CHAPTER 11

Best practices for producing high quality seedlings and establishing main European tree species

Abies alba Mill.

Valeriu-Norocel Nicolescu
Faculty of Silviculture and Forest Engineering, Transilvania University of Braşov, Braşov, Romania

Propagation

Seed propagation

Abies alba (silver fir) trees start bearing seeds between 25 and 30 (35) years of age, but high seed production occurs at ages between 60 and 70 (80) years (Lanier, 1986; Bary-Lenger et al., 1988; Boudru, 1989; Wolf, 2003; Bledý et al., 2024). High seed crops occur every 2–3 years in seed orchards, while in seed stands, they occur every 4–5 years and sometimes every 6 years (Boudru, 1989; Riou-Nivert, 1996; Savill, 2013; McCartan and Jinks, 2015; Mihai et al., 2016). Silver fir cones standing vertically on the uppermost whorls/upper third of the tree crown are harvested maually or using scissors or hooks from trees in seed stands or orchards before they mature, from September to October, 15–20 days before dispersal, when they soften and become brown. Alternatively, cones can be collected on the ground using tarpaulins or plastic sheets after being cut (Iancu, 1999).

As tree height is strongly and positively correlated with seed germination, seeds should be collected from trees that exceed the mean stand height (Ballian, 2013). At harvest, the cones contain 30%–70% water and seed moisture is about 40% (Vlase, 1982; Bledý et al., 2024). Place the cones in ventilated rooms for natural drying; this causes the scales to detach from the cone axis, releasing the seeds. From 1 hL (100 L) of cones, 4–6 kg of winged seeds or 3.5–5.5 kg of dewinged seeds are obtained (Vlase, 1982). The seed wings must be carefully removed manually; damaging the resin vesicles that contain terpenes reduces germination potential, which is typically quite low (ca. 40%) (Iancu, 1999; McCartan and Jinks, 2015; Bledý et al., 2024). Afterward, the seeds can be cleaned of impurities by flotation (in water, alcohol or ether), by using an air separator, or a gravity table (McCartan and Jinks, 2015). Seeds are then sorted by shape, size, and weight, eliminating empty or insect-infected seeds (Damian, 1978; Florescu, 1996). Seed

Guidelines for Climate Adaptive Forest Restoration and Reforestation Projects
ISBN 978-0-443-34086-4
https://doi.org/10.1016/B978-0-443-34086-4.00011-6

© 2026 Elsevier Inc. All rights are reserved, including those for text and data mining, AI training, and similar technologies.

Table 11.1 Characteristics of seed lots of *Abies alba*.

Purity (%)	No. seeds/kg	Germination capacity (%)
80–98 (90)	13,000–30,000 (20,000)	20–40 (30)

Mean values in brackets.
Haralamb (1967); Lanier (1986); Bary-Lenger et al. (1988); Boudru (1992); Florescu (1996); Iancu (1999).

cleaning and sorting are essential, as seed crops of *A. alba* often contain a high proportion of empty seeds, primarily due to inadequate pollination and seed pests such as chalcids (*Megastigmus suspectus*) and midges (*Resseliella piceae*) (McCartan and Jinks, 2015).

The average characteristics of seed lots are summarized in Table 11.1.

Dry the cleaned seeds in an unheated, ventilated room. Spread the seed in 5 cm thick layers, turning them two to three times every day for 2–3 weeks, until seed moisture is reduced to 12%–13% before winter storage (Vlase, 1982). Store the seeds in unheated rooms, on wooden or cement floors in layers 10–15 cm thick.

Alternatively, seeds can be stored for up to 1 year with a moisture content of 8%–10% in hermetically sealed (glass or aluminum) containers in unheated rooms. The maximum summer temperature in the room should not exceed 15°C. Another option for storing seeds for up to 1 year is to dry them to a moisture content of 11%–12%, pack the seeds in hermetically sealed plastic bags and store them at −5°C (±1°C) (Mihai et al., 2016). To store the seeds for up to 1.5 years, seeds can be kept at 8%–10% moisture content but at a temperature between 0° and 5°C (Vlase, 1982). For storage up to 3 years, the temperature should be lowered to −15°C, with seed moisture maintained at 12%–13%. For periods longer than 3 years, moisture content should be reduced to 8%–9% (Vlase, 1982). Finally, seeds can be stored up to 8 years in hermetically sealed plastic bags with a moisture content of 6%–7% at −5°C (±1°C) (Mihai and Alexandru, 2020).

A. alba seeds belong to the category of "short germination period seeds," that is, they germinate, and seedlings emerge immediately after seeding, without any special treatment (Damian, 1978; Florescu, 1996). Seeds show a short dormancy that is easily broken. This process involves soaking the seeds in water for 24 h, draining the water and drying outer surfaces of the seeds, and then placing them in a zip-locked freezer bag in a refrigerator for 6–8 weeks. After storage they are ready for sowing in March (Savill, 2013). Due to the high proportion of empty seeds, germination rates in *A. alba* are highly variable, ranging from 15%–70% (Ballian, 2013), 5%–80% (McCartan and Jinks, 2015), and 25%–35% (Morar et al., 2023), making seed lot upgrading essential. A three-stage process, known as imbibition-drying-separation (IDS) is based on the principle that viable seeds retain imbibed water better than dead seeds during drying and this can improve seed lot quality (McCartan and Jinks, 2015).

Vegetative propagation

A. alba can be vegetatively propagated using herbaceous or lignified cuttings. Cuttings must be harvested during the winter when mother plants are dormant and used immediately. Shoot cuttings must be 15–20 cm long. Bevel the apical part of the cutting just

above a bud; the base of the cutting must be cut perpendicularly just below a bud. Before placing the cutting in a hot bed, the base must be treated with fungicide such as a mixture of 5.5% benomyl and talcum powder (Anonymous, 2022). Insert the cutting into a rooting medium up to one-third of its length.

Cuttings root when the growing media temperature ranges between 10 and 15°C, which can be achieved in greenhouses or in hot beds. The air temperature should be around 20°C, with high relative humidity. The cuttings should be well watered but avoid waterlogging (Florescu, 1996). Treat cuttings with 100 mg/L indole-3-butyric acid to accelerate rooting and shoot growth (Haralamb, 1967). The most common rooting media used are coarse sand, vermiculite, a blend of equal parts peat moss and vermiculite, or a blend of equal parts peat moss and perlite (Fast-growing-trees.com).

Leaf-bearing cuttings show higher rooting than cuttings without leaves. Several studies have shown positive effects of mycorrhiza inoculum on rooting (e.g., Banach et al., 2013). Combining natural or synthetic hormones (e.g., indole-3-acetic acid (IAA), indole-3-butyric acid (IBA); Abrudan, 2006) with a mycorrhizal enriched preparation (Banach et al., 2013) or pieces of root colonized by mycorrhizae increases rooting success.

Seedling cultivation

Container plants

Multipot seed trays, usually with cells of 300 mL volume are recommended for cultivating *A. alba* seedlings (Banach et al., 2013). Peat-based substrates can be used, with a pH of 4.2–6.0. Inoculation with mycorrhizal fungi, such as *Lactarius* spp. or *Hebeloma crustuliniforme* can improve seedling quality and survival (Banach et al., 2013; Bledý et al., 2024). Seedlings usually grow between 2 and 5 cm in the first growing season and after 3 years they are ready to be transplanted in the field (Treeseedonline.com).

Bare root plants

Abies alba seedlings can be produced either in the nursery field or in a dual greenhouse (polyhouse) and nursery field system. Seeds are sown either in spring (March-early April) or fall (September–October) (Lanier, 1986; Bary-Lenger et al., 1988; Iancu, 1999). For spring sowing, seeds are stratified in sand or humus at 3–5°C for 21 days (Bledý et al., 2024). If stored in containers, seeds can be soaked in a 15% potassium permanganate solution, an oxidizing agent with bactericidal properties, for 3–5 days before sowing. No special preparation is needed for fall sowing (Bary-Lenger et al., 1988; Iancu, 1999).

Field sowing is done in rills 1.5–2 cm deep, spaced 20 cm apart. Using top quality seeds (class I—65 g minimum weight of 1000 seeds, 95% minimum purity, 45% minimum germination capacity, 23,000 maximum number of seeds/kg), recommended sowing rates range between 22 g of seeds/m (1100 kg/ha) in Romania (Iancu, 1999) and 30–35 g of seeds/m (1500–2000 kg/ha) in Belgium (Bary-Lenger et al., 1988; Boudru, 1992). The seeds are covered with a homogenous mix of *Fagus sylvatica* humus (50%), *P. abies* humus (20%), *P. abies* litter (20%) and sand (10%). A protecting layer of leaves, 5–10 cm thick, is laid over this mix to protect the seeds against frost in fall sowings or to maintain soil humidity in spring sowing.

Seedlings emerge in 3–4 weeks, sometimes 6 weeks after sowing (Lanier, 1986; Boudru, 1992). The seedling has 5–6 green cotyledons that drop off in year 3 or 4. In the first year, only one whorl of leaves grows. Growth is very slow and only a terminal bud is formed in the fall. In year 2, the terminal twig has stronger growth, and lateral buds are formed in the fall.

The optimum culture density is 30–40 plants/m of rill if the seedlings are to be transplanted but only 20–30 plants/m if not transplanted. The mean collar diameter of plants 1–2 years old is 1 mm (Iancu, 1999).

Seedlings are grown in the field for 2 years then transplanted again in the field to grow for 1, 2, or even 3 more years. Spacing between plants along the row is maintained at 5–10 cm with 20 cm between rows. Consequently, the production cycle is 3–4 (5) years (2 + 1, 2 + 2, or 2 + 3) (Vlase, 1982; Bary-Lenger et al., 1988; Boudru, 1992), with seedlings developing a minimum root collar diameter of 5 mm (Iancu, 1999; Abrudan, 2006). Seedlings grow slowly, reaching only 15–30 cm in height at age 3(5) but they develop strong tap roots and many lateral roots after the first year (Haralamb, 1967).

In the nursery field, any soil crust formed following heavy rains should be broken immediately using mechanical cultivators. Hoeing to a depth of 5–8 cm is also recommended at the beginning of the growing season. Weed control using chemicals with residual substances (e.g., Pitezin, Simazine) is applied the day after sowing (Iancu, 1999).

In the nursery field, plants must be fully shaded during the growing season with lighter shade in later years (Iancu, 1999). *A. alba* seedlings are susceptible to common cockchafer (*Melolontha melolontha*) and European mole cricket (*Gryllotalpa gryllotalpa*), both affecting the roots of young plants (Iancu, 1999). Several biological and chemical methods can be used to control both pests (Held and Cobb, 2016; Woreta, 2015).

The production cycle in a polyhouse takes 2 years. The seedbed is prepared with a mix of 50% *Picea abies* humus, 30% *Picea abies* litter, 20% *Fagus sylvatica* humus as well as nitrogen, phosphorus and potassium fertilizers. Then the seedbed is disinfected with 1% Formalin and seeds are sown (either in the spring or in fall), at a depth of 1–2 cm, in rows 4 cm apart. Finally, seeds are covered with a mixture of three parts conifer humus and one part sand, about 1 cm thick (Iancu, 1999).

The upper part of the seedbed is kept moist until seedlings emerge, by watering every day with 1 L/m^2 for the first 30 days after sowing and then 2–5 L/m^2 every 2–3 days. Watering is stopped at the beginning of August to allow seedlings to harden. Weeding is done manually or with herbicides (0.6 kg of Pitezin or Simazine diluted in 400–600 L of water), immediately after seeding and only once during the growing season (Iancu, 1999). About 1 month after seedlings emerge, the weakest and most crowded plants are thinned with scissors.

The polyhouse should be ventilated when the seedbed temperature reaches 30°C (Iancu, 1999). Disease protection begins before sowing by treating the seeds, seedbeds and paths with hydrogen peroxide, H_2O_2. Seedlings should be sprayed against fusariosis after emerge with 1% Bordeaux mixture (1 kg $CuSO_4$ + 1 kg CaO, mixed in 100 L of water).

Irrigation water needs to be free of harmful salts, toxic substances, and its temperature should be close to that of the soil especially when cultivated in the field. After each watering, the soil should be worked superficially. In mountain areas in Romania, where the



Figure 11.1 *Abies alba* germinants in the seedbed (left) and 2 + 2 seedlings (right). *(Vladan Ivetić.)*

majority of *A. alba* forest nurseries are located, two irrigations per month are needed in field cultivation: 300 m^3/ha in May, 500 m^3/ha in June, 600 m^3/ha in July, and 500 m^3/ha in August (Iancu, 1999).

Seedlings are stored in trenches 30–40 cm deep and the soil compacted to prevent air penetration, and watered if necessary. Seedlings bundles are placed in the trench, leaning against the wall on the windward or southern side. They are covered with 8–10 cm of soil, ensuring that the roots and 4–5 cm of the stem are buried (Damian, 1978; Florescu, 1996; Iancu, 1999). *A. alba* plants must be transported quickly to the planting site, preferably at night or during cool, wet conditions, with the roots treated with anti-desiccants to improve seedling survival (Bledý et al., 2024). Upon arrival at the planting site, seedlings should be stored in a ditch, watered, and covered with soil to prevent root desiccation (Iancu, 1999) (Fig. 11.1).

Field establishment

We recommend spring planting without any previous soil preparation. Plants typically are 3–4 (even 5) years-old, with a minimum root collar diameter of 5 mm and a height of 20–30 cm (Vlase, 1982). They can be planted in holes 20 cm deep within a surrounding weed-free area of 40 × 60 cm to 60 × 80 cm (Haralamb, 1967; Iancu, 1999). The initial planting density ranges from 2500 seedlings/ha (2.0 × 2.0 m) (Bary-Lenger et al., 1988; Boudru, 1992) to 5000 seedlings/ha (2.0 × 1.0 m) (Anonymous, 2022). As seedlings are sensitive to direct sun exposure, they need some shelter in the first years. This can be achieved by planting on the northeast side of stumps, deadwood, large stones, or elderberry or raspberry bushes (Haralamb, 1967; Iancu, 1999).

Direct seeding of *A. alba* has recently been recommended for converting *Picea abies* stands in Central Europe as a close-to-nature approach. Autumn seeding is favored due to the use of fresh seeds and avoidance of costs and equipment associated with seed storage (Huth et al., 2017). However, autumn seeding has disadvantages, including a limited time window between seed harvest and site preparation and increased risks to freshly sown seeds without snow cover from winter frosts (Huth et al., 2017). Alternatively, Kutscher (2011) documented higher germination rates with spring seeding, suggesting

that both seasons are viable options, to be selected based on specific forest conditions and operational requirements (Huth et al., 2017).

A. alba seedlings grow slowly in height the first years, speeding up at 8–10 years of age when they start to take a pyramidal shape (Haralamb, 1967; Iancu, 1999). Early care is important, including mowing or mulching within a 0.5 m radius around the plants in the first 2–3 years after establishment (Damian, 1978). Weeding is recommended, including pioneer woody species (e.g., *Salix capraea*, *Populus tremula*, and *Betula pendula*). Tall weeds (undergrowth, stump sprouts, etc.) can bend over or break the seedlings in the winter.

A. alba seedlings are highly susceptible to animal damage due to their high palatability and slow juvenile growth such as bud browsing, bark stripping, browsing, and fraying by large wild herbivores like red deer (*Cervus elaphus*) and roe deer (*Capreolus capreolus*) (Mauri et al., 2016; Bledý et al., 2024; Frei et al., 2024; Konôpka and Šebeň, 2024). In montane forests of Central and Southeastern Europe, *A. alba* is the most heavily browsed among important tree species, leading for decades to consistently poor or even failed natural regeneration (Konôpka and Šebeň, 2024). Protecting seedlings with repellents or fencing can reduce ungulate damage, although both are costly and only effective during early growth stages (Konôpka and Šebeň, 2024). Sustainable herbivore population management through effective hunting practices is likely the most efficient long-term measure for reducing browsing damage in *A. alba* forests (Wolf, 2003; Frei et al., 2024; Konôpka and Šebeň, 2024).

Fertilizers, tree shelters, and irrigation are generally considered unnecessary for young silver fir plantations (Haralamb, 1967; Damian, 1978; Iancu, 1999). However, when applied, fertilizers (in granular, tablet, or powder form) have been shown to improve seedling height growth (Kupka, 2005). Similarly, using tree shelters (e.g., Lay-flat Shelterguards) has improved survival rates (92% in the fifth year postplanting) and stimulated height growth in young silver fir (Petkova et al., 2022).

Chapter References

Abrudan, I.V., 2006. Împăduriri (Afforestations). Editura Universitătii Transilvania, Brasov.

Anonymous, 2022. Ghidul de bune practici privind compoziţii, scheme şi tehnologii de regenerare a pădurilor şi de împădurire a terenurilor degradate (Good practice guide on compositions, schemes and technologies for reforestation and afforestation of degraded land). In: Monitorul Oficial Al Romaniei, Bucuresti, Partea I, No. 1000 Bis, 14 Octombrie 2022, pp. 5–382.

Ballian, D., 2013. Genetic overload of silver fir (*Abies alba* Mill.) from five populations from central Bosnia and Herzegovina. In: Folia Forestalia Polonica, Series A, 2013, vol 55, pp. 49–57. https://doi.org/10.2478/ffp-2013-0006, 2.

Banach, J., Skrzyszewska, K., Świeboda, L., 2013. Substrate influences the height of one- and two-year-old seedlings of silver fir and European beech growing in polystyrene containers. Leśne Prace Badawcze 74 (2), 117–125. https://doi.org/10.2478/frp-2013-0012 (Forest Research Papers).

Bary-Lenger, A., Evrard, R., Gathy, P., 1988. La forêt: écologie-géstion-economie-conservation. In: Troisième edition (The forest: ecology-management-economy-conservation. 3rd edition). Editions du Perron, Liège.

Bledý, M., Vacek, S., Brabec, P., Vacek, Z., Cukor, J., Černý, J., Ševčík, R., Brynychová, K., 2024. Silver Fir (*Abies alba* Mill.): review of ecological insights, forest management strategies, and climate change's impact on European forests. Forests 15 (6), 998. https://doi.org/10.3390/F15060998.

Boudru, M., 1989. Forêt et sylviculture: sylviculture appliquée (Forest and Silviculture: Applied Silviculture). Les Presses Agronomiques de Gembloux, Gembloux.

Boudru, M., 1992. Forêt et sylviculture: boisements et reboisements artificiels (Forest and Silviculture: Artificial Afforestation and Reforestation). Les Presses Agronomiques de Gembloux, Gembloux.
Damian, I., 1978. Împăduriri (Afforestations). Editura Didactică Si Pedagogică, Bucureşti.
Florescu, G., 1996. Împăduriri. Seminţe Forestiere (Afforestations. Forest Seeds). Universitatea Transilvania din Braşov, Braşov.
Frei, E.R., Conedera, M., Bebi, P., Zürche, S., Bareiss, A., Ramstein, L., Giacomelli, N., Bottero, A., 2024. High Potential but Little Success: Ungulate Browsing Increasingly Impairs Silver Fir Regeneration in Mountain Forests in the Southern Swiss Alps. Forestry, pp. 1–10. https://doi.org/10.1093/forestry/cpae028 cpae028.
Haralamb, A., 1967. Cultura Speciilor Forestiere. Ediţia a III-A Revăzută Si Adăugită (Culture of Forest Species, third ed. Editura Agro-Silvică de Stat, Bucureşti.
Held, D., Cobb, P., 2016. Biology and control of Mole crickets. In: Extension Alabama A&M and Auburn Universities, Technical Report ANR-0176. USA.
Huth, F., Wehnert, A., Tiebel, K., Wagner, S., 2017. Direct seeding of silver fir (*Abies alba* Mill.) to convert Norway spruce (*Picea abies* L.) forests in Europe: a review. For. Ecol. Manag. 403, 61–78. https://doi.org/10.1016/J.FORECO.2017.08.017.
Iancu, I., 1999. Manualul Pădurarului. Teoria Si Practica Meseriei (Manual of Forest Ranger. Theory and Practice). Tipografia Pegasus, Bucureşti.
Konôpka, B., Šebeň, V., 2024. Positive impact of large wild herbivore exclusion on silver fir regeneration: a case study from the Poľana Mountains, Central Slovakia. Central Europ. Forestry J. 70 (4), 248–262.
Kupka, I., 2005. Reaction of silver fir (*Abies alba* Mill.) plantation to fertilization. J. For. Sci. 51, 95–100.
Kutscher, M.T., 2011. Verjüngung von Fichtenreinbeständen in den Flyschbergen und im Kalkalpin mittels Saat (Rejuvenation of pure Norway spruce stands in the Flysch Mountains and the Limestone Alps using seeding). Technische Universität München, Fachgebiet Waldernährung und Wasserhaushalt, p. 234. PhD thesis.
Lanier, L., 1986. Précis de sylviculture (Forestry Handbook). Ecole Nationale du Génie Rural, des Eaux et des Forêts, Nancy.
Mauri, A., de Rigo, D., Caudullo, G., 2016. *Abies alba* in Europe: distribution, habitat, usage and threats. In: San-Miguel-Ayanz, J., de Rigo, D., Caudullo, G., Houston Durant, T., Mauri, A. (Eds.), European Atlas of Forest Tree Species. Publication Office of the EU, Luxembourg e01493b+.
McCartan, S.A., Jinks, R.L., 2015. Upgrading seed lots of European silver fir (*Abies alba* Mill.) using imbibition-drying-separation. Tree Plant. Notes U.S. Dep. Agric. For. Serv. 58 (2), 21–27.
Mihai, G., Mărgineanu, M., Daia, M.L., 2016. Rezultate preliminare privind conservarea semințelor de brad (*Abies alba* Mill) în România (Preliminary results concerning the conservation of European silver fir seeds (*Abies alba* Mill) in Romania). Rev. Padurilor 3–4, 3–6.
Mihai, G., Alexandru, A.M., 2020. Conservarea semințelor de brad (Conservation of silver fir seeds). Editura Silvică, Voluntari.
Morar, I.M., Dan, C., Sestraş, R.E., Stoian-Dod, R.L., Truţă, A.M., Sestraş, A.F., Sestraş, P., 2023. Evaluation of different geographic provenances of silver fir (*Abies alba*) as seed sources, based on seed traits and germination. Forests 14, 2186. https://doi.org/10.3390/f14112186.
Petkova, K., Iliev, N., Borisov, M., Sattler, M., 2022. Effect of tree shelters on the survival and growth of coniferous tree species in Central South Bulgaria. Silva Balcan. 23 (1), 67–78. https://doi.org/10.3897/silvabalcanica.22.e84286.
Riou-Nivert, P., 1996. Les résineux. Tome I: Connaissance et reconnaissance (Conifers. Volume I: Knowledge and recognition). Institut pour le développement forestier, Paris.
Savill, P., 2013. The Silviculture of Trees Used in British Forestry, second ed. CAB International, Wallingford and Boston.
Vlase, I., 1982. Conservarea Semințelor Forestiere (Conservation of Forest Seeds). Editura Ceres, Bucureşti.
Wolf, H., 2003. EUFORGEN Technical Guidelines for Genetic Conservation and Use for Silver Fir (*Abies alba*). International Plant Genetic Resources Institute, Rome, Italy.
Woreta, D., 2015. Control of cockchafer *Melolontha* spp. grubs – a review of methods. Folia Forest. Polon. 57 (1), 33–41.

Web references

https://www.fast-growing-trees.com/pages/propagating-with-cuttings (Accessed October 31, 2024).
https://www.treeseedonline.com/store/p50/European_Silver_Fir_%28abies_alba%29.html (Accessed October 31, 2024).

Acer L. (*Acer campestre* L., *Acer platanoides* L., *Acer pseudoplatanus* L.)

Anastazija Dimitrova[1], Vladan Popović[2] and Aleksandar Vemić[2]
[1]Ss. Cyril and Methodius University in Skopje, Hans Em Faculty of Forest Sciences, Landscape Architecture and Environmental Engineering, Skopje, North Macedonia; [2]Institute of Forestry, Department of Genetics, Plant breeding, Seed and Nursery Production, Belgrade, Serbia

Propagation

Seed propagation

The fruits of *Acer* species, known as samaras, ripen in the mid-autumn (October), when they turn brown, indicating that they are ready for collection (Bacchetta, 2006; Navarro Cerrillo and Herrero Sierra, 2012). Fruit production varies significantly between years. Although most seeds fall easily, some can remain on the tree until the next reproductive season (Bonner and Karrfalt, 2008; Navarro Cerrillo and Herrero Sierra, 2012). Fruits are usually collected manually, using methods such as raking, sweeping, specialized vacuum cleaners, or by shaking or cutting branches (Božič et al., 2021). After collection, fruits of *Acer* with orthodox seeds (see below) should be dried on a nonhumidity absorbing surface, such as wood, in a well-ventilated place, with regular turning to ensure uniform drying (Burdekin and Rowe, 1982). Removing the wings from the samaras with machines, like bending-type devices, is uncommon as it can reduce the fruit quality (Gordon, 1992; Piotto and Di Noi, 2003; Navarro Cerrillo and Herrero Sierra, 2012).

The orthodox seeds of *A. campestre* and *A. platanoides* remain viable up to 4 years, though viability gradually decreases, when properly stored, with a moisture content between 10% and 15% and temperatures <0°C (down to −10°C) (Burdekin and Rowe, 1982; Gordon, 1992; Navarro Cerrillo and Herrero Sierra, 2012). The seeds often have endogenous dormancy, requiring a period of cold stratification. *A. campestre* seeds show high dormancy heterogeneity and germination can be done in different ways (Navarro Cerrillo and Herrero Sierra, 2012). The seeds may be sown in autumn, immediately after harvest, or dormancy can be broken within 90 days by soaking in water at 40°C for 3 days (Navarro Cerrillo and Herrero Sierra, 2012). If sown in spring, the seeds need either 3–8 weeks of warm stratification followed by 12–24 weeks of cold stratification or, simply 13 weeks of cold stratification (Piotto and Di Noi, 2003). *A. platanoides* seeds are deeply dormant at maturity, and cold stratification for 13 weeks at 3–4°C is needed to release dormancy and achieve high germination rates (Pawlowski and Szczotka, 1997). In both species, seeds must be monitored during stratification and sown as soon as they begin to swell as they easily germinate at low temperatures (3–5°C) (Piotto and Di Noi, 2003). Seed size and viability in *A. platanoides* are greater in populations from colder, wetter environments compared with those from warmer, drier areas (Carón et al., 2014).

Unlike *A. campestre and A. platanoides*, *A. pseudoplatanus* seeds are recalcitrant, and desiccation or freezing hinders their vitality (Dickie et al., 1991; Pukacki and Juszczyk, 2015). They can germinate within a range of 3–20°C (Tylkowski, 1989). Generally, warming and drought reduce germination (Carón, 2014). For short-term storage, the

seeds of *A. pseudoplatanus* should be stratified at −3°C and those that have started germinating can be stored at 3°C for up to 12 months. For optimal storage, the seeds should be kept until the following spring at a temperature of −3 to +5°C with high humidity (Gosling, 2007). Gibberellic acid enhances germination of *A. pseudoplatanus* seeds (Stejskalová et al., 2015), which can germinate during cold stratification after 42 days (Stejskalová et al., 2014). The highest germination rates occur with autumn seeding (Yücesan and Bayram, 2021). For spring seeding, it is recommended to stratify seeds at 0–5°C for 3–12 weeks, with a humidity of 44%–58% (Iliev et al., 2022). The characteristic of seed lots of the three *Acer* species are shown in Table 11.2.

Vegetative propagation

Vegetative propagation of *A. campestre* is uncommon due to the difficulty and the poor vigor of resulting plants (Navarro Cerrillo and Herrero Sierra, 2012). If used, hardwood or semihardwood cuttings, 20–25 cm long, have the lower 10 cm buried in a loose substrate, typically a 2:1 mixture of peat: perlite (Navarro Cerrillo and Herrero Sierra, 2012; Piotto and Di Noi, 2003). Rooting may be enhanced by applying hormone treatments, such as 8000 mg/L of indole-3-butyric acid (IBA), and the cuttings should be placed on rooting tables with basal heating (18–20°C) and misting (Navarro Cerrillo and Herrero Sierra, 2012).

Although *A. platanoides* can be easily propagated by seeds, vegetative propagation is preferred for cultivars, especially in urban areas, to ensure consistent phenotypic traits (Tomov, 2007). As cuttings root poorly, grafting onto seedling rootstock is more commonly used (Iliev and Tomov, 2017). Treatments such as shoot etiolation, basal banding with black tape, and applying 0.8% IBA powder can promote rooting (Tomov, 2007).

A. pseudoplatanus plants propagated from cuttings outperform those grown from seed in the early years of growth (Iliev et al., 2022; Strimbu and Nicolescu, 2023). Softwood or semihardwood shoots should be collected from spring to mid-summer (Hartmann and Kester, 2002; Spethmann, 2007). As with *A. platanoides,* auxin also stimulates rooting in *A. pseudoplatanus* cuttings (Hartmann and Kester, 2002). Cuttings should be 28–35 cm in length. Rooting *A. pseudoplatanus* is difficult but can be improved by pruning and shading donor plants in the previous season before collecting cuttings (Maynard et al., 1996). Applying fungicide treatments to prevent bark damage, a common issue for *A. pseudoplatanus*, is also recommended. Suitable fungicides include copper-based compounds, dithiocarbamates, triazoles, dinitrophenols, and phthalimides.

Table 11.2 Characteristics of seed lots of *Acer campestre, Acer platanoides,* and *Acer pseudoplatanus.*

Species	Purity (%)	No. seeds/kg	Germination capacity (%)
Acer campestre	92	8,600–25,000 (16,800)	40–80 (60)
Acer platanoides	86	2,800–10,300 (6550)	40–70 (55)
Acer pseudoplatanus	90	5400–15,800 (10,600)	40

Mean values in brackets.
Božič et al. (2021); Burdekin and Rowe (1982); Piotto and Di Noi (2003).

A substrate of peat or a peat-perlite mix is recommended for rooting cuttings (Hartmann and Kester, 2002). While *A. pseudoplatanus* forms arbuscular mycorrhizae (Rusterholz et al., 2020), the specific mycorrhizal type is not critical for root development in temperate regions (Kubisch et al., 2015). However, intensive activities in stands established from vegetative material should be avoided in the first few years to preserve the diversity of mycorrhizal fungi in the roots (Heklau et al., 2023).

Seedling cultivation

Container plants

Because nursery germination is often low (around 14%), germination can be delayed, and seedlings tolerate transplanting, it is recommended to use seedbeds (Navarro Cerrillo and Herrero Sierra, 2012). The same seedbed can be used for two growing seasons, and the seedlings can be transplanted once the two embryonic leaves are fully developed and perpendicular to the stem, just before the true leaves emerge (Navarro Cerrillo and Herrero Sierra, 2012). As container plants, the seedlings need to be transplanted into 300–400 cm^3 containers, in which plants can grow to 30–40 cm tall with a well-formed root system. In some cases, especially for restoration and landscaping use, containers with larger volumes (<3500 cm^3) can be used to obtain larger plants (40–60 cm) (Navarro Cerrillo and Herrero Sierra, 2012). Periodic treatments with broad-spectrum fungicides and soil insecticides are needed to prevent nematodes (Navarro Cerrillo and Herrero Sierra, 2012).

A. campestre is considered a slow-growing species, so maximum growth is obtained by longer growing cycles that result in compact plants, but not shorter than 30 cm. The container substrate can be various mixtures, between a minimum of 75% common material (peat moss, peat hummus, or coconut fiber) and a maximum 25% of an inorganic material (perlite, vermiculite, or sand). As there is no specific knowledge about fertilization composition and dosage, slow-release fertilizers are commonly used, for example, 18-11-10 NPK over 8–9 months with 2 g/L substrate or 14-8-15 NPK over 8–9 months with 2.5 g/L substrate (Navarro Cerrillo and Herrero Sierra, 2012). For a shorter cultivation period, an alternative is fertilized peat (with base fertilizer 16-8-16 NPK and pH correction), but control and possible adjustments need to be considered (Navarro Cerrillo and Herrero Sierra, 2012). Inoculation with arbuscular mycorrhizae fungi in the nursery could increase the survival rate and the growth after transplanting (Corsini et al., 2022).

Bare root plants

A. pseudoplatanus seeds cannot be stored for extended periods, so it's best to sow them in the spring or autumn at the beginning of the cultivation period. The growth of *A. pseudoplatanus* is hindered in dry winters; the weather conditions in July and October of the previous year and March of the current year significantly impact its radial growth (Vacek et al., 2017). In areas where it behaves as an invasive species, *A. pseudoplatanus* demonstrates better growth due to its adaptability to light conditions and phenotypic plasticity (Shouman et al., 2017). In addition, climate warming contributes to the regeneration of *A. pseudoplatanus* (Carón et al., 2015).

Liming low pH soils increases the growth of *A. pseudoplatanus* (Balcar et al., 2011). Intensive irrigation increases the mass of fine roots but also results in a reduction in the root:shoot dry-weight ratio compared with low irrigation (Hipps et al., 1996). Avoiding graywater irrigation is recommended as *A. pseudoplatanus* is sensitive to high salinity (Gräf et al., 2022). *A. pseudoplatanus* seedlings face attacks from various harmful organisms, including *Rhytisma acerinum, Nectria cinnabarina, Eutypella parasitica, Verticillium dahliae, Verticillium albo-atrum, Phytophthora* spp., and chewing insects (Sinclair and Lyon, 2005). Copper fungicides and triazoles are effective against most of these pathogens, while phenylamides (such as metalaxyl, benalaxyl, cymoxanil) are recommended against *Phytophthora* spp. Neonicotinoids or bioinsecticides are used against the insects damaging the leaves.

Recommended stock type for *A. pseudoplatanus* is 1 + 1 seedlings, with a height of 25–50 cm (Evans 1984). Regarding *A. campestre*, Navarro Cerrillo and Herrero Sierra (2012) indicated that it can be cultivated as 1 + 2 bare root stock with plants reaching a height of 80–100 cm. Among the provenances in Western Europe, no differences in growth and tree form have been found (Whittet et al., 2021). In large trees for urban forestry and ornamental purposes, there is no optimal period for pruning *A. pseudoplatanus* (Fini et al., 2013) (Fig. 11.2).

Field establishment

A. campestre is naturally present across Europe, except in the northern parts. It does not form pure stands and it is commonly a subdominant species in numerous plant communities (Chybicki et al., 2014). The main limitations for seedling establishment are the low viability of the seeds, insufficient growth, and sensitivity of the seedlings in the first 3 years to stressors, especially drought. Summer watering and mulching with straw can partially overcome drought (Navarro Cerrillo and Herrero Sierra, 2012). Planting seedlings is the recommended revegetation method. If direct seeding is chosen, it should be performed in autumn (Navarro Cerrillo and Herrero Sierra, 2012). If planting is preferred, using larger seedlings, especially those cultivated in large containers (>1000 mL) increases



Figure 11.2 *Acer pseudoplatanus* 1 + 0 container seedlings (left) and bare root seedlings (right). *(Vladan Ivetić.)*

establishment rates. In sites with large herbivores, seedlings must be protected by fencing or by using tall tree shelters (Navarro Cerrillo and Herrero Sierra, 2012).

A. campestre seedlings can be planted in groups at favorable locations to increase survival rates. For instance, high planting densities (200 plants/ha) are recommended on lower parts of slopes, where water availability and soil depth are greater. This density should be reduced to 100 plants/ha on the upper part of slopes, promoting more complex vegetation with moderate competition and healthy plants with good branching and vigor (Navarro Cerrillo and Herrero Sierra, 2012).

We recommend digging large, deep holes (40 × 40 × 60 cm) as a soil preparation method with removal of weeds. Although *A. campestre* is tolerant of poor soil conditions, soils prone to waterlogging should be avoided (Banks et al., 2019; Navarro Cerrillo and Herrero Sierra, 2012).

A. platanoides is generally tolerant to prolonged periods of dry and very wet soil, but this can vary among cultivars, which is important to know during the selection of reproductive material (Banks et al., 2019).

The main factors that limit the establishment of *A. pseudoplatanus* are closed-canopy stands, competition from weeds, and mammal damage (Hein et al., 2009). *A. pseudoplatanus* thrives on different types of soil, and with its developed roots, it can rehabilitate places exposed to erosion (Caudullo and de Rigo, 2016). Seedlings develop poorly in contaminated soils, but there are several tolerant genotypes that show better health (Turner and Dickinson, 1993).

Climate change threatens the development of *A. pseudoplatanus* seedlings (Carón et al., 2015). Seeds germinate best in autumn (Yücesan and Bayram, 2021), and seedlings generally have high frost resistance (Piovesan et al., 2005). Therefore, regeneration in autumn is recommended. Waterlogged soils must be avoided and planting should be concentrated on well drained soils with higher clay and nitrogen content (Jensen et al., 2008). In mixed plantations of *A. pseudoplatanus* and *Fagus sylvatica*, it is crucial to increase the density of *A. pseudoplatanus,* as *F. sylvatica* absorbs N faster and is more competitive (Li et al., 2015). To protect seedlings against harsh weather conditions, managing canopy openings is important to prevent microclimate disturbances in the stands. Manual removal of weeds should be favored over herbicides to avoid environmental issues.

Chapter References

Bacchetta, G., 2006. Manuale Per La Raccolta, Studio, Conservazione E Gestione Ex Situ Del Germoplasma. APAT, Roma.

Balcar, V., Kacalek, D., Kunes, I., Dusek, D., 2011. Effect of soil liming on European beech (*Fagus sylvatica* L.) and sycamore maple (*Acer pseudoplatanus* L.) plantations. Folia Forestalia Polonica. Series A. Forestry 53, 85–92.

Banks, J.M., Percival, G.C., Rose, G., 2019. Variations in seasonal drought tolerance rankings. Trees 33, 1063–1072. https://doi.org/10.1007/s00468-019-01842-5.

Bonner, F.T., Karrfalt, R.P., 2008. The woody plant seed manual. In: Agriculture Handbook 727. US Department of Agriculture Forest Service, Washington, DC.

Božič, G., Westergren, M., Konrad, H., Lanšćak, M., Nagy, L., Novčić, Z., Rukavina, S., Schüler, S., Stojnić, S., Železnik, P., 2021. Guidelines for the Production of Planting Material for Restoration of

Riparian Forests. Slovenian Forestry Institute, Silva Slovenica Publishing Centre. https://doi.org/10.20315/SFS.170.
Burdekin, D.A., Rowe, D.C.F., 1982. Seed Manual for Ornamental Trees and Shrubs, Forestry Commission Bulletin. HMSO, London.
Carón, M., 2014. Regeneration from Seeds of Two *Acer* Species in the Face of Climate Change (PhD Thesis). Ghent University. Faculty of Bioscience Engineering, Ghent, Belgium.
Carón, M.M., De Frenne, P., Brunet, J., Chabrerie, O., Cousins, S.A.O., De Backer, L., Decocq, G., Diekmann, M., Heinken, T., Kolb, A., Naaf, T., Plue, J., Selvi, F., Strimbeck, G.R., Wulf, M., Verheyen, K., 2015. Interacting effects of warming and drought on regeneration and early growth of *Acer pseudoplatanus* and *Acer platanoides*. Plant Biol. J. 17, 52–62. https://doi.org/10.1111/plb.12177.
Carón, M.M., De Frenne, P., Brunet, J., Chabrerie, O., Cousins, S.A.O., De Backer, L., Diekmann, M., Graae, B.J., Heinken, T., Kolb, A., Naaf, T., Plue, J., Selvi, F., Strimbeck, G.R., Wulf, M., Verheyen, K., 2014. Latitudinal variation in seeds characteristics of *Acer platanoides* and *A. pseudoplatanus*. Plant Ecol. 215, 911–925. https://doi.org/10.1007/s11258-014-0343-x.
Caudullo, G., de Rigo, D., 2016. *Acer platanoides* in Europe: distribution, habitat, usage and threats. In: San-Miguel-Ayanz, J., de Rigo, D., Caudullo, G., Houston Durrant, T., Mauri, A. (Eds.), European Atlas of Forest Tree Species. Publ. Off. EU, Luxembourg p. e019159+.
Chybicki, I.J., Waldon-Rudzionek, B., Meyza, K., 2014. Population at the edge: increased divergence but not inbreeding towards northern range limit in *Acer campestre*. Tree Genet. Genom. 10, 1739–1753. https://doi.org/10.1007/s11295-014-0793-2.
Corsini, D., Vigevani, I., Oggioni, S.D., Frangi, P., Brunetti, C., Mori, J., Viti, C., Ferrini, F., Fini, A., 2022. Effects of controlled mycorrhization and deficit irrigation in the nursery on post-transplant growth and physiology of *Acer campestre* L. and *Tilia cordata* Mill. Forests 13, 658. https://doi.org/10.3390/f13050658.
Dickie, J.B., May, K., Morris, S.V.A., Titley, S.E., 1991. The effects of desiccation on seed survival in *Acer platanoides* L. and *Acer pseudoplatanus* L. Seed Sci. Res. 1, 149–162. https://doi.org/10.1017/S0960258500000829.
Evans, J., 1984. Silviculture of Broadleaved Woodland, Forestry Commission Bulletin. HMSO, London.
Fini, A., Ferrini, F., Frangi, P., Piatti, R., Faoro, M., Amoroso, G., 2013. Effect of pruning time on growth, wound closure and physiology of sycamore maple (*Acer pseudoplatanus* L.). Acta Hortic. 99–104. https://doi.org/10.17660/ActaHortic.2013.990.9.
Gordon, A.G. (Ed.), 1992. Seed Manual for Forest Trees, 1. Publ. Ed, Forestry Commission Bulletin. HMSO, London.
Gosling, P., 2007. Raising Trees and Shrubs from Seed, Practice Guide. Forestry Commission, Edinburgh.
Gräf, M., Pucher, B., Hietz, P., Hofbauer, K., Allabashi, R., Pitha, U., Hood-Nowotny, R., Stangl, R., 2022. Application of leaf analysis in addition to growth assessment to evaluate the suitability of grey-water for irrigation of *Tilia cordata* and *Acer pseudoplatanus*. Sci. Total Environ. 836, 155745. https://doi.org/10.1016/j.scitotenv.2022.155745.
Hartmann, H.T., Kester, D.E., 2002. Plant Propagation: Principles and Practices, seventh ed. Prentice Hall, Upper Saddle River, N.J.
Hein, S., Collet, C., Ammer, C., Goff, N.L., Skovsgaard, J.P., Savill, P., 2009. A review of growth and stand dynamics of *Acer pseudoplatanus* L. in Europe: implications for silviculture. Forestry 82, 361–385. https://doi.org/10.1093/forestry/cpn043.
Heklau, H., Schindler, N., Eisenhauer, N., Ferlian, O., Bruelheide, H., 2023. Temporal variation of mycorrhization rates in a tree diversity experiment. Ecol. Evol. 13 (4), e10002. https://doi.org/10.1002/ece3.10002.
Hipps, N.A., Higgs, K.H., Collard, L.G., 1996. The effect of irrigation and root pruning on the growth of sycamore (*Acer pseudoplatanus*) seedlings in nursery beds and after transplantation. J. Horticult. Sci. 71, 819–828. https://doi.org/10.1080/14620316.1996.11515464.
Iliev, N., Tomov, V., 2017. Factors affecting the grafting/budding of *Acer platanoides* L. cultivars. Propag. Ornam. Plants 17, 29–36.
Iliev, N., Varbeva, L., Alexandrov, N., 2022. Silvicultural aspects of the sycamore (*Acer pseudoplatanus* L.) propagation - overview. SBAL 23, 37–55. https://doi.org/10.3897/silvabalcanica.22.e82756.
Jensen, J.K., Rasmussen, L.H., Raulund-Rasmussen, K., Borggaard, O.K., 2008. Influence of soil properties on the growth of sycamore (*Acer pseudoplatanus* L.) in Denmark. Eur. J. For. Res. 127, 263–274. https://doi.org/10.1007/s10342-008-0202-1.

Kubisch, P., Hertel, D., Leuschner, C., 2015. Do ectomycorrhizal and arbuscular mycorrhizal temperate tree species systematically differ in root order-related fine root morphology and biomass? Front. Plant Sci. 6. https://doi.org/10.3389/fpls.2015.00064.

Li, X., Rennenberg, H., Simon, J., 2015. Competition for nitrogen between *Fagus sylvatica* and *Acer pseudoplatanus* seedlings depends on soil nitrogen availability. Front. Plant Sci. 6. https://doi.org/10.3389/fpls.2015.00302.

Maynard, B., Johnson, W., Holt, T., Hoogendoorn, D., 1996. Stock plant shading to increase rooting of paperbark maple cuttings. In: Combined Proceedings-International Plant Propagators Society, pp. 611–613, 46.

Navarro Cerrillo, R.M., Herrero Sierra, N., 2012. *Acer campestre* L. In: Pemán García, J., Navarro Cerillo, R.M., Serrada, R., Nicolás Peragón, J.L. (Eds.), Producción y manejo de semillas y plantas forestales. Organismo Autónomo Parques Nacionales, Madrid, pp. 80–90.

Pawlowski, T., Szczotka, Z., 1997. Qualitative changes in protein content during cold and warm stratification of Norway maple (*Acer platanoides* L.) seeds. Seed Sci. Res. 7, 385–390. https://doi.org/10.1017/S0960258500003792.

Piotto, B., Di Noi, A., 2003. Seed Propagation of Mediterranean Trees and Shrubs. APAT, Roma.

Piovesan, G., Di Filippo, A., Alessandrini, A., Biondi, F., Schirone, B., 2005. Structure, dynamics and dendroecology of an old-growth *Fagus* forest in the Apennines. J. Veg. Sci. 16, 13–28. https://doi.org/10.1111/j.1654-1103.2005.tb02334.x.

Pukacki, P.M., Juszczyk, K., 2015. Desiccation sensitivity and cryopreservation of the embryonic axes of the seeds of two *Acer* species. Trees 29, 385–396. https://doi.org/10.1007/s00468-014-1118-7.

Rusterholz, H.-P., Studer, M., Zwahlen, V., Baur, B., 2020. Plant-mycorrhiza association in urban forests: effects of the degree of urbanization and forest size on the performance of sycamore (*Acer pseudoplatanus*) saplings. Urban For Urban Green. 56, 126872. https://doi.org/10.1016/j.ufug.2020.126872.

Shouman, S., Mason, N., Kichey, T., Closset-Kopp, D., Heberling, J.M., Kobeissi, A., Decocq, G., 2017. Functional shift of sycamore maple (*Acer pseudoplatanus*) towards greater plasticity and shade tolerance in its invasive range. Perspectives in Plant Ecology. Evolut. Systemat. 29, 30–40. https://doi.org/10.1016/j.ppees.2017.11.001.

Sinclair, W.A., Lyon, H.H., 2005. Diseases of Trees and Shrubs, 2. ed. Cornell Univ. Press, Ithaca, NY.

Spethmann, W., 2007. Increase of rooting success and further shoot growth by long cuttings of woody plants. Propag. Ornam. Plants 7 (3), 160–166.

Stejskalová, J., Kupka, I., Miltner, S., 2015. Effect of gibberellic acid on germination capacity and emergence rate of Sycamore maple (*Acer pseudoplatanus* L.) seeds. J. For. Sci. 61, 325–331. https://doi.org/10.17221/22/2015-JFS.

Stejskalová, J., Kupka, I., Nováková, O., 2014. Influence of sycamore seed stratification length on their germination capacity. J. For. Sci. 60, 212–217. https://jfs.agriculturejournals.cz/artkey/jfs-201405-0005_influence-of-sycamore-seed-stratification-length-on-their-germination-capacity.php.

Strimbu, B.M., Nicolescu, V.-N., 2023. Coppice management for young sycamore maple (*Acer pseudoplatanus* L.). Forests 14, 297. https://doi.org/10.3390/f14020297.

Tomov, V., 2007. Rooting of Norway maple (*Acer platanoides* L.) cuttings. Forest. Ideas 23, 57–64.

Turner, A.P., Dickinson, N.M., 1993. Survival of *Acer pseudoplatanus* L. (sycamore) seedlings on metalliferous soils. New Phytol. 123, 509–521. https://doi.org/10.1111/j.1469-8137.1993.tb03763.x.

Tylkowski, T., 1989. Short-term storage of after-ripened seeds of *Acer platanoides* L. and *A. pseudoplatanus* L. Arboret. Konic. 34, 135–141.

Vacek, S., Vacek, Z., Kalousková, I., Cukor, J., Bílek, L., Moser, W.K., Bulušek, D., Podrázský, V., Řeháček, D., 2017. Sycamore maple (*Acer pseudoplatanus* L.) stands on former agricultural land in the Sudetes – evaluation of ecological value and production potential. Dendrobiology 79, 61–76. https://doi.org/10.12657/denbio.079.006.

Whittet, R., López, G., Rosique-Esplugas, C., 2021. Mid-rotation variation in growth, form and phenology of sycamore (*Acer pseudoplatanus* L.) provenances in field trials in England. Forestry 94, 704–713. https://doi.org/10.1093/forestry/cpab012.

Yücesan, Z., Bayram, D., 2021. Effects of pretreatment, sowing time, sowing environment and climate factors on germination in *Acer pseudoplatanus*. L. Šumar. List 145, 557–566. https://doi.org/10.31298/sl.145.11-12.5.

Carpinus L. (*Carpinus orientalis* Mill., *Carpinus betulus* L.)

Anastazija Dimitrova[1], Halil Barõş Özel[2] and Hakan Sevik[3]
[1]Ss. Cyril and Methodius University in Skopje, Hans Em Faculty of Forest Sciences, Landscape Architecture and Environmental Engineering, Skopje, North Macedonia; [2]Faculty of Forestry, Department of Silviculture, University of Bartõn, Bartõn, Türkiye; [3]Department of Environmental Sciences, Faculty of Engineering and Architecture, University of Kastamonu, Kastamonu, Türkiye

Propagation

Seed propagation

The fruits of *Carpinus orientalis* and *Carpinus betulus* ripen between September and January and are dispersed by wind (short distance) or birds (long distance) from fall to spring (Pijut, 2008). Seed production starts when trees are 10–12 years old, with abundant seed years occurring every 1–2 years (Gültekin, 2008). Fruits can be harvested when green-brown as soon as the wings turn yellow and are soft and flexible, or when fully ripened and brown (Pijut, 2008; Božič et al., 2021). If broadcasted, the fruit bracts do not need to be removed (Pijut, 2008). Bract removal can be done manually by rubbing the fruits or placing the fruits inside bags and beating them. If large quantities of seeds are needed, the bracts can be removed by dewinging machines. Debris can be removed by screening and fanning (Pijut, 2008; Piotto and Di Noi, 2003).

Harvesting immature seeds from green fruits of both species prevents the formation of a hard seed coat and these seeds germinate in the first winter after collection, whereas fully mature seeds germinate in the second spring after dispersal (Suszka et al., 2022). *Carpinus* species show physical and physiological dormancy (Güney et al., 2015). Fresh seeds require 90 days of chilling (2–4°C) to break dormancy. In addition, fresh *Carpinus* seeds have a relatively high moisture content (around 25%) (Suszka et al., 2022) that favors high seed respiration rates and consequently necessitates careful seed processing before storage or use (Piotto and Di Noi, 2003). If green seeds are planned to be used soon, they need 3–4 months of cold stratification or a combination of warm stratification (3–4 weeks at 20°C) followed by cold stratification (12–15 weeks at 3–5°C) before spring sowing (Pijut, 2008; Piotto and Di Noi, 2003). Alternatively, seeds can be sown directly in the fall without pretreatments and expect normal fall and winter conditions to promote germination the following spring. To prevent hard seed coat formation in green seeds collected for storage, avoid full drying and check for well-developed embryos before sowing (Pijut, 2008).

Other dormancy-breaking methods include treatment with gibberellic acid (GA) combined with cold stratification (i.e., GA with 25 or 100 ppm concentration, followed by cold stratification at 4°C for 6, 12 or 18 weeks); or exposing the seeds to alternating cycles of 8 h with light at 25°C and 16 h in the dark at 20°C, or even more intensive temperature fluctuations such as 25°C/10°C (Pijut, 2008; Piotto and Di Noi, 2003).

In *C. betulus,* a combination of 2 months of warm stratification followed by 4-month cold stratification is effective for increasing the germination; however, it can also be replaced by GA treatment instead of the warm stratification and 3–4 months cold stratification period (Pipinis et al., 2012). In *C. orientalis,* warm stratification does not seem to be as effective; instead, a combination of GA treatment (500, 1000 and 2000 ppm) followed by 4 months of cold stratification has shown to be the best combination (Pipinis et al., 2012). Neither immersion of *C. orientalis* seeds in GA (500 ppm) at 25°C for 24 h or mechanical or chemical seed coat abrasive treatments (sandpaper and H_2SO_4) improved germination (Tsitsoni et al., 2013). If dry seeds with hardened seed coats are harvested, they should be spread in thin layers in a cool, aerated space for drying, with occasional stirring. Stratified seeds can be stored for up to 2 years. Seeds with 10% moisture content can be stored in closed containers at 0–2°C for extended periods. Partially dried seeds with 8%–10% moisture content can be stored in sealed containers at −3°C for up to 5 years, while stored at 3°C they remain viable up to 14–18 months (Pijut, 2008; Piotto and Di Noi, 2003). Seed characteristics of both *Carpinus* species are shown in Table 11.3.

Vegetative propagation

C. orientalis can be vegetatively propagated from cuttings and by grafting (Piotto and Di Noi, 2003), although it is challenging with low rooting success caused by several physiological and genetic factors (Çalıkoğlu, 2017). Given that rooting success is low, tissue culture can be an option for critical purposes such as species conservation.

C. betulus can be propagated from cuttings or by grafting, with success depending on the cultivar (Pijut, 2008; Piotto and Di Noi, 2003). To maximize rooting, cuttings should be taken within the first 8 weeks after budbreak (Gültekin, 2008). The use of 2% indole-3-butyric acid promotes rooting, after which the cuttings require a dormancy period by storing them at 0°C during winter (Pijut, 2008). Stock plant etiolation and stem banding can also improve the rooting, with etiolation being particularly effective for *C. betulus* (Božič et al., 2021; Maynard and Bassuk, 1992).

Table 11.3 Characteristics of seed lots of *Carpinus orientalis* and *Carpinus betulus.*

	Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
Carpinus orientalis	Unknown	Unknown	70,000–85,000 (77,500)	65–85 (75)
Carpinus betulus	50	97	14,000–28,660 (21,330)	30–85 (58)

Mean values in brackets.
Pijut (2008); Piotto and Di Noi (2003); Türker et al. (2009).

Seedling cultivation

Container plants

Sowing is conducted in late winter or early spring using cold-wet stratified seeds. Seeds are sown at a depth of 1–2 mm in 45- or 24-cell multicavity containers, with three to five seeds per cell. Seeds can be covered with perlite and short, light irrigations should be applied several times a day until seeds germinate. The growing medium can be a mixture of 80% peat and 20% pumice (3–5 mm) or vermiculite, and the optimum pH varies between 5.0 and 6.5. A slow-release fertilizer with a supply of sulfur can be mixed with the growing medium at a rate of 0.5 kg/m^3.

Seedlings are sensitive during the growth of their secondary leaves and some mortality may occur due to drought, drying of the root collar, or fungal diseases. *Carpinus* seedlings should be cultivated under 35%–50% shade, with the shade cloth removed after September. Daily watering is essential, with saturation of the growing medium provided through a sprinkler system during the cooler parts of the day. Fertilization can also be applied via irrigation (i.e., fertigation) and then only sulfur should be added to the growing medium.

1 + 0 seedlings grown in containers, particularly those that favor air pruning of lateral roots, can be used for plantations. For specific forestation activities (roadside afforestation, landscaping works, etc.), 1 + 1 and 1 + 2 seedlings can be used. In some countries such as Türkiye, 1 + 1 plants can be cultivated in 16 × 28 cm polybags and 1 + 2 plants can be cultivated in 18 × 28 cm polybags (Gültekin, 2014).

Bare root plants

Seeds can be sown directly immediately after collection in the fall or in early spring. If spring sowing is preferred, seeds need to be stratified (see Section 1.1). Seeds should be soaked in water for 1–2 days before sowing and covered after sowing to prevent rapid drying. Water regularly during dry periods until germination occurs. Seeds are sown manually on 20–25 cm high seedbeds (120 cm wide) in five or seven rows, at a depth of 4–5 mm. The seeds should be covered with a 0.6–1.3 cm layer of a 1:1 mixture of organic material and stream silt (maximum 1.5 times the thickness of the seed). *Carpinus* species grow well in slightly acidic or neutral sandy loam soils containing 3–5% organic matter (Gültekin, 2014; Maynard and Bassuk, 1992). The seedbeds need to be continuously moist, rich loamy soil that is protected from extreme atmospheric changes. Seeding rates vary between 323 and 43 1/m^2. If fall-sown beds are used, they should be mulched with burlap, pine straw, or another material until the last spring frost. The soil surface should be kept moist until after germination and the beds should be lightly shaded during the first year (Pijut, 2008).

Watering frequency depends on the weather and growing conditions. Generally, water every 2–3 days in spring and early autumn, every 1–2 days in summer, and every 3–6 days in late autumn. The optimal seedling spacing is 10 cm (Atar, 2021). If the density of emerged seedlings is high, thin them in May (Gültekin, 2014). Fertilization rate depends on soil analysis, with the first application beginning once seedling roots reach 10–15 cm depth.

Bare root stock (1–0) can be planted when they reach >30 cm height and a root collar diameter >0.4 cm at lifting (Gültekin, 2014). Herbicides and synthetic sheet mulches such as black polythene sheets 125 m thick have been beneficial for weed control (Pijut, 2008).

Field establishment

Carpinus species tolerate a wide range of soil and light conditions. However, they grow best in adequate sun exposure, although shading might be required in harsh sites to protect the plants from sunburn. Rich, moist, and well drained soils are best for planting *Carpinus* species (Pijut, 2008). No special planting practices are needed for *Carpinus* species other than used in deciduous broadleaf species from wet temperate climates.

Chapter References

Atar, F., 2021. Effects of altitude on some seedling quality characteristics of *Carpinus betulus* L. (Common hornbeam) and *Carpinus orientalis* Mill. (Oriental hornbeam). J. Fores. Fac. 22 (2), 257–265.

Božič, G., Westergren, M., Heino, K., Lanšćak, M., Nagy, L., Novčić, Z., Rukavina, S., Schüler, S., Stojnić, S., Železnik, P., 2021. Guidelines for the Production of Planting Material for Restoration of Riparian Forests. Slovenian Forestry Institute, Silva Slovenica Publishing Centre.

Çalıkoğlu, E., 2017. Germination Characteristics of Eastern Hornbeam (*Carpinus orientalis* Mill.) Seed. Master Thesis. Karadeniz Technical University, Institute of Science and Technology, Department of Forestry Engineering, p. 77.

Gültekin, H.C., 2008. Some sexual characteristics of angiosperm trees and shrubs. Forest Hunt. J., July-August (4), 42–47 (in Turkish).

Gültekin, H.C., 2014. *Carpinus* L. (Hornbeam). Seedling production techniques of important forest trees. Ministry of Forestry and Water Affairs, General Directorate of Forestry, Poplar and Fast Growing Forest Trees Research Institute. Various Publications Series No: 26, pp. 188–192. Izmir.

Güney, D., Atar, F., Atar, E., Turna, İ., Kulaç, S., 2015. The effect of pre-treatments and seed collection time on the germination characteristics of common hornbeam (*Carpinus betulus*) seeds in the Eastern Black Sea Region, Turkey. Seed Sci. Technol. 43, 1–9.

Maynard, B.K., Bassuk, N.L., 1992. Stock plant etiolation, shading, and banding effects on cutting propagation of *Carpinus betulus*. J. Am. Soc. Hortic. Sci. 117, 740-740.

Pijut, P.M., 2008. Carpinus. In: Bonner, F.T., Karrfalt, R.P. (Eds.), Woody Plant Seed Manual. US Department of Agriculture Forest Service, vol 727. Agriculture Handbook, Washington, DC, pp. 328–332.

Piotto, B., Di Noi, A., 2003. Seed propagation of Mediterranean trees and shrubs. Dendrobiology 47, 109–111.

Pipinis, E., Milios, E., Kiamos, N., Mavrokordopoulou, O., Simiris, P., 2012. Effect of stratification and pre-treatment with gibberellic acid on seed germination of two *Carpinus* species. Seed Sci. Technol. 40, 21–31.

Suszka, J., Bujarska-Borkowska, B., Tylkowski, T., Wawrzyniak, M.K., 2022. Effects of temperature, moisture content and storage on dormancy release and germination of European hornbeam (*Carpinus betulus* L.) seeds. Forestry 95 (3), 428–436. https://doi.org/10.1093/forestry/cpab053.

Tsitsoni, T., Tsakaldimi, M., Tsouri, C., 2013. Seed treatments to break dormancy and stimulate germination in *Cercis siliquastrum* L. and *Carpinus orientalis* Mill. Afr. J. Agric. Res. 8, 4501–4505.

Türker, A.H., Gülbaba, A.G., Öztürk, N., Taşdelen, A., Gültekin, H.C., 2009. Seed production of some leave species that can be used in afforestation in the eastern Mediterranean region. Eastern Mediterranean Forestry Research Directorate. Project No:, 20. 1203/2003-2006.

Castanea sativa Mill.

Alberto Maltoni[1], Beatriz Cuenca[2], Milan Mataruga[3] and Barbara Mariotti[1]
[1]Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy; [2]Vivero de Maceda, Empresa de Transformación Agraria S.A., S.M.E., M.P. (TRAGSA), Ourense, Spain; [3]Faculty of Forestry, University of Banja Luka, Republic of Srpska, Bosnia and Herzegovina

Propagation

Seed propagation

Castanea sativa flowers from May to early July according to climate and latitude, and the flowers remain highly receptive for 3–4 weeks. Pollination involves both wind and insect vectors, especially bees, and it is influenced by climatic conditions. In dry conditions, wind disperses pollen very effectively, while in wet conditions, pollen viscosity limits dispersal (Cuenca Valera and Majada Guijo, 2012). Strong and persistent precipitations can limit pollination.

Seeds (nuts) are enclosed in a prickly, hard involucre (bur). Most seeds fall to the ground free from the burs and can be collected manually, using special vacuum cleaners, or by self-propelled harvesters from late September to early November. Manual harvesting can be done directly with rakes and gloves or with nets placed under the trees during thinning or before to catch spontaneous falls. Nets, which can be laid on the ground or suspended, speed up collection and cleaning but involve installation costs and can be damaged by wildlife (Cuenca Valera and Majada Guijo, 2012). Sweeping machines or vacuum cleaners are attached to a tractor, are self-propelled, or in some cases portable. Their use will depend on the characteristics of the terrain and the plantation (access, slope, size of the plot, arrangement of the trees, soil cleanliness) (Cuenca Valera and Majada Guijo, 2012).

Damaged seeds (e.g., those affected by insects) can be removed using densitometric methods in water. Cuenca Valera and Majada Guijo (2012) suggested the following procedure to prevent insect larval development and enhance nut preservation: a) immersion in hot water (48–50°C) for 30–45 min followed by 8–12 h in cold water to restore initial thermal conditions and avoid overheating; b) immersion in cold water for 5–8 days (nuts to water volume ratio, 2:3) to create low-oxygen conditions. This eliminates aerobic microorganisms causing molds and rots, and promotes anaerobic microorganisms that induce slight lactic fermentation, reducing the sugar content and thus the food source for rot fungi. The presence of characteristic CO_2 bubbles on the water surface indicates that the treatment is complete. Water renewal, usually partial, depends on both the quantity of nuts and the renewal frequency (more frequent renewals use smaller amounts of water).

The seeds are recalcitrant (Pence, 1992) and to preserve them they are typically stratified in moist sand or peat moss at 1–4°C for 2–3 months before sowing. Seeds cannot be stored for long periods even under refrigeration, as they will lose their germination

ability. *C. sativa* nuts can also be stored for a few months by sealing them in a polyethylene, 50-μm thick bag at 4°C. Alternatively, seeds can be stored at 50%–55% relative humidity at 0–3°C overwinter (Cuenca Valera and Majada Guijo, 2012). When stored in dry conditions, germination capacity rapidly declines. In addition, when stored at temperatures below 0°C, nuts freeze and become unusable (Stilinović, 1987).

A high germination rate occurs after cooling in humid conditions to break seed dormancy (Mencarelli, 2001; Fernandes et al., 2022). To achieve more synchronized germination, nuts are stratified in moist sand or peat moss at 1–2°C for at least 2 months, but no more than 4–5 months. At the end of stratification, higher temperatures promote germination (Beccaro et al., 2019a).

Germination tests, following ISTA standards, involve placing seeds on filter paper under high humidity conditions at a night-day temperature of 20–30°C. The first count is performed after 7 days, with the final count after 21 days. The quantitative characteristics of *C. sativa* seeds are shown in Table 11.4.

Vegetative propagation

Traditional *C. sativa* orchards consist of grafted plants, as grafting is the most effective method for vegetative propagation of selected fruit varieties. Other propagation methods are less suitable for large-scale production of trees intended for fruit production, due to cost, low effectiveness, or both, as mature trees (those bearing fruits) are usually unable to root. Techniques such as layering and rooting cuttings (both soft- and hardwood), and micropropagation are used to produce self-rooted rootstocks or self-rooted plants for orchards.

Grafting is done in different seasons according to the technique used: in winter, whip graft; in winter–spring, triangle (inlay) graft, splice (slice) graft; in early spring, lateral graft and Cadillac; in spring, under bark graft and crown graft; in spring-summer, T-budding; in summer, chip budding, and semisoft scion graft (Bounous, 2014). The scions used for winter grafting are collected from healthy and well-lignified one-year-old branches. If the mother plants are old, severe pruning can induce new vigorous shoots suitable for scions. The scions are generally cut when the trees are completely dormant in winter and are stored in closed polyethylene bags at 1–2°C. The scions

Table 11.4 Characteristics of seed lots of *Castanea sativa*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
100 (80–100)	100 (100)	50–257 (120)	60–90 (79)

Mean values in brackets.
Gradi (1980), Piotto (1992), Berrocal et al. (1998), Catalán (1991), Ribeiro et al. (2001), Navarro Cerrillo and Gálvez (2001), Louro and Pinto (2011), Cuenca Valera and Majada Guijo (2012), Moraldi (2024).

used for summer grafts are normally cut from the mother trees right before the moment of grafting. Graft wounds must be protected to prevent tissue dehydration and pathogen attack. Cold (for field use) or warm (for nursery use) commercially available dressings are composed of fats, natural and synthetic resins, or vegetable oils. To prevent *Cryphonectria parasitica* infection, grafts can be treated with formulations containing biocontrol agents, copper, or benzimidazole fungicides (Beccaro et al., 2019a). Hot water treatment has been tested to control pest and diseases in propagation processes and it is very effective for the disinfection of scions against *Dryocosmus kuriphilus* (Ciordia et al., 2020).

Vegetative propagation uses the low layering or stooling method (Cuenca Valera and Majada Guijo, 2012; Beccaro et al., 2019a; Serdar et al., 2019). Briefly, the plant is cut at ground level and new shoots formed in spring are covered with soil to promote rooting. The plant is cut annually, and the best shoots are selected while removing the rest. Selected shoots are defoliated at the base between mid-May and mid-June, lightly scraped, and treated with a paste containing rooting hormones (mainly auxins). Traditionally, pastes for rooting have been made by dissolving indolebutyric acid (IBA) in petroleum jelly, with concentrations ranging from 4%–8% based on shoot lignification. Shoots are then banded with a wire ring below the hormone-treated area to enhance photosynthate accumulation and promote root formation. To improve rooting, select young shoots and use loose, moderately acidic, and well-drained soils. Rooted sprouts are detached form mother plants and lifted from mid-November, after leaf drop. These plants often have poorly balanced root systems, with peripheral shoots showing base curvature, which can be corrected by cultivating the plants for an additional season in the nursery (Cuenca Valera and Majada Guijo, 2012).

Cuttings are an alternative vegetative propagation method that requires less land for the deployment of mother plants, but it needs more specific and engineered facilities, such as air-humidity control systems. Semiherbaceous stems are collected in June from mother plants grown in big pots under shade, and they are cut into one to three bud sections. The base of these sections is dipped in a hormonal solution (1000 ppm of IBA) for some minutes and placed in trays filled with a highly draining substrate to root inside rooting tunnels in the greenhouse. A fog system in the summer ensures high air humidity to prevent dehydration of cuttings during the rooting process. Rooted cuttings are transplanted to pots the next spring to resume growth. These plants develop a higher-quality rooting system compared with those produced by layering. In addition, as they are potted, they have a longer period for commercialization (Beccaro et al., 2019a; Serdar et al., 2019).

Micropropagation is another vegetative propagation method that allows quick propagation starting with very little material, although it is limited to technologically advanced nurseries (Beccaro et al., 2019a). Successful results require healthy, juvenile shoots (Vieitez, 1992), but the plants produced by this method are free from viruses and diseases, making it an ideal system for moving plants around the world.

The micropropagation protocol for *C. sativa* through axillary shoots can be divided into four stages: establishment, shoot proliferation, shoot rooting, and hardening. Various media have been used, but the most adopted are the GD mineral medium (Gresshoff and Doy, 1972) and the MS medium (Murashige and Skoog, 1962). Apical necrosis and chlorosis can be avoided by using half-strength nitrate in the culture media (Vieitez et al., 1986).

Seedling cultivation

To establish forest stands, sexually propagated trees are used in compliance with legal requirements (Directive 1999/105/CE). *Castanea sativa* seeds are typically collected from coppice stands or orchards (Ciordia et al., 2012), and rarely from wild populations. However, it must be taken into account that orchard stands exhibit less genetic differentiation compared with coppices and old high forest stands (Seabra et al., 2001). For fruit orchards, nursery production includes seedlings used as rootstocks, grafted plants, and vegetative copies of selected material (e.g., clonal rootstocks, grafted plants, or other asexually propagated plants of selected cultivars). Clonal rootstocks are produced by rooting shoots from clones selected for their tolerance to root diseases, dwarfing effects, and grafting compatibility.

There is scant literature on the quality of *C. sativa* reproductive forest material. This is because the species rarely loses its ability to produce new shoots from stumps, which ensures its persistence and regeneration in forests. Moreover, many *C. sativa* plantations have been established for creating or restoring orchards, where grafted nursery material is more commonly used. This has led to a lack of an approach for evaluating the material that aligns with the target plant concept or assesses the field performance of the material (Clark et al., 2023). Moreover, planting *C. sativa* is not currently a priority for species maintenance as it is for *Castanea dentata* in the US. Since *C. sativa* is widely distributed across Europe, the main objective is growing forest reproductive material to restore stool density to suitable levels in coppice stands (Manetti et al., 2022).

The main problem in establishing nursery seedlings of *C. sativa* is the availability of healthy nursery material free from the pests *Phytophthora cambivora* and *Cryphonectria parasitica* that are the two most influential factors hindering the success of reforestation projects. For this reason, the breeding and production of planting material is mostly directed toward the creation of tolerant genotypes, both for rootstocks and for cultivars (which are mainly propagated by grafting).

Container plants

High germination rates occur after cooling in humid conditions to remove seed dormancy. To synchronize germination, the nuts should be stratified in moist sand or peat moss at 1–2°C for at least 2 months (maximum 4–5 months). At the end of the

stratification, increased temperatures promote germination and both the primordial root (radicle and hypocotyl), and the young shoot (epicotyl) emerge from the torch at the top of the seed. After removing the root apex, the pregerminated nuts are sown (flat side down) in raised beds, trays, or pots. The soil should be soft and well drained. The sowing depth is generally no more than 3–5 cm. In *C. sativa*, as in other recalcitrant, large, nutrient-rich seeds; the weight of the seed affects seedling development (Tumpa et al., 2021). Nursery cultivation can begin in March–April, though if the cultivation is done in a greenhouse, sowing can start in February. The latter is useful to obtain larger 1-year-old rootstock.

The volume of the container can range between 400 and 1000 mL to grow 1- or 2-year-old seedlings. The growing medium can be 70%–80% peat (or coir) mixed with 20%–30% perlite to improve aeration. Compost can replace peat up to 50%, obtaining plants of the same size and quality. Pine bark is not recommended due to the occurrence of *Fusarium circinatum* (Beccaro et al., 2019a).

Cultivation density is crucial for promoting healthy growth and producing high quality seedlings. If it is too high, overlapping leaves can result in shading and reduce water availability in each container cell. An optimal density is 100 plants/m^2 (Beccaro et al., 2019a) and density should not exceed 250 plants/m^2 (Cuenca Valera and Majada Guijo, 2012). Using hydrogels mixed with the substrate can help maintain water levels between irrigations (Cuenca Valera and Majada Guijo, 2012).

Although slow-release fertilizers mixed in the initial substrate can be used, fertilization of the container plant is usually carried out through fertigation systems; as an example, NPK (14:7:14) mineral fertilizer can be applied once a month at a rate of 23.3 mg/plant of N in each application (Fuertes-Mendizábal et al., 2021). Initial fertilization should be high in nitrogen to support plant growth and phosphorus to establish the root system. During the hardening phase, we recommend reducing nitrogen and increasing potassium to enhance resistance to water and heat stress. High nitrogen fertilization should be avoided to prevent strong leaf growth, which can expand the canopy and make irrigation uniformity difficult (Cuenca Valera and Majada Guijo, 2012). Plant growth-promoting (PGP) biostimulants can complement fertilization since these microorganisms provide multiple benefits to plant growth; in addition, they can significantly improve plant tolerance to water deficit (Fuertes-Mendizábal et al., 2021).

Inoculation with ectomycorrhizal fungi has been proven to improve water uptake and mineral nutrition and to prevent fungal root diseases (Crawford, 1995). Inoculation with *Boletus* spp. and *Morchella conica* can improve seedling quality (Gonzalez et al., 2010); valuable *Tuber* spp. have also been tested with some success (Crawford, 1995).

For container-grown plants, observed and recommended values of morphological attributes are reported in Table 11.5. A straight stem with no branches can be recommended for reforestation or forest restoration projects (Fig. 11.3).

Table 11.5 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Castanea sativa*.

	Container seedlings		Bare root seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	33–77	(40) 50–70	10–86	40–60
Root collar diameter (cm)	0.96–1.15	>0.6	0.52–1.34	>0.6
Shoot dry weight (g)	1.4–9.3		3.1–28.5	
Root dry weight (g)	2.2–8.7		14.7–22.2	
Plant dry weight (g)			35.1–58	
Shoot/root dry weight ratio			1.1–1.8	

Hipps et al. (1997), Çiçek and Tilki (2007), Álvarez-Lafuente et al. (2018), Tumpa et al. (2021), Fuertes-Mendizábal et al. (2021).



Figure 11.3 One-year old seedlings of *Castanea sativa*. *(Barbara Mariotti.)*

Bare root plants

The same recommendations as for container cultivation apply to seeds. The soil should be soft and well drained, with a seeding depth generally not exceeding 3–5 cm, and covered with a sterile substrate, such as peat, to prevent dehydration and weed growth (Cuenca Valera and Majada Guijo, 2012). The soil should be compacted with a roller to

eliminate air pockets that could hinder germination. It is advisable to protect the sowing from predators using repellents or nets, which can also provide shade. Depending on climatic conditions and the seed stratification period, germination will typically be completed within 4–6 weeks (Cuenca Valera and Majada Guijo, 2012). For producing 1-year-old seed rootstock, sowing in raised beds is recommended, with row spacing typically 30–40 cm—at least 20 cm (Cuenca Valera and Majada Guijo, 2012) - and rows 80–100 cm apart (Beccaro et al., 2019a). Irrigation, fertilization, and control of weeds and pathogens are crucial. Hoeing or mechanical weed control methods are used, although plastic mulch can also be employed; however, this is not considered an environmentally sustainable practice (Beccaro et al., 2019a). Root pruning can improve root system structure, making it shorter and more fibrous, which enhances plant lifting and survival (Cuenca Valera and Majada Guijo, 2012). Root pruning also favors the ratio between the shoots and roots by slowing shoot growth. Lifting is carried out in winter, and planting should be done during the same period before the growing season starts. Small plants can be transplanted and cultivated for an additional year in the nursery.

Observed and recommended values of morphological attributes of bare root plants are reported in Table 11.5. Plants with straight stems and no branches can be recommended for reforestation or forest restoration projects where timber is the main objective.

Field establishment

C. sativa plantings for silvicultural purposes are established to create new high-forest stands (Balandier and Dupraz, 1999; Bourgeois et al., 2004). Artificial regeneration is not needed when stumps are present, as this species has a strong and long-lasting resprouting capacity. In such cases, seedlings grown from seeds cannot survive the competition. In Italy, *C. sativa* has been introduced into both pure and mixed productive plantations to provide high-value timber (Buresti-Lates and Mori, 2009), with recommendations to use protective measures against browsing. Similar plantings have been implemented in other EU countries with funding from European Community Support Frameworks (Bourgeois et al., 1991; Patrício, 2006; Patrício and Nunes, 2017). The afforestation rate of *Castanea sativa* in Portugal, northern Spain, and France was estimated at 4500 ha/year (Alvarez-Alvarez, 2004). In France, for timber production, a recommended planting density is up to 1200 plants/ha (IDF, 1990; Patrício, 2006; Benedetti-Ruiz et al., 2023).

C. sativa trees thrive in deep, cool, well-drained, and limestone-free soils. Therefore, conducting a soil analysis is recommended. Ideal soils should be light to medium-textured to prevent the risk of fungal infections like *Phytophthora* sp., which causes ink disease. Soils should be at least 40 cm deep and moderately acidic, with a pH no lower than 4.2 (Cuenca Valera and Majada Guijo, 2012). While *C. sativa* can tolerate

frost, they are sensitive to late spring frosts. The ideal water supply is at least 600 mm during the vegetative period.

It is possible to establish plantations by direct seeding, sowing at a depth of 4–5 cm, typically with two to three seeds per hole. Spring sowing is recommended with stratified seeds to minimize the time the seeds remain in the soil without germinating, thus reducing damage from rodents and birds. Seed shelter, a new tool to protect seeds from rodents (Castro et al., 2015), can promote seeding success.

For timber plantations, one seedling per hole is preferred as it is more robust than two seedlings. *C. sativa* is challenging to establish, so taking great care and monitoring the seedlings during their initial 2–3 years is essential. Fertilizing the planting hole, ensuring adequate water during the first summers, and shielding the plants from herbivores and sunburn with 1.20 m protective tubes are recommended (Cuenca Valera and Majada Guijo, 2012). The optimal planting time is late autumn or early winter, while the plant is dormant, and avoiding planting on excessively windy or frosty days is crucial.

The choice of planting density depends on whether the objective is to produce timber in monospecific or mixed plantations, timber and fruit, or only fruit. Planting density can be high, medium or low, with or without intercropping. Low-density plantations require more intensive early intervention to develop straight trunks and balanced crowns. Timber plantations generally have higher densities, while fruit plantations are more widely spaced to promote flowering and fruit production. In timber plantations, an ideal density is one that yields 180–250 high quality stems by the end of the rotation period (Cuenca Valera and Majada Guijo, 2012).

For timber production, using average quality plants with minimal pruning typically requires a density of 950–1100 plants/ha, with spacings of 3.5 × 3 m, 4 × 2.5 m, or 3 × 3 m (Cuenca Valera and Majada Guijo, 2012). If high-quality plants are used, density can be lower, but pruning needs to be more frequent. At densities of 400–830 plants/ha, the recommended spacing is 5 × 5 m, 4 × 4 m, or 4 × 3 m (Cuenca Valera and Majada Guijo, 2012). Currently, the 4 × 4 m spacing is common, which reduces planting costs though it requires more intensive pruning. Bourgeois et al. (2004) suggested a minimum row distance of 4 m, with planting densities of 625–1200 plants/ha on agricultural land and 1100 to 1430 plants/ha on forestland. For fruit production, new plantings densities can be as low as 100 trees/ha (10 × 10 m) or much more (250–300 trees/ha) in intensive management. Agroforestry schemes can also be done in lines with the tress spaced 3 m, and the rows separated by 10–12 m, allowing grazing or crop production between the rows (Cuenca Valera and Majada Guijo, 2012).

Several ecological factors can hinder planting success, including early frosts, summer droughts during the first 2–3 years, high soil lime, and temporary waterlogging. In addition, young trees are also highly sensitive to weed competition. Initially, herbicides should be used to control weeds, as mechanical methods can damage roots and potentially spread diseases like ink disease in areas with high soil moisture (Cuenca Valera and

Majada Guijo, 2012). Disc harrows, cultivators, or rotovators are not recommended due to the shallow root system of *C. sativa* trees. Instead, control weeds with manual brush cutters or herbicides in early summer, taking care to damaging the tree if using systemic products. Mulching with cut grass or brush around the base of the trunk helps suppress weeds and maintain soil moisture. In addition to grass, it is essential to control woody shrubs in the early years, possibly using a chain brush cutter. In some cases, such as with brambles, ferns, or broom, competition is minimal, and they can be beneficial by keeping the soil cool, providing shade, and protecting against large game damage (Cuenca Valera and Majada Guijo, 2012). Avoid carrying out these operations during very hot periods to prevent trunk damage and burn issues (Cuenca Valera and Majada Guijo, 2012). Timber plantations are not commonly fertilized, although leaving leaf litter is beneficial for organic matter contribution. Fertilization is crucial in agricultural soils with nutrient deficiencies, however, particularly in fruit production plantations. Recommendations can be found in Cuenca Valera and Majada Guijo (2012) and Beccaro et al. (2019b).

Chapter References

Alvarez-Alvarez, P., 2004. Viveros forestales y uso de planta en repoblación en Galicia [Forest nurseries and plant use in afforestations in Galicia]. PhD Thesis. Universidad de Santiago de Compostela, Santiago de Compostela, Spain, p. 261.

Álvarez-Lafuente, A., Benito-Matías, L.F., Peñuelas-Rubira, J.L., Suz, L.M., 2018. Multi-cropping edible truffles and sweet chestnuts: production of high-quality *Castanea sativa* seedlings inoculated with *Tuber aestivum*, its ecotype *T. uncinatum, T. brumale,* and *T. macrosporum*. Mycorrhiza 28 (1), 29–38. https://doi.org/10.1007/s00572-017-0805-9.

Balandier, P., Dupraz, C., 1999. Growth of widely spaced trees, a case study from young agroforestry plantations in France. Agrofor. Syst. 43, 151–167. https://doi.org/10.1023/A:1026480028915.

Beccaro, G.L., Bounous, G., Cuenca, B., Bounous, M., Warmund, M., Xiong, H., Zhang, L., Zou, F., Serdar, U., Akyü, B., Mellano, M.G., Lourenço, C.R., 2019a. Nursery techniques. In: Beccaro, et al. (Eds.), The Chestnut Handbook: Crop & Forest Management. CRC Press, pp. 119–154.

Beccaro, G., Alma, A., Bounous, G., Gomes-Laranjo, J., 2019b. European chestnut traditional and high-density orchards. In: Beccaro, et al. (Eds.), The Chestnut Handbook: Crop & Forest Management. CRC Press, pp. 203–254.

Benedetti-Ruiz, S., Loewe-Munoz, V., Del Río, R., Delard, C., Barrales, L., Balzarini, M., 2023. Effect of thinning on growth and shape of *Castanea sativa* adult tree plantations for timber production in Chile. For. Ecol. Manag. 530, 120762. https://doi.org/10.1016/j.foreco.2022.120762.

Berrocal, M., Gallardo, J.F., Cardenoso, J.M., 1998. El Castaño. Ed. Mundi-Prensa, Madrid.

Bounous, G., 2014. Il castagno: Risorsa multifunzionale in Italia e nel mondo. Edagricole, Bologna, Italy, p. 420.

Bourgeois, C., Evans, J., Rollinson, T.J.D., 1991. Le chataignier en Europe. Foret Entreprise 76 (4), 25–42.

Bourgeois, C., Sevrin, E., Lemaire, J., 2004. Le Chataignier Un Arbre, Un Bois, second ed. Institute pour le Developpement Forestiere, Paris.

Buresti-Lates, E., Mori, P., 2009. L'indice di qualità di piantagioni pure. Sherwood 157, 17–21.

Castro, J., Leverkus, A.B., Fuster, F., 2015. A new device to foster oak forest restoration via seed sowing. N. For. 46, 919–929. https://doi.org/10.1007/s11056-015-9478-4.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: Instituto Nacional para la Conservación de la Naturaleza. Ministerio de Agricultura, Pesca y Alimentación, Madrid, pp. 156–158.

Çiçek, E., Tilki, F., 2007. Seed size effects on germination, survival and seedling growth of *Castanea sativa* Mill. J. Biol. Sci. 7 (2), 438–441.

Ciordia, M., Feitoa, I., Pereira-Lorenzo, S., Fernandez, A., Majada, J., 2012. Adaptive diversity in *Castanea sativa* Mill. half-sib progenies in response to drought stress. Environ. Exp. Bot. 78, 56–63. https://doi.org/10.1016/j.envexpbot.2011.12.018.

Ciordia, M., García, J.C., Loureiro, M.D., 2020. Hot water treatment: an effective method for disinfecting *Castanea sativa* Mill. dormant scions against *Dryocosmus kuriphilus* Yasumatsu. Pest Manag. Sci. 76 (5), 1944–1948. https://doi.org/10.1002/ps.5727.

Clark, S.L., Marcolin, E., Patrício, M.S., Loewe-Munoz, V., 2023. A silvicultural synthesis of sweet (*Castanea sativa*) and American (*C. dentata*) chestnuts. For. Ecol. Manag. 539, 121041. https://doi.org/10.1016/j.foreco.2023.121041.

Crawford, M., 1995. Chestnuts, Production and Culture. Agroforestry Research Trust, UK, p. 52.

Cuenca Valera, B., Majada Guijo, J., 2012. *Castanea sativa* mill. In: García, J.P., Cerrillo, R.M.N., Peragón, J.L.N., Prada Sáez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Tomos I y II. Organismo autónomo Parques Nacionales. Ministerio de Agricultura, Alimentación y Medio Ambiente.

Fernandes, P., Colavolpe, M.B., Serrazina, S., Costa, R.L., 2022. European and American chestnuts: an overview of the main threats and control efforts. Front. Plant Sci. 13, 951844. https://www.frontiersin.org/journals/plant-science/articles/10.3389/fpls.2022.951844/full.

Fuertes-Mendizábal, T., Huérfano, X., Ortega, U., González-Murua, C., Estavillo, J.M., Salcedo, I., Duñabeitia, M.K., 2021. Compost and PGP-based biostimulant as alternative to peat and NPK fertilization in chestnut (*Castanea sativa* Mill.) nursery production. Forests 12, 850. https://doi.org/10.3390/f12070850.

Gonzalez, O.M., Chung, G.P., Loewe, M.V., Reinoso, R., October, 2010. Efecto de micorrizas comestibles en el crecimiento en vivero y campo de plantas de castaño (*Castanea sativa*). Chilean Congress of Forest Sciences. Temuco, Chile.

Gradi, A., 1980. Vivaistica Forestale. Edagricole, Bologna, p. 568.

Gresshoff, P.M., Doy, C.H., 1972. Development and differentiation of haploid *Lycopersicon esculentum* (tomato). Planta 107 (2), 161–170. https://doi.org/10.1007/BF00387721.

Hipps, N.A., Hipps, K.H., Collard, L.G., 1997. Effects of root wrenching and irrigation rate on the growth and water relations of *Castanea sativa* and *Quercus robur* seedlings in the nursery and after outplanting. Can. J. For. Res. 27 (2), 180–188. https://doi.org/10.1139/x96-175.

Institute pour le Developpement Forestier (IDF), 1990. Boiser Une Terre Agricole. Paris, France, p. 64.

Louro, V., Pinto, G., 2011. Sementes, uma ponte entra o passado e o futuro sa floresta. Ministerio da Agricultura, Mar, Ambiente e Ordenamento do Territorio. CENASEF, pp. 31–38.

Manetti, M.C., Conedera, M., Pelleri, F., Montini, P., Maltoni, A., Mariotti, B., Pividori, M., Marcolin, E., 2022. Optimizing quality wood production in chestnut coppices. For. Ecol. Manag. 523, 120490. https://doi.org/10.1016/j. foreco.2022.120490.

Mencarelli, F., 2001. Postharvest handling and storage of chestnuts. Working document of the project: TCP/CPR/8925 integrated pest management and storage of chestnuts in XinXian county, Henan province, China. Food Agricult. Organizat. U. Nat. On-line https://www.fao.org/4/AC645E/ac645e00.htm#Table%20of%20Contents.

Moraldi, M., 2023. Compendio di Vivaistica Forestale. ARSIAL Regione Lazio, Roma, Italy.

Murashige, T., Skoog, F., 1962. A revised medium for rapid growth and bio assays with tobacco tissue cultures. Physiol. Plant. 15 (3), 473–497. https://doi.org/10.1111/j.1399-3054.1962.tb08052.x.

Navarro Cerillo, R.M., Galvez, C., 2001. Manual Para la Identificación y Reproducción de Semillas de Especies Vegetales Autóctonas de Andalucía. Tomo I. Consejería de Medio Ambiente. Junta de Andalucía, Córdoba. Spain, pp. 100–102.

Patrício, M.S., 2006. Analise da Potencialidade Produtiva do Castanheiro em Portugal. Dissertation, Universidade Tecnica de Lisboa. Instituto Superior de Agronomia, Lisbon, Portugal.

Patrício, M.S., Nunes, L., 2017. Density management diagrams for sweet chestnut high-forest stands in Portugal. iForest 10, 865–870. https://doi.org/10.3832/ifor2411-010.

Pence, A.V., 1992. Desiccation and survival of *Aesculus, Castanea* and *Quercus* embryo axes through cryoconservation. Cryobiology 29, 391–399. https://doi.org/10.1016/0011-2240(92)90040-9.

Piotto, B., 1992. Semi di Alberi e Arbusti in Italia: Come e Quando Seminarli. Società Agricola e Forestale, ENCC, Roma. Italy.

Ribeiro, D., Ribeiro, H., Louro, V., 2001. Producao im Viveiros Florestais. Direcao General de Desenvolvimento Rural, Lisboa, Portugal.

Seabra, R.C., Simões, A.M., Baeta, J., Pais, M.S., 2001. Evaluation of Portuguese chestnut stands by RAPDs. For. Snow Landsc. Res. 76 (3), 435–438.

Serdar, Ü., Saito, T., Cuenca, B., Burak, A.B., Gomes, L.J., Beccaro, G., Bounous, G., Rita Lourenço, C. R., Fernandes, P., Mellano, M.G., Pereira, L.S., 2019. Advances in cultivation of chestnuts. In: Serdar, Ü., Fulbright, D. (Eds.), Achiving Sustainable Cultivation of Tree Nuts. Burleigh Dodds Science Publishing, Cambridge, UK. https://doi.org/10.19103/AS.2018.0042.22. e-chapter.

Stilinović, S., 1987. Proizvodnja Sadnog Materijala Šumskog i Ukrasnog Drveća i Žbunja. Univerzitet U Beogradu, Šumarski Fakultet, Udžbenik; P:303./Seedling Production of Forest and Ornamental Trees and Shrubs. University of Belgrade, Faculty of Forestry (Textbook).

Tumpa, K., Vidakovic, A., Drvodelic, D., Sango, M., Idzojtic, M., Perkovic, I., Poljak, I., 2021. The effect of seed size on germination and seedling growth in sweet chestnut (*Castanea sativa* Mill.). Forests 12 (7), 858. https://doi.org/10.3390/f12070858.

Vieitez, A.M., Vieitez, M.L., Vieitez, E., 1986. Chestnut (Castanea spp). In: Bajaj, Y.P.S. (Ed.), Biotechnology in Agriculture and Forestry, vol 1. Springer, Berlin Heidelberg New York, pp. 393–414.

Vieitez, E., 1992. The lack of rootability of chestnut cuttings. In: Double, M.L., MacDonald, W.L. (Eds.), Proceedings International Chestnut Conference. West Virginia University Press, Morgantown, WV, USA, pp. 82–88.

Corylus colurna L.

Petar Zhelev[1], Muhidin Šeho[2], Krasimira Petkova[1] and Ina Aneva[3]
[1]University of Forestry, Sofia, Bulgaria; [2]Bavarian Office for Forest Genetics (AWG), Teisendorf, Germany; [3]Department of Plant and Fungal Diversity and Resources, Institute of Biodiversity and Ecosystem Research, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

Harvesting of *Corylus colurna* (Turkish hazel) fruit is done manually from late August to October when the sheath turns brown (Alexandrov, 1995; Alteheld, 1996; Šeho et al., 2019). The nuts fall close to the parent tree and are mainly spread by wildlife. The sheath must be removed manually, and seeds can be stored for up to 6 months at room temperature, up to 1 year in a refrigerator at 40%–60% relative air humidity and 2–5°C, or up to 3 years at relative air humidity 15%–40% and −2 to −4°C (Šeho et al., 2019; Nikolova et al., 2021). Viability after long term storage is low (Šeho et al., 2016). Fruit seed composition depends on the variety, harvest year, soil, climate, and method of cultivation (Köksal et al., 2006; Oliveira et al., 2008; Alasalvar et al., 2009).

Germination can vary from 30% to 90% depending on the year and locality (Šeho et al., 2019). Due to the high moisture content of the seeds, autumn sowing is recommended. If stored and sown in spring, seeds need stratification for 5–7 months before sowing (Šeho et al., 2019; Nikolova et al., 2021; Ninić-Todorović et al., 2012). One type of stratification is placing seeds in wet sand at a constant temperature of 1–5°C. Another method involves soaking seeds in water at room temperature for 4–5 days, changing the water every day, followed by 4–6 months in sand at 1–5°C (Milev et al., 2004; Aygün et al., 2009, Vilmorin Tree Seeds, 2024). Germination from autumn sowing could reach 80%, but if sown in spring after stratification germination is strongly reduced to about 20% (Šeho et al., 2019). Treatment with gibberellic acid increases germination and removing the pericarp could increase germination by up to 30% (Šeho et al., 2019; Jansky et al., 1988.). The average characteristics of seed lots are summarized in Table 11.6.

Vegetative propagation

The experience with vegetative propagation of *C. colurna* is relatively limited (Alexandrov, 1995, Young, 2001). The species has been used as a rootstock for grafting *Corylus avellana* cultivars because it demonstrates a wide tolerance to different soil conditions, a high resistance to frost and drought, and does not form suckers (Ninić-Todorović et al., 2012). Semihardwood cuttings should be collected between July and November (Young, 2001). They must be at least 1 cm in diameter at the lower end and a minimum length of 12 cm, with at least three nodes, and should be kept moist and cool prior to

Table 11.6 Characteristics of seed lots of *Corylus colurna*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
45–70 (60)	85–98 (90)	550–820 (700)	34–90 (70)

Mean values in brackets.
Šeho et al. (2019); Nikolova et al. (2021); Aygün et al. (2009); Ninić Todorović (2000).

treatment. The procedure consists of dipping cuttings in a mild bleach solution for 30 s, then treatment with Hormex (3000 ppm IBA) rooting powder and placing the cuttings in the greenhouse in 5 cm deep trays. Regular watering with misting is necessary until the roots form. The rooted cuttings could be transplanted after 50 days. The rooting success rate is ~50% (Young, 2001). Although in vitro propagation of *C. colurna* has been demonstrated, final acclimatization of micropropagated plants has not yet been achieved (Kosenko et al., 2009), indicating a need for further refinement of this method.

Seedling cultivation

Container plants

Container plant production for forestry purposes is currently not done. There is some information on container production for ornamental and/or horticultural purposes. Container production can be recommended only for one-year-old seedlings, because studies in Bavaria have shown that the root development of container plants is not optimal. The roots do not develop further after the first year (Šeho et al., 2019).

Bare root plants

Natural populations of *C. colurna* are located in regions with continental or Mediterranean mountain climate in the Balkans, Türkiye, Caucasus, Iran, and Afghanistan. Environmental conditions in the nursery must be close to the conditions of the planting sites. Seeds are sown in rows at a depth of 2–4 cm. Length of the rows can vary depending on the specific size of the nursery. The distance between rows depends on the number of rows per bed; at 4, 5, and 6 rows per bed, distance is 30, 22.5, and 18 cm, respectively. One kg of seeds could yield about 440 1-year-old seedlings. The production norms are 25–30 1-year old seedlings/m, and 20 2-year old seedlings/m (Milev et al., 2004; Iliev et al., 2015). Seedlings can be temporarily stored outside in the nursery. For grafting, seedlings $2 + 0$ are preferred, as $1 + 0$ seedlings are not considered viable rootstocks (Šeho et al., 2019). Morphological characteristics of bare root seedlings are summarized in Table 11.7.

Table 11.7 Observed and recommended morphological attributes of different stock types of bare root *Corylus colurna* seedlings.

	1 + 0 bareroot seedlings		2 + 0 bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	13–26	>20	42–110	>50
Root collar diameter (cm)	0.56–0.59		0.9–1.2	
Shoot weight (g)	1.8–2.8		9.2–12.6	
Root weight (g)	2.9–4.2		11.0–17.5	

Alexandrov (1995); Iliev et al. (2015); Šeho et al. (2019).

Field establishment

Water stress and herbivory by deer, rabbits, and mice are the main environmental limitations for seedling establishment of *C. colurna*. We recommend 1 + 0 and 2 + 0 seedlings for reforestation and afforestation. Planting can be performed in the autumn or in early spring (not later than the end of March). Seedling growth can differ substantially on the same site, depending on the genotype (Šeho et al., 2023). The species thrives across a wide range of site conditions, including flat to sloping terrain, dry to nearly wet environments, gravelly areas, older terraces, and soils varying from nutrient-poor to nutrient-rich. However, sites with excessive moisture or extremely dry soils are generally unsuitable for this species (Alteheld, 1996). *C. colurna* is also sensitive to soil compaction (Pauls, 2006), exhibits only partial tolerance to saline soils, and is rarely found on raw or peaty substrates (Bärtels, 1985).

C. colurna develops a robust taproot, reaching depths of 3–4 m, which enables it to thrive on skeletal, nutrient-poor, rocky, or shallow soils (Alteheld, 1996; Šeho et al., 2019). It shows optimal growth on nitrogen-poor sites and requires high base availability. The ideal pH range is between 5.5 and 8; the species can tolerate acidic soils, but they are not optimal for growth (Petkova et al., 2017). Root protection treatment with alginates is recommended to reduce desiccation. Protection against wildlife and rodents may be needed in the early years postplanting (Richter, 2017; Šeho et al., 2019).

Mature trees can develop extensive lateral roots that extend beyond the crown, providing significant stem stability (Alteheld, 1996). The species' leaf litter decomposes efficiently and has a beneficial effect on soil quality, particularly when mixed with hornbeam litter. Owing to its robust root system, Turkish hazel is well-suited for erosion control.

Chapter References

Alasalvar, C., Amaral, J.S., Satir, G., Shahidi, F., 2009. Lipid characteristics and essential minerals of native Turkish hazelnut varieties (*Corylus avellana* L.). Food Chem. 113 (4), 919–925. https://doi.org/10.1016/j.foodchem.2008.08.019.

Alexandrov, A.H., 1995. Corylus colurna. In: Schütt, P. (Ed.), Enzyklopädie der Holzgewächse. Handbuch und Atlas der Holzgewächse. 2. Erg.Lfg. Landsberg am Lech: Ecomed-Verlag. Band III-2. S. 215 – 222.

Alteheld, R., 1996. Die Turkish hazel: Monographie einer Baumart. In: Baumkunde. Band 1. Eching. IHW-Verlag, Germany, pp. 39–75.

Aygün, A., Erdogan, V., Bozkurt, E., 2009. Effect of some pretreatments on seed germination of Turkish Hazel (*Corylus colurna* L.). Acta Hortic. 845, 203–206. https://doi.org/10.17660/ActaHortic.2009.845.27.

Bärtels, A., 1985. Der Baumschulbetrieb. 3. Aufl. Stuttgart: Ulmer, Germany.

Jansky, A., Hartmann, W., Stoesser, R., 1988. Untersuchungen zur Keimung der Baumhasel. Deutsche Baumschule 1\. - agris.fao.org.

Iliev, N., Milev, M., Alexandrov, P., Petkova, K., 2015. Production of Seedlings of Tree and Shrub Species. University of Forestry Publishing House, Sofia, 333 pp. (in Bulgarian).

Köksal, A.İ., Artik, N., Simşek, A., Güneş, N., 2006. Nutrient composition of hazelnut (*Corylus avellana* L.) varieties cultivated in Turkey. Food Chem. 99 (3), 509–515. https://doi.org/10.1016/j.foodchem.2005.08.013.

Kosenko, I.S., Boyko, A.L., Opalko, A.I., Nebykov, M.V., Tarasenko, G.A., 2009. Micropropagation of *Corylus colurna* L. Acta Hortic. 845, 261–266. https://doi.org/10.17660/ActaHortic.2009.845.37.

Milev, M., Alexandrov, P., Petkova, K., Iliev, N., 2004. Sowing Materials of Broadleaved Species. Videnov & Son Publ, House, Sofia (in Bulgarian).

Nikolova, A., Tsvetanova, Z., Koyuva, L., 2021. Guidebook LIFEFORHAB. Forest Seed Control Station, Sofia (in Bulgarian). https://www.lifeforhab.eu/wp-content/uploads/2021/03/Guidebook-LIFEFORHAB-text.pdf.

Ninić-Todorović, J., 2000. Postharvest physiology of Turkish Filbert (*Corylus colurna* L.) seeds. NUCIS Newslett. 9, 27–32.

Ninić-Todorović, J., Ognjanov, V., Keserović, Z., Cerović, S., Bijelić, S., Cukanović, J., Kurjakov, A., Cabilovski, R., 2012. Turkish Hazel (*Corylus colurna* L.) offspring variability as a foundation for grafting rootstock production. Bulgar. J. Agricult. Sci. 18 (6), 883–888.

Oliveira, I., Sousa, A., Morais, J.S., Ferreira, I.C.F.R., Bento, A., Estevinho, L., Pereira, J.A., 2008. Chemical composition, and anti-oxidant and antimicrobial activities of three hazelnut (*Corylus avellana* L.) cultivars. Food Chem. Toxicol. 46 (5), 1801–1807. https://doi.org/10.1016/j.fct.2008.01.026.

Pauls, T., 2006. Die Turkish hazel (*Corylus colurna* L.) – mehr als ein Alleebaum. Mitt. Dtsch. Dendrol. Ges. 91, 91–199.

Petkova, K., Huber, G., Baier, R., Šeho, M., 2017. Turkish hazel in Bulgaria-an autochthonous and valuable tree species for the climate change. In: IUFRO 125th Anniversary Congress, 18–22 September 2017, Freiburg, Germany. Book of Abstracts, p. 459.

Richter, E., 2017. Baumhasel – Datensammlung Waldbau und Holzeigenschaften. https://www.baumhasel.info/datensammlung. (Accessed 30 October 2024).

Šeho, M., Ebinger, T., Huber, G., Konnert, M., 2016. Baumhasel - Saatgut und Vermehrung im Fokus. Dtsch. Baumsch. 8, 42–45.

Šeho, M., Ayan, S., Huber, G., Kahveci, G., 2019. A review on Turkish hazel (*Corylus colurna* L.): a promising tree species for future assisted migration attempts. South-East Europ. Forest. 10 (1), 53–63. https://doi.org/10.15177/seefor.19-04.

Šeho, M., Kavaliauskas, D., Ayan, S., Čokeša, V., Petkova, K., Fussi, B., 2023. First insights into genetic structure and diversity of *Corylus colurna* L. for conservation of its genetic resources and possible assisted migration. For. Ecol. Manag. 549, 121480. https://doi.org/10.1016/j.foreco.2023.121480.

Vilmorin, 2024. Tree Seeds. https://www.vilmorin-tree-seeds.com/seeds/ornamental-nurseries/1108-corylus-colurna.html. (Accessed 30 October 2024).

Young, B., 2001. Propagation protocol for vegetative production of *container Corylus cornuta* (A. DC.) Sharp var. californica (A. DC.) (Depot 40 and Treeband #10); Golden Gate National Parks, San Francisco, California. In: Native Plant Network. University of Idaho, College of Natural Resources, Forest Research Nursery, Moscow (ID). http://www.nativeplantnetwork.org.

Fagus sylvatica L.

Ivona Kerkez Janković[1], Juan A. Oliet[2] and Heinrich Lösing[3]
[1]University of Belgrade, Faculty of Forestry, Belgrade, Serbia; [2]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [3]VuB Versuchs-und Beratungsring Baumschulen, Ellerhoop, Germany

Propagation

Seed propagation

Fagus sylvatica (beech) produces irregular nut crops, with mast years occurring every 5–8 years (Houston Durrant et al., 2016). Nuts ripen between September and October, and early frosts help the splitting of the husks and nut release. Early seed falls usually have empty shells (Fletcher, 1992). Harvesting nuts too early can reduce seed viability (Ulbricht et al., 2016), which is also highly influenced by storage conditions (Procházková et al., 2002). Drought and high temperature in summer significantly affect fruiting success (Hilton and Packham 1997; Hacket-Pain et al., 2015; Nussbaumer et al., 2016).

The quality of fruits depends on the presence of other *F. sylvatica* trees. Isolated trees located >100 m from other reproductive individuals can produce up to 92% empty nuts due to self-incompatibility (Packham et al., 2012). Collecting seeds from large areas can result in significant variability in germination rates and subsequent nursery growth (Bezděčková and Matějka, 2015).

Seed harvesting is done manually, either from the ground or using seed traps or nets. Impurities, such as cupules and damaged seeds, can be removed manually or through water floating (Simak, 1973). Cleaning stones from seed collections can be challenging due to their similar size to *F. sylvatica* nuts, but this can be resolved using specific gravity tables. When using water flotation, seeds must be dried afterward. The optimal process is to spread the beech nuts on a flat surface and blow air over them while turning the bulk repeatedly until surface moisture evaporates. For long-term storage, nuts are dried down to 9% moisture content by blowing air at a maximum temperature around 18–20°C and afterward stored in sealed bags at −5 to −10°C. Before sowing, beechnuts have to be rehydrated and stratified for 8–12 weeks at 3–5°C aiming to increase nut moisture content to 30%–32%. However, if sowing is done immediately after collection, additional

Table 11.8 Characteristics of seed lots of *Fagus sylvatica*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
84–93 (88)	90–99 (97)	3,000–10,000 (4866)	37–89 (57)

Mean values in brackets.
Regent (1980); Suszka et al. (1996); Procházková et al. (2002); Šijačić-Nikolić et al. (2007); Bonner and Leak (2008); Drvodelić et al. (2011); Gavranović et al. (2018).

drying is unnecessary (Gordon, 1992). The average characteristics of seed lots are summarized in Table 11.8.

F. sylvatica seeds show a deep embryo dormancy, which significantly influences germination and seedling growth (Pawłowski et al., 2024). Dormancy release is associated with a reduction in the abscisic acid concentration in the seed (Vondráková et al., 2020) and changes in the expression of key proteins and hormones during cold stratification (Bezděčková and Matějka, 2015). At harvest, *F. sylvatica* seeds can have a moisture content of up to 40%. Seeds sown immediately after collection from late October to mid-November do not need pretreatment as in winter the nuts are exposed to natural cold stratification. However, nuts should be sown in containers and kept in a greenhouse during the winter to avoid frost damage. If not sown right after harvest, storage conditions and pretreatment methods such as cold stratification and moisture control are critical to optimize germination rate and speed. The most common storage method involves keeping the seeds in a refrigerator at 5°C in paper or plastic bags for up to 6 months (Kerkez et al., 2018).

The effect of storage on seed quality depends largely on moisture content, making it more critical than the duration of storage (Suszka, 1974; Suszka and Zieta, 1977; Muller and Bonnet-Masimbert, 1982; Muller, 1993; Procházková and Bezděčková, 2008). During storage and stratification, mold fungi can grow on seeds, potentially leading to decay. Nuts with an 8% moisture content can be stored at −3°C for up to 5 years without significant viability loss (Gordon, 1992). Optimal germination for stored seeds requires cold, moist stratification for 8–19 weeks as a presowing treatment (Szczotka et al., 2003; Rezah et al., 2010). Moisture levels should not be < 25%–30% at sowing, as a strong reduction in moisture can damage seeds and reduce germination (Gordon, 1992; Falleri et al., 2004).

Vegetative propagation

Although not commonly practiced in forestry, *F. sylvatica* can be propagated vegetatively through cuttings, grafting, and in vitro techniques. Research on *F. sylvatica* propagation by cuttings is relatively scarce, likely due its difficulty, although some success has been reported with stem cuttings taken in late summer (Bonner and Leak, 2008; Jurásek, 2018). However, these sources did not provide information on harvesting time, preparation, processing, or storage of vegetative material. Cuttings from crown branches of mature trees generally do not root but those harvested from 1- to 5-year-old plants can have rooting rates of up to 90%, although with low survival rates during propagation (Vieitez et al., 2003). Jurásek (2018) found that cuttings cultivated in the nursery for three or 4 years in 1 L containers to produce large plants, can be successfully used to establish forest stands with <10% mortality.

In vitro propagation techniques for *F. sylvatica* include micropropagation and protoplast culture mainly aimed at propagating pathogen-resistant genotypes (Vieitez et al.,

2003). Explant sources include shoot tips, buds of mature trees and seedlings, embryo explants, leaves of juvenile and mature trees, immature zygotic embryo, anthers; they are mostly cultivated on Murashige and Skoog and woody plant media. Few studies have successfully produced plantlets and even fewer have reached the acclimatization stage. Large-scale regeneration with conventional propagation methods remains uncertain, requiring further improvement. Grafting is a method common for ornamental purposes (Bonner and Leak, 2008; Nonić et al., 2017).

Seedling cultivation

Container plants

F. sylvatica is mainly produced as bare root stock in Europe. However, facing climate change, especially in the southern range of its distribution and in areas prone to summer drought, container seedlings may offer some advantages (Scott and Skibsted, 2007; Oliet et al., 2012; Mauer et al., 2018). In Northern and Central Europe, container stock is less competitive due to higher production cost (Scott and Skibsted, 2007).

For container plants it is recommended to use products with structures such as vertical ridges or lateral openings to prevent root spiraling and promote proper root growth and structure (Oliet et al., 2012; Škorić and Devetaković, 2015). A minimum container volume of 200 mL is recommended (Oliet et al., 2012), although evidence of performance benefits from larger containers (e.g., 265 mL) compared to 150 mL container is limited (Scott and Skibsted, 2007). However, seedlings cultivated in 200 mL containers had higher survival than those cultivated in 93 and 50 mL containers, but still lower survival compared with survival of bare root plants (Madsen et al., 2006). According to our experience container seedlings have lower growth than bare root plants of the same age (Kerkez Janković, unpublished results).

Outplanting success of *F. sylvatica* depends on the seedlings' competitive capacity against natural vegetation, which can be enhanced by planting larger seedlings. To produce seedlings at least 10–15 cm in height and 4 mm in diameter, cultivation density should be < 250 plants/m^2 (Oliet et al., 2012). While peat is the most common growing medium, alternative, cheaper, and peat-free substrates can be used, such as composted bark from pines or other conifers (Oliet et al., 2012). Mixtures of wood shavings and chips, straw, bark, perlite, core wood, and mixed silage, supplemented with liquid fertilizers has shown positive effects on root morphology and growth 1 year after planting (Rotowa et al., 2023, 2024).

Controlled-release fertilizers with a minimum release period of 5 months (e.g., 15-15-15 or 15-10-15 NPK + essential micronutrients) are recommended at rate of 2–4 g/L of substrate with up to 6 g/L for producing larger plants (Oliet et al., 2004). Combining Yara Mila Complex and Osmocote Exact fertilizers in sawdust-and-peat

growing medium is effective for increasing growth, but increases costs (Banach et al., 2021). Additions of Ca, Mg, and K can improve growth (Pająk et al., 2002; Balcar et al., 2011; Wang et al., 2020), with Mg fertilizers being particularly effective (Wang et al., 2020). The use of dolomite (3 kg/m^3) to achieve a pH of 5.5 increases seedlings Mg content in plant and enzymatic action in growing media, decreases acidification, and has a positive effect on seedlings growth, particularly on their root systems (Lasota et al., 2021; Rotowa et al., 2024).

Inoculating peat with mycorrhiza increases seedling establishment (Banach et al., 2013) and drought resistance (Manzanedo et al., 2018). Excessive irrigation during the germination phase can favor *Phytophthora* spp. proliferation, which can be managed with preventive fungicides like Dimethomorph and Fosetyl (Oliet et al., 2012). Weed control can be done manually and through different types of mulches such as perlite or vermiculite, although these do not completely inhibit weed growth (Rys et al., 2014).

Compared with bareroot, container seedlings are more resistant to storage conditions, in air-conditioned environments, or open storage with stable temperatures (e.g., 2–6°C) and moderate humidity (58%–75%) to prevent desiccation (Pantová et al., 2021). For overwinter storage, the optimal temperature varies between −1.7 and 3.4°C. Short-term storage (up to 1 month) is feasible at temperatures between −5.6



Figure 11.4 *Fagus sylvatica* container seedlings ready for transport. *(Vladan Ivetić.)*

and −5.9°C with mortality risk increasing at temperatures between −6.6 and −8.4°C (Pantová et al., 2021) (Fig. 11.4).

Bareroot plants

Nursery cultivation of bareroot stock can be done in seedbeds or Dunemann beds (deep beds of loosely packed leaf-mold) sown either in autumn (November) or in spring (late February–March). Before sowing, treating seeds with fungicides and rodenticides is recommended. Since provenance significantly influences *F. sylvatica* seedling quality, seeds should ideally come from sources with environmental conditions similar to the nursery.

Seedbed preparation is mainly done just before sowing and involves mowing, chopping, and disking cover crops (if any), deep ripping (if needed), soil disking and plowing, and bedding. Sowing density depends on seed quality, the desired final plant size, and machinery use. Final cultivation densities vary from 120 to 150 seedlings/m^2 for 1-year-old plants, and about 120 seedlings/m^2 for 2-year-old seedlings. Row spacing is typically 20 cm between rows, with 3.3–4.2 cm between plants within a row. Sowing can be done using a seeder with sowing depths of 2–3 cm in spring and up to 6 cm in autumn (Oliet et al., 2012).

Several stock types can be found in *F. sylvatica,* which include 1 + 0, 2 + 0, 1 + 1, 1 + 2, 2 + 2, and 1 + 3 plants (Oliet et al., 2012), but 1 + 0 and 2 + 0 seedlings are the most common (Ivetić et al., 2017). For producing 2 + 0 stock, root pruning in autumn is recommended (Lösing, 1997). Root undercutting promotes the formation of lateral roots, which enhances outplanting survival (McKay et al., 1999).

Shading is recommended during the first vegetation period. Irrigation should be done regularly, especially during summer, and if needed in spring before germination, due to the sensitivity of *F. sylvatica* to drying out. Fertilization can be done using granular NPK fertilizers applied at approximately 300 kg/ha, once or twice annually, while foliar fertilizers with micronutrients at a dose of approximately 3 mL/L can be applied monthly.

Excessive watering of container stock during germination should be avoided to prevent the development of *Phytophthora* spp. Preventive treatments include fungicides such as Dimethomorph, Fosetyl, Metalaxyl, or Propamocarb in combination with a contact fungicide like Mancozeb after germination (Oliet et al., 2012). Crop rotation is recommended to mitigate fungal issues. The *F. sylvatica* woolly aphid (*Phyllaphis fagi*) can be controlled with neonicotinoid products, such as Clothianidin, Imidacloprid, Thiacloprid, and Thiamethoxam (Oliet et al., 2012). Acetamiprid is also effective for gall-forming pests (*Dasyneura fagicola* and *Contarinia fagi*) that can damage terminal buds. Pyrethroid-based products, such as Cyhalothrin, can be used in combination (Oliet et al., 2012). Weed control is crucial as grasses significantly reduce seedling growth (Coll et al., 2004). It can be done manually or using selective herbicides such as

Clethodim and Pendimethalin for narrow- and broadleaved weeds, particularly for seedlings with woody stems. Hardening seedlings through controlled water deprivation can improve drought resilience (Yang et al., 2022).

Root pruning is common for bareroot stock after lifting, but we consider that it should be avoided, regardless of transplanting conditions as it increases outplanting mortality particularly in *F. sylvatica* bareroot seedlings (Andersen, 2001). Dormant seedlings can be stored in polyethylene bags (protecting the root system and the entire plant, separately) in freezers or air-conditioned warehouses, maintaining the temperature between −3.4 and −1.7°C (Pantová et al., 2021) and the relative humidity above 80% (Wilnen and Vaartaja, 1958).

Plant quality is important; seedlings with significant physical damage, poor root development, signs of disease, dehydration, or incomplete lignification of stem and branches should not be used in forest plantations (Alía et al., 2005). Root electrolyte leakage tests can be used to assess the quality of large seedling batches at lifting and for cold storage. The acceptable value for bareroot seedlings is 30%, with the baseline when fully dormant being 10%–20%. These electrolyte leakage values indicate a high outplanting survival potential of over 90% (Morgan, 1999).

Table 11.9 depicts observed and recommended values for bareroot and containerized *F. sylvatica* seedlings. General requirements for *F. sylvatica* seedlings according to current standard in Serbia (SRPS D.Z2.110:1968) are at least 4 mm in diameter and 15 cm tall for 1 + 0 seedlings and 5 mm in diameter and 30 cm height for 2 + 0 seedlings. British Commission standards require seedlings taller than 20 cm (Oliet al., 2012).

Table 11.9 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 and 2 + 0 container seedlings of *Fagus sylvatica*.

Attributes	Observed values		Recommended values
	Container seedlings	Bareroot seedlings	
1 + 0 seedlings			
Shoot height (cm)	8–48	10–51	>15
Root collar diameter (cm)	0.29–0.54	0.24–0.60	>0.4
Shoot dry weight (g)	0.71–2.71	–	–
Root dry weight (g)	0.63–1.69	–	–
2 + 0 seedlings			
Shoot height (cm)	23–28	21.2–64.5	>30
Root collar diameter (cm)	0.56–0.66	0.32–0.85	>0.5
Shoot dry weight (g)	2.67–4.10	–	–
Root dry weight (g)	4.21–5.24	–	–

SRPS D.Z2.110: (1968); Jurásek et al. (2010); Oliet al. (2012); Banach et al. (2013), Ivetić et al. (2017); Popović et al. (2021); Banach et al. (2021), Pająk et al. (2022); Kormanek et al. (2024); Popović et al. (2024).

Field establishment

Establishment of *F. sylvatica* is mainly influenced by water availability and drought stress, temperature extremes, light, and provenance (Jump et al., 2007; Stojnić et al., 2015; Wang et al., 2021; Prigoliti et al., 2023). Direct seeding is an option but faces many challenges, such as seed predation by rodents, herbivory of emerging seedlings, and competition from vegetation (Löf et al., 2004; Birkedal, 2010). Planting therefore remains the preferred forestation method. According to our experience, planting 2 + 0 (or 3 + 0) seedlings, taller than 20 cm with well-developed fine roots, results in optimum results, with mortality <10%. However, for bareroot 3 + 0 seedlings taller than 45 cm we have observed die back and resprouting from the stem base, suggesting that this stock type is more prone to stress.

To ensure establishment, competitive vegetation should be removed, and planting holes of 40 × 40 cm and up to 50 cm deep (depending on root development) can be dug manually or by mechanical methods. Chemical control of weed competition can be accomplished by using herbicides such as glyphosate during the vegetative period and preemergent herbicides like propyzamide. In the short-term (3–6 years), planting density has little influence on outplanting performance (Houšková and Mauer, 2013). Typical densities are 1.5–2 × 1.5–2 m spacing, but studies in *Fagus orientalis* indicate that closer spacing (0.7 × 2 m) can result in greater biomass production and carbon storage in 25-year-old plantations, despite smaller tree dimensions, compared with wider 2 × 2 m spacing (Güner et al., 2010). For artificial regeneration, mostly involving planting, large seedlings (bareroot or container) are recommended at densities >1500 seedlings/ha (Wagner et al., 2010) and up to 10,000 plants/ha (Buesa, 2006) to help maintaining apical dominance and prevent the growth of thick lateral branches (Oliet et al., 2012).

Survival of plantings can vary widely (Catorci et al., 2012; Hasstedt and Annighöfer, 2020). Key factors reducing survival include poor weed control (Repáč et al., 2018), lack of pioneer shrubs that protect seedlings from browsers and extreme radiation, low soil moisture during summer, and browsing (Catorci et al., 2012). Survival is increased in fenced plots as it reduces browser damage and is decreased under strong shade conditions (Hasstedt and Annighöfer, 2020).

Mycorrhization with ectomycorrhiza before planting showed little effect on seedling growth (Repáč et al., 2018), but combining it with hydrogel significantly increased the survival under dry conditions (Repáč et al., 2023).

Chapter References

Alía, R., Alba, N., Agúndez, D., Iglesias, S., 2005. Manual para la comercialización y producción de semillas y plantas forestales. Materiales de base y de reproducción. Serie Forestal. DGB, Madrid, 384 pp.

Andersen, L., 2001. Survival and growth of *Fagus sylvatica* seedlings root-pruned prior to transplanting under competitive conditions. Scand. J. For. Res. 16, 318–323.
Banach, J., Skrzyszewska, K., Świeboda, Ł., 2013. Substrate influences the height of one- and two-year-old seedlings of silver fir and European beech growing in polystyrene containers. Leśne Prace Badawcze 74, 117–125. https://doi.org/10.2478/frp-2013-0012.
Banach, J., Kempf, M., Skrzyszewska, K., Olejnik, K., 2021. The effect of starter fertilization on the growth of seedlings of European beech (*Fagus sylvatica* L.). Sylwan 165, 565–576.
Bezděčková, L., Matějka, K., 2015. The quality and the depth of dormancy of beechnuts in individual stand groups with varying climatic conditions within a single unit of approval. J. For. Sci. 61, 382–392.
Birkedal, M., 2010. Reforestation by Direct Seeding of Beech and Oak: Influence of Granivorous Rodents and Site Preparation. Doctoral Thesis. Swedish University of Agricultural Sciences, Alnarp.
Bonner, F.T., Leak, W.B., 2008. Fagaceae – beech family. *Fagus L. beech*. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual. USDA Forest Service, Washington, DC, pp. 520–524. Agriculture Handbook No. 727.
Buesa, A., 2006. El haya en Bizkaya. Diputación Foral de Bizkaia. Spain.
Catorci, A., Scapin, W., Tardella, F.M., Vitanzi, A., 2012. Seedling survival and dynamics of upper timberline in Central Apennines. Pol. J. Ecol. 60, 79–94.
Coll, L., Balandier, P., Picon-Cochard, C., 2004. Morphological and physiological responses of beech (*Fagus sylvatica*) seedlings to grass-induced belowground competition. Tree Physiol. 24, 45–54.
Drvodelić, D., Oršanić, M., Paulić, V., Rožman, M., 2011. Morphological-biological properties of fruit and seed of beech (*Fagus sylvatica* L.) growing at different altitudes. Glas. šum. pokuse: Annales Experim. Silv. Cult. Provehen. 44, 1–18.
Falleri, E., Muller, C., Laroppe, E., 2004. Effect of water stress on germination of beechnuts treated before and after storage. Canad. J. Forest. Res. 34, 1204–1209.
Fletcher, A.M., 1992. Flower, fruit and seed development and morphology. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees, Forestry Commission Bulletin, vol 83. HMSO, London, pp. 59–70.
Gavranović, A., Bogdan, S., Lanšćak, M., Čehulić, I., Ivanković, M., 2018. Seed yield and morphological variations of beechnuts in four European beech (*Fagus sylvatica* L.) populations in Croatia. South-East Eur. For. 9 (1), 17–27. https://doi.org/10.15177/SEEFOR.18-06.
Gordon, A.G., 1992. The processing of cones and seeds. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees, Forestry Commission Bulletin, vol 83. HMSO, London, pp. 86–97.
Güner, S., Yagci, V., Tilki, F., Celik, N., 2010. The effects of initial planting density on above- and belowground biomass in a 25-year-old *Fagus orientalis* Lipsky plantation in Hopa, Turkey. Sci. Res. Essays 5, 1856–1860. https://doi.org/10.5897/SRE.9000909.
Hacket-Pain, A.J., Friend, A.D., Lageard, J.G., Thomas, P.A., 2015. The influence of masting phenomenon on growth–climate relationships in trees: explaining the influence of previous summers' climate on ring width. Tree Physiol. 35, 319–330.
Hasstedt, S.L., Annighöfer, P., 2020. Initial survival and development of planted European beech (*Fagus sylvatica* L.) and small-leaved lime (*Tilia cordata* Mill.) seedlings competing with black cherry (*Prunus serotina* Ehrh.). Plants 9, 677. https://doi.org/10.3390/PLANTS9060677.
Hilton, G.M., Packham, J.R., 1997. A sixteen-year record of regional and temporal variation in the fruiting of beech (*Fagus sylvatica* L.) in England (1980–1995). Forestry 70, 7–16.
Houšková, K., Mauer, O., 2013. Effect of the density of transplants in reforestation on the morphological quality of the above-ground part of European beech (*Fagus sylvatica* L.) six years after planting. Acta Univ. Agric. Silvic. Mendelianae Brun. 61, 1703–1713. https://doi.org/10.11118/ACTAUN201361061703.
Houston Durrant, T., de Rigo, D., Caudullo, G., 2016. *Fagus sylvatica* and other beeches in Europe: distribution, habitat, usage and threats. In: San-Miguel-Ayanz, J., de Rigo, D., Caudullo, G., Houston Durrant, T., Mauri, A. (Eds.), European Atlas of Forest Tree Species. Publ. Off. EU, Luxembourg e012b90+.
Ivetić, V., Maksimović, Z., Kerkez, I., Devetaković, J., 2017. Seedling quality in Serbia - results from a three-year survey. Reforesta 4, 27–53. https://doi.org/10.21750/REFOR.4.04.43.

Jump, A.S., Hunt, J.M., Peñuelas, J., 2007. Climate relationships of growth and establishment across the altitudinal range of *Fagus sylvatica* in the Montseny Mountains, northeast Spain. Écoscience 14, 507–518. https://doi.org/10.2980/1195-6860(2007)14[507:CROGAE]2.0.CO;2.

Jurásek, A., 2018. Possibilities of using rooted cuttings of European beech (*Fagus sylvatica* L.) for stabilisation of forest ecosystems. J. For. Sci. 53, 498–504.

Jurásek, A., Leugner, J., Martincová, J., 2010. Growth and physiological state of beech seedlings grown in a nursery in different light conditions. J. For. Sci. 56, 442–450.

Kerkez, I., Pavlović, S., Lučić, A., Devetaković, J., Šijačić, N.M., Popović, V., 2018. Different methods for beech seed quality testing. Sustain. Forest. Collect. 77–78, 1–10.

Kormanek, M., Małek, S., Tabor, S., 2024. Resistance to pulling seedlings out of the nursery container. Forests 15, 2157.

Lasota, J., Kempf, M., Kempf, P., Błońska, E., 2021. Effect of dolomite fertilization on nutritional status of seedlings and soil properties in forest nursery. Soil Sci. Ann. 72, 132236.

Löf, M., Thomsen, A., Madsen, P., 2004. Sowing and transplanting of broadleaves (*Fagus sylvatica* L., *Quercus robur* L., *Prunus avium* L. and *Crataegus monogyna* Jacq.) for afforestation of farmland. For. Ecol. Manag. 188 (1–3), 113–123.

Lösing, H., 1997. Erste Versuchsergebnisse zum Knospensterben an *Fagus sylvatica*. Anuell Report, Versuchs- und Beratungsring, pp. 29–32.

Madsen, P., Bentsen, N., Madsen, T.L., Olesen, C.R., 2006. Artificial beech regeneration in Denmark - development of direct seeding and planting methods. In: Beech Silviculture in Europe's Largest Beech Country. Proceedings, pp. 64–66.

Manzanedo, R.D., Schanz, F.R., Fischer, M., Allan, E., 2018. *Fagus sylvatica* seedlings show provenance differentiation rather than adaptation to soil in a transplant experiment. BMC Ecol. 18, 42.

Mauer, O., Rozmánek, M., Houšková, K., 2018. Drought spells and their impact on the growth of young plantations established with the containerized planting stock. Acta Univ. Agric. Silvic. Mendelianae Brunensis 66, 86–99.

McKay, H.M., Jinks, R.L., McEvoy, C., 1999. The effect of desiccation and rough-handling on the survival and early growth of ash, beech, birch and oak seedlings. Ann. For. Sci. 56, 391–402.

Morgan, J.L., 1999. Forest tree seedlings: best practice in supply, treatment and planting. For. Commun. Bull. 121.

Muller, C., 1993. Combination of dormancy-breaking and storage for tree seeds: new strategies for hardwood species. In: Edwards, D.G.W. (Ed.), Dormancy and Barriers to Germination. Proceedings of an International Symposium of IUFRO Project Group P2.04-00 (Seed Problems), Victoria, British Columbia, Canada, 23–26 April 1991, pp. 79–85.

Muller, C., Bonnet-Masimbert, M., 1982. Long term storage of beechnuts: results of large-scale trials. In: Wang, B.S.P., Pitel, J.A. (Eds.), Proceedings International Symposium on Forest Seed Storage; 23–27 Sep. 1980, Canadian Forestry Service Publication, pp. 178–183.

Nonić, M., Skocajic, D., Grbic, M., Šijačić-Nikolić, M., 2017. Variability of quantitative and qualitative characteristics of *Fagus sylvatica* 'Purpurea' clones produced by grafting. Not. Bot. Horti Agrobot. Cluj-Napoca 45 (2), 400–407.

Nussbaumer, A., Waldner, P., Etzold, S., Gessler, A., Benham, S., Thomsen, I., Jørgensen, B., Timmermann, V., Verstraeten, A., Sioen, G., Rautio, P., Ukonmaanaho, L., Skudnik, M., Apuhtin, V., Braun, S., Wauer, A., 2016. Patterns of mast fruiting of common beech, sessile and common oak, Norway spruce and Scots pine in Central and Northern Europe. For. Ecol. Manag. 363, 237–251.

Oliet, J., Planelles, R., Segura, M.L., Artero, F., Jacobs, D.F., 2004. Mineral nutrition and growth of containerized *Pinus halepensis* seedlings under controlled-release fertilization. Sci. Hortic. 103, 113–129.

Oliet, J.A., Pardo Navarro, F., Lösing, H., 2012. *Fagus sylvatica* L. In: Pemán Garcia, J., Navarro Cerrillo, R., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y manejo de semillas y plantas forestales. Organismo Autónomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, Spain, pp. 492–515.

Packham, J.R., Thomas, P.A., Atkinson, M.D., Degen, T., 2012. Biological flora of the British Isles: *Fagus sylvatica*. J. Ecol. 100, 1557–1608.

Pająk, K., Kormanek, M., Małek, S., Banach, J., 2022. Effect of peat-perlite substrate compaction in Hiko V265 trays on the growth of *Fagus sylvatica* L. seedlings. Sustainability 14, 4585.

Pająk, K., Małek, S., Kormanek, M., Jasik, M., Banach, J., 2002. Macronutrient content in European beech (*Fagus sylvatica* L.) seedlings grown in differently compacted peat substrates in a container nursery. Forests 13, 1793.

Pantová, P., Houšková, K., Mauer, O., 2021. Overwinter storage of European beech and Norway spruce planting stock: effect of different methods and temperature conditions. Forests 12, 1286.

Pawłowski, T.A., Suszka, J., Mucha, J., Zadworny, M., Alipour, S., Kurpisz, B., Chmielarz, P., Jagodziński, A.M., Chmura, D.J., 2024. Climate legacy in seed and seedling traits of European beech populations. Front. Plant Sci. 15, 1355328.

Popović, V., Lučić, A., Rakonjac, L., 2021. Variability of morphological traits of European beech (*Fagus sylvatica* L.) seedlings in Serbia. South-East Europ. Forest. 12, 83–89.

Popović, V., Vemić, A., Rakonjac, L., Jovanović, S., Hadrović, S., Lučić, A., 2024. The use of morphological traits of European beech (*Fagus sylvatica* L.) seedlings for selection of reproductive material from the provenances likely affected by climate change. Šumarski List 7–8, 353–362.

Prigoliti, M., Chiofalo, M.T., Petruzzellis, F., Lo Gullo, M.A., Trifilò, P., 2023. Ecophysiological behavior of *Fagus sylvatica* L. growing at its southern distribution limit: insights for understanding the fate of the European beech under warmer and drier growth conditions. Forests 14, 2058. https://doi.org/10.3390/f14102058.

Procházková, Z., Bezděčková, L., 2008. Effects of moisture content, storage temperature and type of storage bag on the germination and viability of stored European beech (*Fagus sylvatica* L.) seeds. J. For. Sci. 54, 287–293.

Procházková, Z., Bezdecková, L., Martincová, J., Palátová, E., 2002. Quality of beechnuts from different crop years. Dendrobiology 47, 39–42.

Regent, B., 1980. Šumsko sjemenarstvo. In: Jugoslovenski Poljoprivredni Gumarski Centar-Sluzba Gumske Proizvodnje, Beograd, p. 201.

Repáč, I., Tučeková, A., Sarvašová, I., Vencurik, J., 2018. Survival and growth of outplanted seedlings of selected tree species on the High Tatra Mts. windthrow area after the first growing season. J. For. Sci. 57, 349–358. https://doi.org/10.17221/130/2010-JFS.

Repáč, I., Belko, M., Krajmerová, D., Kmeť, J., Pavlík, M., 2023. Induced drought stress response of European beech seedlings treated with hydrogel and ectomycorrhizal inoculum. Forests 14, 1749. https://doi.org/10.3390/F14091749.

Rezah, A., Nasery, B., Yazdian, F., Hedayati, M.A., Kolar, I., 2010. Pregermination treatment and germination characteristics of oriental beech (*Fagus orientalis* Lipsky) in the Caspian region. Acta Silvatica Lignaria Hung. 6, 143–148.

Rotowa, O.J., Małek, S., Banach, J., Pach, M., 2023. Effect of different innovative substrate mediums on roots characterization of European beech (*Fagus sylvatica* L.) and pedunculate oak (*Quercus robur* L.) seedlings. Sylwan 167, 09.

Rotowa, O.J., Małek, S., Kupka, D., Pach, M., Banach, J., 2024. Impact of innovative peat-free organic substrates and fertilizers on root morphology and growth dynamics of *Fagus sylvatica* L. and *Quercus robur* L. seedlings one year after planting. SSRN. https://doi.org/10.2139/ssrn.4872724.

Rys, F., van Wesemael, D., van Haecke, D., Mechant, E., Gobin, B., 2014. Mulches and other cover materials to reduce weed growth in container-grown nursery stock. Commun. Agric. Appl. Biol. Sci. 79, 47–52.

Scott, B.N., Skibsted, J.H., 2007. Field performance of containerized beech seedlings: preliminary results. In: Abstract from the International Beech Symposium IUFRO Research Group, Teheran, Islamic Republic of Iran, p. 25.

Simak, M., 1973. Separation of forest seed through flotation. In: Seed Processing, Proc. IUFRO Wkg. Group on Seed Problems, Bergen. Vol. I, Paper 16.

Šijačić, N.M., Ivetić, V., Knežević, R., Milovanović, J., 2007. Analysis of seed and seedling traits of different provenances of beech. Acta Herbol. 16 (1), 15–27.

Škorić, M., Devetaković, J., 2015. Seedling production in Gočko container. In: Ivetić, V., Stanković, D. (Eds.), Proceedings: International Conference Reforestation Challenges, 03–06 June 2015, Belgrade, Serbia. Reforesta, pp. 99–103.

SRPS D.Z2.110:1968. 1968 - Forest seedlings - General requirements. Yugoslavia.

Stojnić, S., Orlović, S., Ballian, D., Ivanković, M., Šijačić-Nikolić, M., Pilipović, A., Bogdan, S., Kvesić, S., Mataruga, M., Daničić, V., Cvjetković, B., Miljkovic, D., Von Wuehlisch, G., 2015. Provenance by site interaction and stability analysis of European beech (*Fagus sylvatica* L.) provenances grown in common garden experiments. Silvae Genet. 64, 133–147. https://doi.org/10.1515/SG-2015-0013.

Suszka, B., 1974. Storage of beech (*Fagus sylvatica* L.) seeds for up to 5 winters. Arbor. Kornickie 19, 105–128.

Suszka, B., Muller, C., Bonnet-Masimbert, M., 1996. Seeds of Forest Broadleaves: From Harvest to Sowing Practiques. INRA, 334 pp.

Suszka, B., Zieta, L., 1977. A new presowing treatment for cold stored beech (*Fagus sylvatica* L.) seed chilled without medium at a controlled hydration level and pregerminated in cold moist conditions. Arbor. Kornickie 22, 237–255.

Szczotka, Z., Pawłowski, T., Krawiarz, K., 2003. Proteins and polyamines during dormancy breaking of European beech (*Fagus sylvatica* L.) seeds. Acta Physiolog. Plant. 25, 423–435.

Ulbricht, M., Göttlein, A., Biber, P., Dieler, J., Pretzsch, H., 2016. Variations of nutrient concentrations and contents between summer and autumn within tree compartments of European beech (*Fagus sylvatica*). J. Plant Nutr. Soil Sci. 179, 746–757.

Vieitez, A.M., San-Jose, M.C., Sánchez, M.C., Ballester, A., 2003. Micropropagation of Fagus spp. In: Jain, S.M., Ishii, K. (Eds.), Micropropagation of Woody Trees and Fruits. Kluwer Academic Publishers, pp. 181–215.

Vondráková, Z., Pešek, B., Malbeck, J., Bezděčková, L., Vondrák, T., Fischerová, L., Eliášová, K., 2020. Dormancy breaking in *Fagus sylvatica* seeds is linked to formation of abscisic acid-glucosyl ester. New For. 51, 671–688.

Wagner, S., Collet, C., Madsen, P., Nakashizuka, T., Nyland, R.D., Sagheb-Talebi, K., 2010. Beech regeneration research: from ecological to silvicultural aspects. For. Ecol. Manag. 259, 2172–2182.

Wang, Z., Hassan, M.U., Nadeem, F., Wu, L., Zhang, F., Li, X., 2020. Magnesium fertilization improves crop yield in most production systems: A meta-analysis. Front. Plant Sci. 10, 495191.

Wang, F., Israel, D., Ramírez-Valiente, J.A., Sánchez-Gómez, D., Aranda, I., Aphalo, P.J., Robson, T.M., 2021. Seedlings from marginal and core populations of European beech (*Fagus sylvatica* L.) respond differently to imposed drought and shade. Trees 35, 53–67. https://doi.org/10.1007/S00468-020-02011-9.

Wilnen, J., Vaartaja, O., 1958. Prevention of injury to tree seedlings during cellar storage. For. Chron. 34, 132–138.

Yang, F., Burzlaff, T., Rennenberg, H., 2022. Drought hardening of European beech (*Fagus sylvatica* L.) and silver fir (*Abies alba* Mill.) seedlings in mixed cultivation. Forests 13, 1386.

Fraxinus angustifolia Vahl.

Nebi Bilir[1], Georgi Hinkov[2] and Ivaylo Tsvetkov[2]
[1]Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye; [2]Forest Research Institute, Department Forest Genetics, Physiology and Plantations, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

Seeds of *Fraxinus angustifolia* Vahl. (narrow-leaved ash) ripen between October and November when they are brown (del Campo et al., 2012). Seeds enter a deep physiological dormancy that needs to be released through pretreatments if sown in spring. However, if sown in autumn the seeds do not need any pretreatment (Piotto, 1997; del Campo et al., 2012). Dormancy can be broken by the following two methods. The first procedure involves in vitro embryo culture, which provides the best results compared with intact or partially cut seeds, producing nearly 90% viable plants (Raquin et al., 2002). This method, however, is difficult to implement at the operational scale. The second method involves 4 weeks of warm stratification at +25°C and 4–8 weeks of cold stratification at +5°C; or 6 weeks of cold stratification at +5°C. Alternatively, Piotto (1997) recommends combining stratification of 15 days at +25°C with an additional 15 days at +3–5°C and drying to a 9.5% moisture content. Draghici and Aburdan (2011) tested four seed stratification treatments and concluded that a cold stratification at 3°C in a mixture of sand and peat showed the highest germination. In an experiment with nine stratification treatments in moistened sand, Tilki and Cicek (2005) concluded that the most effective pretreatments were: 16 weeks at 4°C, 4 weeks at 20°C followed by 4 weeks at 4°C, or 4 weeks at 20°C followed by 8 weeks at 20°C; germination varied between 40% and 45%, while control seeds showed 19% germination.

Seeds can be stored in bags and boxes for 2–3 years under conditions of −3 to +5°C and 9%–15% air humidity. When hermetically sealed, seeds can be stored for up to 5 years with a moisture content of 6%–8% at a temperature of −4 to −10°C (Nikolova et al., 2020). Cryopreservation of seeds at low moisture content (3%) allows for germplasm conservation and high (70%) germination in water (Lombardo et al., 2013).

The average seed characteristics of *F. angustifolia* are shown in Table 11.10. Seed weight and germination capacity can show notable differences among provenances (Cicek et al., 2005).

Vegetative propagation

Vegetative propagation by cuttings is not a common practice in *F. angustifolia* due to its low rooting capacity (Fraxigen, 2005). Nevertheless, *Fraxinus* species could be

Table 11.10 Characteristics of seed lots of *Fraxinus angustifolia*.

Purity (%)	No. seeds/kg	Germination capacity (%)
84–100 (92)	10,000–25,000 (17,500)	42–80 (61)

Mean values in brackets.
Piotto and Piccini (1998); Cicek et al. (2005); del Campo et al. (2012); Prada and Arizpe (2008); Nikolova et al. (2020).

propagated by budding, grafting, and layering (Hartmann et al., 1997). Cicek (2005) observed that cutting age and the position in the mother tree had a significant effect on rooting capacity, plant survival, and diameter growth, while height growth was only affected by position in the mother tree. One-year-old cuttings showed a higher rooting (56.0%) than 2-year-old cuttings (35.3%).

An alternative to classical vegetative propagation is in vitro propagation. Pérez-Parrón et al. (1994) reported successful micropropagation of *F. angustifolia* using both mature and juvenile plant material as initial explants. Studying the organogenic capacity of different embryo portions of mature *F. angustifolia* seeds, Tonon et al. (2001a) succeeded in regenerating shoots, which were easily rooted in hormone-free Woody Plant Medium (with calcium chloride, vitamins and sucrose without agar) (Lloyd and McCown, 1980). Most (80%) of the formed plantlets were successfully acclimated in the greenhouse. Induction of somatic embryogenesis from immature embryos of *F. angustifolia*, and the sequential development, encapsulation, germination, and conversion of somatic embryos to plantlets, have been reported by Tonon et al. (2001a, b).

Seedling cultivation

Container seedlings

Container production of seedlings is an alternative to bareroot stock, and it is particularly useful if shorter production time is required and it can extend the planting season (Bozic et al., 2021). The volume of the most used containers ranges from 250 to 400 cm^3 (sometimes up to 500 cm^3) and 15 cm depth. Recommended cultivation density varies between 200 and 350 seedlings/m^2 with seedlings grown under a moderate 20% shade especially in hot summer locations. Common organic growing media are blonde peat and coconut fiber (coir) mixed with 10%–15% vermiculite with a pH 6–7 (del Campo et al., 2012). Drvodelić and Oršanić (2020) do not recommend to use black peat for cultivating this species, as it dries out easily during the summer, particularly in hot locations, making subsequent rehydration and water uptake difficult. Slow-release fertilizers are recommended for extended periods, with a preference for thermostable brands that

do not release nutrients too quickly. Observed and recommended morphological characteristics of *F. angustifolia* container stock are shown in Table 11.11.

Bareroot seedlings

Bareroot cultivation of *F. angustifolia* is relatively common and recommended, although today container production is more widespread (del Campo et al., 2012). However, bareroot stock is a common practice in central Europe, while container stock is used more in Mediterranean countries (del Campo et al., 2012). The germination period is usually about 3–4 weeks. In the case of soil deficiencies or low pH, amendments must be made before sowing using organic amendments, base fertilization, etc (del Campo et al., 2012). The height of 1 + 0 plants can reach 20–30. According to Bobinac et al. (2010), 1 + 0 bareroot seedlings of *F. angustifolia* are bigger than container-grown seedlings. As in some other riparian tree species, the preferred planting stock type is 1 + 2 (Bozic et al., 2021).

For ashes, Nikolova et al. (2020) recommend a sowing rate in seedbeds of 75 (137) seeds per linear/m. del Campo et al. (2012) recommend cultivation densities between 80 and 150 plants/m^2. Cicek et al. (2007a) conducted a nursery experiment and tested densities ranging between 24 and 120 seedlings/m^2. As in many other species, seedling growth decreased with closer spacing. The highest growth was observed at 24 seedlings/m^2 (rows 33 cm apart, plants at 12.5 cm the within row). Fidan (2017) suggests adjusting cultivation density based on soil fertility. The recommended cultivation densities are 50 seedlings/m^2 if the soil is amended with 2 m^3 of organic matter per 150 m^2 of seed bed or 25 seedlings/m^2 with 1 m^3 of organic matter or no amendment.

Leaf parameters, especially total leaf area are valuable morphological attributes of seedling quality in *F. angustifolia* as they are strongly correlated with seedling growth

Table 11.11 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 seedlings of *Fraxinus angustifolia*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	21.0	–	29–71	40.0–70.0
Root collar diameter (cm)	0.5	–	0.4–1.2	0.6–0.8
Shoot dry weight (g)	–	–	2.6–15.8	–
Root dry weight (g)	–	–	0.5–21.1	–
Plant dry weight (g)	–	–	7.6–16.3	–
Shoot/root dry weight ratio	–	–	1.0–1.7	–

Anonymous (1988); Cicek et al. (2007a); Fidan (2017); Fidan et al. (2020); Drvodelic and Orsanic (2020).

and can complement other morphological attributes (Kerkez Janković et al., 2020). Some morphological characteristics of *F. angustifolia* bareroot stock are shown in Table 11.11. Attributes can strongly vary depending on provenance, nursery practices, and environment (Cicek et al., 2007a; Fidan, 2017; Fidan et al., 2020).

Field establishment

F. angustifolia seedlings produced at low density (24 seedlings/m^2) in the nursery showed higher outplanting growth when established at 2 × 2.5 m spacing (Cicek et al., 2007b). In a later study, Cicek et al. (2011) reported that the optimal field growth was observed in seedlings cultivated in the nursery at 40 seedlings/m^2. Growth, but not survival of rooted cuttings has been reported to be higher than the growth of 1 + 0 bareroot seedlings after one and three growing seasons (Cicek et al., 2006, 2010). *F. angustifolia* seedlings had higher survival but lower growth after two growing seasons compared with *Ulmus laevis* and *U. minor* on a site prepared by cleaning vegetation, and ripping and disking soil (Cicek et al., 2007b).

Soil preparation can vary depending on the stock type and soil features. In compacted or poor soils, subsoiling to 40–60 cm deep is recommended, whereas in high-quality alluvial plains, conventional tillage can be used. For large plants, deep holes excavated with a backhoe is frequently used; auger-dug holes are discouraged in heavy soils (del Campo et al., 2012). Mechanical control is recommended over chemical methods for weedy competition and other woody species. In Mediterranean climate, irrigation with 20–40 L per plant may be necessary to mitigate summer drought during the first growing season, particularly if the water table is not close to the surface (del Campo et al., 2012). Planting densities can vary according the objectives, stock type, and mixture with other species, typically ranging between 700 and 1200 plants/ha. Ventilated tree shelters can increase growth if they do not strongly reduce irradiation (del Campo et al., 2012).

Chapter references

Anonymous, 1988. Seedling Quality Classification of Conifers. Turkish Standard Institute Press, Ankara, Türkiye.

Bobinac, M., Andrasav, S., Sijacic-Nikolic, M., 2010. Elements of growth and structure of narrow-leaved ash (*Fraxinus angustifolia* Vahl.) annual seedlings in the nursery of fluvisol. Period. Biol. 112 (3), 341–351.

Bozic, G., Westergren, M., Zeleznik, P., Konrad, H., Nagy, L., Lanscak, M., Novcic, Z., Rukavina, S., Schueler, S., Stojnic, S., 2021. Planting Material Use and Conservation Guidelines, Output 5.2, REFOCuS Interreg Project. https://www.interreg-danube.eu/approved-projects/refocus.

Cicek, E., 2005. Vegetative propagation of narrow-leaved ash (*Fr. angustifolia* Vahl.) by stem cuttings. J. of For. Facul. Gazi Univ. Kastamonu 5 (1), 112–127.

Cicek, N., Cicek, E., Bilir, N., 2005. Some seed and seedling characteristics in Narrow-leaved ash (*Fraxinus angustifolia* Vahl.). J. Forest. Facul. Suleyman Dem. Univers. A (1), 17–24.

Cicek, E., Tilki, F., Cicek, N., 2006. Field performance of narrow-leaved ash (*Fraxinus angustifolia* Vahl.) rooted cuttings and seedlings. J. Biol. Sci. 6 (4), 750–753.
Cicek, E., Cicek, N., Bilir, N., 2007a. Effects of seedbed density on one-year-old *Fraxinus angustifolia* seedling characteristics and outplanting performance. N. For. 33, 81–91.
Cicek, E., Tilki, F., Kulac, S., Yilmaz, M., Yilmaz, F., 2007b. Survival and growth of three hardwood species (*Fraxinus angustifolia, U. laevis* and *U. minor*) on a bottomland site with heavy clay soil. J. Plant Sci. 2 (2), 233–237.
Cicek, E., Tilki, F., Ozbayram, A., Cetin, B., 2010. Three-year growth comparison between rooted cuttings and seedlings of. *Fr. angustifolia* and *U. laevis*, J. of Appl. Sci. 6 (3), 199–204.
Cicek, E., Cicek, N., Tilki, F., 2011. Four-year field performance of *Fr. angustifolia* Vahl. and *Ulmus laevis* Pall. Seedlings growth at different seedbed densities. Res. J. Forest. 5 (2), 89–98.
del Campo, A.G., del Campo, M.V., Sáez, M.A., 2012. Fraxinus angustifolia. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), *Producción y manejo de semillas y plantas forestales*. Organismo Autónomo Parques Nacionales. Serie Forestal, Madrid, Spain, pp. 523–539 (in Spanish) Prada.
Draghici, C., Aburdan, I.V., 2011. The Effect of different stratification conditions on the germination of *Fraxinus angustifolia* Vahl. and *Fraxinus ornus* L. seeds. Not. Bot. Agrobot. Cluj. 39 (1), 283–287.
Drvodelić, D., Oršanić, M., 2020. Planting seedlings of narrow-leaved ash (*Fraxinus angustifolia* Vahl.) to recover dieback-affected forest stands. Sumarski list 5–6, 289–301.
Fidan, C., 2017. Effects of organic matter content and seedling density for growing bare-root narrow-leaved ash (*Fraxinus angustifolia* Vahl.) seedlings. Euras. J. For. Sci. 5 (2), 87–93.
Fidan, C., Ozbayram, A.K., Gultekin, H.C., Tamyuksel, H., 2020. Effect of sowing type and seedbed density on the morphological characteristics of Narrow-leaved ash seedlings in different nurseries. J. Forest. Facult. Kastamonu Univers. 20 (2), 74–85.
FRAXIGEN, 2005. Ash species in Europe: biological characteristics and practical guidelines for sustainable use. Oxford Forestry Institute, Department of Plant Sciences, University of Oxford, UK, p. 128.
Hartmann, H.T., Kester, D.E., Davies Jr., F.T., Geneve, R.L., 1997. Plant Propagation – Principles and Practices, sixth ed. Prentice Hall, Upper Saddle River, NJ, p. 770.
Kerkez Janković, I., Šijačić-Nikolić, M., Nonić, M., Devetaković, J., 2020. The growth of one-year-old narrow-leaved ash seedlings is strongly related to the leaf area parameters. Reforesta 10, 31–39.
Lombardo, G., Scialabba, A., Schicchi, R., 2013. Seed cryopreservation of *Fraxinus angustifolia* Vahl. African J. of Biotechnologie 12 (16), 1930–1936.
Lloyd, G., McCown, B., 1980. Commercially feasible micropropagation of mountain laurel, *Kalmia latifolia*, by use of shoot tip culture. Comb. Proc. Int. Plant Prop. Soc. 30, 421–427.
Nikolova, A., Tsvetanova, Z., Koyuva, L., 2020. Technical manual with information on characteristics, extraction, processing, sowing and storage of seeds of target tree species under the project: life16-NAT/BG/000817. In: Restoration and Improvement of the Nature Protection Status of Priority Forest Habitats from the NATURA 2000 Network in Bulgaria, 88 p. (in Bulgarian).
Pérez-Parrón, M.A., Gonzalez-Benito, M.E., Perez, C., 1994. Micropropagation of *Fraxinus angustifolia* from mature and juvenile plant material. Plant Cell. Tiss. Org. Cult. 37, 297–302.
Piotto, B., 1994. Effects of temperature on germination of stratified seeds of three ash species. Seed Sci. and Tech. 22, 519–529.
Piotto, B., 1997. Storage of non-dormant seeds of *Fraxinus angustifolia* Vahl. New For 14, 157–166.
Piotto, B., Piccini, C., 1998. Influence of pretreatment and temperature on the germination of *Fraxinus angustifolia*. Seeds. Seed Sci. and Tech. 26, 799–812.
Prada, M., Arizpe, D., 2008. Riparian Tree and Shrub Propagation Handbook: An Aid to Riverine Restoration in the Mediterranean Region. *Fraxinus angustifolia* Vahl. Publisher Generalitat Valenciana, Valencia, Spain, pp. 59–62.
Raquin, C., Jung-Muller, B., Dufour, J., Frascaria-Lacoste, N., 2002. Rapid seedling obtaining from European ash species *Fraxinus excelsior* L. and *Fraxinus angustifolia* Vahl. Ann. For. Sci. 59, 219–224.

Tilki, F., Cicek, E., 2005. Effects of stratification, temperature and storage on germination in three provenances of *Fraxinus angustifolia* subsp. *oxycarpa* seeds. Turk. J. Agric. For. 29, 12. Available at: https://journals.tubitak.gov.tr/agriculture/vol29/iss4/12.

Tonon, G., Capuana, M., Di Marco, A., 2001a. Plant regeneration of *Fraxinus angustifolia* by *in vitro* shoot organogenesis. Sci. Hortic. 87, 291–301.

Tonon, G., Capuana, M., Rossi, C., 2001b. Somatic embryogenesis and embryo encapsulation in *Fraxinus angustifolia* Vahl. J. Hortic. Sci. Biotech 76, 753–757.

Fraxinus excelsior L.

Barbara Mariotti[1], Halil Barõş Özel[2] and Nebi Bilir[3]
[1]Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy; [2]Faculty of Forestry, Department of Silviculture, University of Bartõn, Bartõn, Türkiye; [3]Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye

Propagation

Seed propagation

Fraxinus excelsior L., common ash, produces fruits called samaras, which typically ripen from late September to November and are commonly harvested in late October or November (Betti and Cappanelli, 2009). Ripe fruits persist on the tree for an extended period, facilitating prolonged collection if needed (Cisneros González et al., 2014). Seed characteristics can vary according to regions and altitude (Kollas et al., 2012). High-quality seeds sourced from *Source Identified Seed Stands* to *Selected Seed Stands* and *Clonal Seed Orchards* (as per EU Dir. 1999/105 - Marketing of forest reproductive material) are accessible across the EU. Provenance trials and breeding programs focusing on wood productivity are conducted in many EU countries (Douglas et al., 2013). Seed orchards, capable of significant seed production within 7–10 years (Douglas et al., 2013), are established throughout Europe, including in Sweden, Denmark, Germany, Slovakia, France, and Italy. Harvesting involves manual methods such as using sacks or nets from the ground, vibrators, pole pruners, ropes, aerial platforms, ladders, or tree-climbing.

Fruit maturity is assessed visually by the transition from green to brown. Manual or mechanical sifting and sieving is used to separate seeds from fruit residues and impurities; usually samara wings are not removed. Assessing seed maturity is considerably more complex. *F. excelsior* exhibits one of the most intricate dormancy types, namely endogenous morpho-physiological dormancy (Bacchetta et al., 2006; Piotto and Noi, 2001), necessitating pretreatments to induce germination. Rapid tests, such as dissection of sample seeds to examine endosperm development and consistency, and assessment of seed moisture content above 50%, can assist in determining harvest time, but may not be conclusive.

Embryo size in autumn ranges between 45% and 55% (Suszka et al., 1994), requiring a warm phase to maturity. The process involves humid stratification in sand at 20°C for 8–16 weeks, followed by a cold phase at 3–4°C for 16–32 weeks (Cisneros González et al., 2014; Sambeek and Preece, 2007; Piotto and Noi, 2001). The warm phase could be reduced to 4 weeks if the seed is cleaned so that samara tissues are not a barrier to hydration. However, this could damage the seed, so it is generally not recommended (Cisneros González et al., 2014). A widely used method to estimate the time needed to overcome seed dormancy involves determining the required cold stratification time (Cisneros González et al., 2014). Freshly collected seeds lack sufficient moisture for stratification, therefore, a sample of seeds is moistened and stored at 3°C until approximately

10% of the sample seeds begin to germinate, as indicated by the presence of rootlets. This period determines the dormancy time up to emergence. The remaining seeds are moistened to 28–30% and stored in airtight flasks at 4°C for the estimated dormancy period plus an additional 2 weeks for safety (Cisneros González et al., 2014).

Storage method depends on the duration of storage. If seeds are stored until sowing in the spring following harvest (short-term conservation), it is sufficient to wash the seeds and store them in bags without any substrate at −3°C to −5°C, as long as the moisture content of the fruit is between 30% and 32% (Cisneros González et al., 2014; Zocca, 1999). Ash seeds are orthodox, so they can be stored at low humidity and temperature for a long time. Once dormancy has been broken, the seeds can be stored using a stratification process that blocks germination. The seeds can be slowly dehydrated to reach a water content of 8–10%, then stored at −3 and −5°C for several years (Cisneros González et al., 2014). Another method is storage under 8–10% humidity and 2–4°C (Zocca, 1999), highlighting that subzero conservation up to 7–10 years with little deterioration is increasingly common (Zocca, 1999). Particular attention is required during seed drying for long-term conservation, which should always be carried out at temperatures below 20°C (Cisneros González et al., 2014). The seed must regain a moisture content of 30–32% before sowing; this is achieved by stratification for about 2 weeks at about 5°C, providing the necessary cold stratification for germination of *F. excelsior*.

An alternative to storage is to sow seeds in autumn to naturally overcome dormancy. This requires placing containers with stratified seeds at −1 to −3°C up to 8–20 weeks. However, this method is considered less dependable (Cisneros González et al., 2014). If the seed lot is pretreated, sowing is done in early spring.

To assess seed viability by rapid tests, we recommend following the ISTA methods and rules (ISTA, 2021) or, for quicker results, the Forestry Commission suggests alternating temperatures from 3 to 20°C, reducing the expected germination time to 35 days (Cisneros González et al., 2014). A widely recommended option is to use a tetrazolium test. The average characteristics of *Fraxinus excelsior* seed lots are summarized in Table 11.12.

Movement of seed is permissible since the transmission of *Hymenoscyphus fraxineus*, the ascomycete fungus that causes ash dieback, from infested seed to germinating

Table 11.12 Characteristics of seed lots of *Fraxinus excelsior*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
65 (65)	90–98 (94.5)	9,000–26,000 (13,467)	50–83 (71)

Mean values in brackets.
Cisneros González et al. (2014); Gradi (1996, 1980); Gültekin (2008); Moraldi (2023); Piotto and Noi (2001); Takos (1998); Zocca (1999).

seedlings is negligible in this species. As a quarantine measure, however, any surface contamination by pathogen spores could be washed off (Marčiulynienė et al., 2018).

Vegetative propagation

F. excelsior, unlike other *Fraxinus* species, can be propagated by cuttings and grafting with encouraging results, up to 65% according to Cisneros González et al. (2014). This is not high enough, however, to be a widespread practice to produce nursery seedlings but in-vitro and grafting are used to propagate selected varieties or clones or to establish seed orchards (Piotto and Noi, 2001).

For vegetative propagation, both woody and semiwoody cuttings can be used. Woody cuttings about 15–20 cm long, can be obtained from branches (about 5 years old) on mature plants from June to July. Before placing cuttings in sand and perlite (1/1 v/v) for rooting, the bottoms of the cuttings should be submerged for a few seconds in a solution of IBA at 5000 ppm. Rooting should take place in a protected room at high humidity and seeds treated weekly with Captan for the first 30 days (Zocca, 1999). Semiwoody cuttings (15–20 cm long) should be taken in July from young shoots on young mother plants (2 or 3 years old). If such plants are not available, young suckers developed in the collar area of stumps or from pollarding can be used. Cuttings can be treated with solutions of IAA at 1000 ppm if they are placed in sand and perlite, or with IBA (5000 ppm) + Benlate if peat and sand substrates are used. The rooting benches should be placed in a protected humidified room (Zocca, 1999) or the substrate should be misted and heated, maintaining a temperature of 21°C. After about 6 weeks, the rooted cuttings can be transferred to unheated protected rooms where they will spend the fall-winter. Immediately before the beginning of the growing season, the rooted cuttings should be pruned leaving a single bud (Zocca, 1999).

Grafted material has the advantage of accelerating the onset of flowering compared with seeded plants and is therefore widely used in seed orchards. Grafting techniques depend on the size of scions and rootstocks available and the season. Cleft grafting and whip grafting are the most used techniques, which require woody scions and for rootstock, 2 to 3 year-old seedlings in containers (Zocca, 1999). Scions, 1-year-old shoots, are collected during the winter and cold stored at about 4°C in plastic bags (e.g., a garbage bag) to prevent the wood to dry out or in a moist (but not saturated) medium such as sand or peat moss. Properly stored scion wood should remain in good condition until ready for use in late winter or early spring. Grafting using these methods is done during early bud burst (March to April). The graft is wrapped with an elastic grafting band to keep it tight and prevent drying; if the wrapping material does not decompose naturally, it must be removed in about a month. To prevent the graft from drying out, the wrapped area must be evenly covered with wax. Among the bud grafting techniques, chip budding made in July to August has given good results (Zocca, 1999).

In vitro micropropagation can be successful in this species using stored seed, which can provide material for micropropagation regardless of the season, or using a shoot tip and nodal segment. The use of epicormic sprouts has not been reported for this species. In vitro micropropagation has been widely studied and is used to select specific genotypes (e.g., resistant to pest or disease). The establishment of *Fraxinus* spp. cultures in vitro is quite difficult, however, due to damaging bacterial flora and it is clone-dependent (Douglas et al., 2013).

Seedling cultivation

The estimated annual need for *F. excelsior* plants in reforestation and afforestation efforts reportedly exceeded 15 million seedlings across 18 countries, though these numbers probably were underestimated (Douglas et al., 2013). In the last decades, however, the spread of ash dieback (primarily caused by the fungus *Hymenoscyphus fraxineus*) has likely reduced production in the EU.

Container plants

Seeding time depends on whether dormancy treatments have been applied. If no treatments have been applied, seeding should be done outdoors in the fall and to ensure environmental conditions for breaking dormancy. If the winter at the nursery site is harsh, it is advisable to prevent damage from low temperatures and early freezing by placing containers in a greenhouse and then continue outdoors in mid-spring. Treated seeds are sown at the end of the winter (February to March) outdoors or in a greenhouse according to the climate.

One or two seeds can be directly sown in each container/pot/cell at about 2 cm depth with the samara placed horizontally with the main long axis. After seedling emergence, only one seedling should be left per container. *F. excelsior* is a tap-rooted species, thus it can be grown in containers used for similar hardwood species, such as single pots or cavities of 0.4–1.3 L volume. Root pruning is recommended or use containers with a structure that prevents root-system defects, such as air pruning (side slits, mesh) or mechanical obstacles (ribs, groves). Cultivation for 1 or 2 years is enough to produce high-quality seedlings (Armand, 1992; Calvo and D'Ambrosi, 1995; Maltoni et al., 2010). Seedlings are stored outdoors in the nursery. Further general information about cultivation practices for plants grown in containers can be found in Landis and Nisley (1990). Observed and recommended characteristics of container stock are shown in Table 11.13.

In the last 2 decades, the fungus *Hymenoscyphus fraxineus* (and less frequently other fungal species) has caused ash dieback, i.e., the dieback of the crown, branches, and shoots leading up to the death of young and old trees. Ash dieback has been a problem in many European countries in forest plantations, roadside trees, and in nurseries (EPPO, 2008; Van Der Linde et al., 2021). In the nursery, this can be controlled by spraying the

Table 11.13 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Fraxinus excelsior*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	15–125	40–60	24–120	40–60
Root collar diameter (cm)	0.4–1.4	>0.8	0.6–1.6	>0.8

Armand (1992); Božič et al. (2021); Gradi (1980); Ivetić et al. (2016); Ivetić et al. (2017); Maltoni et al. (2010); O'Reilly et al. (2003); Tani et al. (2008); Zocca (1999).

seedlings every 3 weeks with fungicides (Božič et al., 2021). Efforts are underway to breed for resistance; a scoring of resistance/tolerance to *Hymenoscyphus fraxineus* for young trees of *Fraxinus excelsior* has been provided to select resistant plants (Douglas et al., 2013).

Bareroot plants

As for container seedling, seeding time depends on pretreatments. Seeding density can range from 20 to 30 g (sowing in rows) to 60–90 g m^2 (Cisneros González et al., 2014) or 400–500 seeds (sowing in rows) per m^2 to produce 1-year-old seedlings or 250 to 300 seeds/m^2 for 2 years cultivation (Bellari et al., 1994; Takos, 1998). Seeds germinate in 14–25 days (Gradi, 1980); 40 to 60 viable seeds are recommended to obtain 30 to 50 seedlings/m^2 (Cisneros González et al., 2014; Gradi, 1980).

Sowing date can affect seedling height (Espahbodi and Khorankeh, 2013). When growth is pivotal to the project success (beyond survival), the window of opportunity for placing plants in cold storage is narrowed (O'Reilly et al., 2003), and it could cause shoot dieback (O'Reilly et al., 2003). Cold storage can reduce height growth once seedlings are planted in comparison with freshly lifted seedlings, but the effect may vary with lift date (O'Reilly et al., 2002, 2003); seedlings lifted to storage in early October grew less than seedlings lifted from December to March. Field performance trials in a northern Mediterranean floodplain environment have identified as more effective among different size classes of bareroot seedlings those seedlings that were 40–50 cm tall and equipped with a deep (40 cm), fasciculate root system rich in root fibrosity (Maltoni et al., 2010). For further information about bareroot seedlings grown in the nursery, tailored specifically for hardwood species, refer to the manual by McNabb and Pike (2019) (Fig. 11.5).

Field establishment

F. excelsior thrives best in fertile, pH-neutral, deep, moist, and well-drained soils, often dominating in floodplains and on moist clay-loam lowland sites. Soils conducive to ash



Figure 11.5 One-year old container (left) and bareroot (right) seedlings of *Fraxinus excelsior*. *(Vladan Ivetić.)*

growth are typically derived from calcareous, marl, or sedimentary parent material, rich in clay or silt (pH 5–7.5), moist but well-drained (Dobrowolska et al., 2011). This species exhibits sensitivity to severe winter and late spring frosts (Dobrowolska et al., 2011). Nevertheless, *F. excelsior* is a versatile species, thriving across diverse site types, except for acid soils. While the species demonstrates broad tolerance for moderate water supply variation, extended waterlogging is detrimental to growth. Optimal growth rates are achievable on sites with over 110 days of readily available water during the growing season (Dobrowolska et al., 2011). Conversely, growth falters on sites with more than 30 days of waterlogging in the upper half of the primary rooting horizon at the start of the growing season (Peltier and Marigo, 1998). Sites subjected to regular flooding, persisting for more than 30 days are unsuitable for planting, especially when the aim is quality timber production.

The light requirement of *F. excelsior* evolves with the developmental stage: seedlings and young plants can exhibit a slight shade tolerance that tends to diminish in adults. Consequently, regular interventions and wide spacing are crucial to maintain optimal light conditions and to provide sufficient soil volume for the roots to exploit. The root system establishes quickly and explores up to 1 meter depth in the first year, at least in southern Europe (Cisneros González et al., 2014). At higher latitudes, wide spacings (up to 4.96 m) were detrimental, with initial height growth and shoot and root biomass reduced compared with lower distances (up to 0.77 m) (Kerr, 2003).

To promote initial fast root growth, spot soil preparation is recommended, by removing soil in a $60 \times 60 \times 100$ cm area, preferably using a backhoe (Cisneros González et al., 2014). Proper drainage is crucial, so preliminary deep plowing before planting is recommended (Cisneros González et al., 2014). Plant density should be between 600 and 800 trees/ha, with the flexibility to lower this range based on high-quality seed sources (Cisneros González et al., 2014). Herb competition can be addressed by current local methods, including mulching and the judicious use of herbicides. The

protection from herbivores with tubes or netting is strongly recommended, especially if timber production is the goal. Field experience has shown that seedlings taller than 40–60 cm showed a better field performance (Ivetić et al., 2016; Maltoni et al., 2010; Tani et al., 2007). Side shelter, such as straw balls or plastic netting, are recommended when planting in windy sites (Willoughby et al., 2009).

Chapter References

Armand, G., 1992. Techniques de plantation des feuillus précieux: caractéristiques des plants de Merisier, Frêne et Érable sycomore. Rev. For. Française 44 (S), 66–70. https://doi.org/10.4267/2042/26384.

Bacchetta, G., Belletti, P., Brullo, S., Cagelli, L., Carasso, V., Casas, J.L., Cervelli, C., Escribà, M.C., Fenu, G., Gorian, F., Güemes, J., Mattana, E., Nepi, M., Pacini, E., Pavone, P., Piotto, B., Pontecorvo, C., Prada, A., Venora, G., Vietto, L., Virevair, M., 2006. Manuale per la raccolta, studio, conservazione e gestione del germoplasma. In: APAT- Agenzia per la protezione dell'ambiente e per i servizi tecnici, Roma (Italy).

Bellari, C., Giannini, R., Proietti Placidi, A.M., 1994. Semi e piantine di latifoglie arboree e arbustive. Provincia Autonoma di Trento. Servizio Foreste Caccia e Pesca, Trento, Italy.

Betti, A., Cappanelli, D., 2009. Le sementi Forestali. Manuale operativo per la produzione del germoplasma. In: Corpo forestale dello Stato - Centro Nazionale per lo Studio e la Conservazione della Biodiversità Forestale. Pieve Santo Stefano, Arezzo (Italy).

Božič, G., Westergren, M., Železnik, P., Konrad, H., Nagy, L., Lanšćak, M., Novčić, Z., Rukavina, S., Schueler, S., Stojnić, S., 2021. Planting Material Use and Conservation Guidelines - INTERREG REFOCUS.

Calvo, E., D'Ambrosi, E., 1995. Proposte di standard di idoneità colturale per il postime vivaistico di alcune latifoglie nobili. Monti Boschi 47, 22–24.

Cisneros González, Ó., Santana Pérez, J., Tomás Ferré, E., Aletà Soler, N., Ligos Martínez, J.T., Turrientes Calzada, A., 2014. *Fraxinus excelsior* L. In: Pemán Garcia, J., Navarro Cerrillo, R., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y manejo de semillas y plantas forestales. Organismo Autonomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio ambiente, Madrid, Spain, 2012.

Dobrowolska, D., Hein, S., Oosterbaan, a., Wagner, S., Clark, J., Skovsgaard, J.P., 2011. A review of European ash (*Fraxinus excelsior* L.): implications for silviculture. Forestry 84, 133–148. https://doi.org/10.1093/forestry/cpr001.

Douglas, G., Pliura, A., Dufour, J., Mertens, P., Jacques, D., Fernandez-Manjares, J., Buiteveld, J., Parnuta, G., Tudoroiu, M., Curnel, Y., Thomasset, M., 2013. Common ash (*Fraxinus excelsior* L.). In: Forest Tree Breeding in Europe. Springer, Netherlands, pp. 403–462.

EPPO, 2008. Hymenoscyphus Fraxineus. Eur. Mediterr. Plant Prot. Organ. (EPPO). https//gd.eppo.int/taxon/CHAAFR. (Accessed 11 December 2023).

Espahbodi, K., Khorankeh, S., 2013. Effect of planting date and seedling cover on seed germination of mountain ash (*Fraxinus excelsior* L.) and decrease of spring late cold damage. Iran J. For Poplar Res. 21, 126–141.

Gradi, A., 1980. Vivaistica Forestale. Edagricole, Bologna (Italy).

Gradi, A., 1996. Manuale pratico per l'allevamento in vivaio delle piantine forestali. In: Con integrazioni di vivaistica ornamentale. Regione Autonoma Fiuli-Venezia Giulia. Direzione Regionale Foreste e Parchi. Servizio Selvicoltura, Udine, Italy.

Gültekin, H.C., 2008. Some sexual characteristics of angiosperm trees and shrubs. Forest Hunt. J. (4), 42–47 (in Turkish).

ISTA, 2021. International rules for seed testing 2022. Int. Rules Seed Test. 2022.

Ivetić, V., Devetaković, J., Maksimović, Z., 2016. Initial height and diameter are equally related to survival and growth of hardwood seedlings in first year after field planting. Reforesta 2, 6–21. https://doi.org/10.21750/REFOR.2.02.17.

Ivetić, V., Maksimović, Z., Kerkez, I., Devetaković, J.R., 2017. Seedling quality in Serbia – results from a three-year survey. Reforesta 4. https://doi.org/10.21750/refor.4.04.43.

Kerr, G., 2003. Effects of spacing on the early growth of planted *Fraxinus excelsior* L. Can. J. Res. 33, 1196–1207. https://doi.org/10.1139/x03-041.

Kollas, C., Vitasse, Y., Randin, C.F., Hoch, G., Körner, C., 2012. Unrestricted quality of seeds in European broad-leaved tree species growing at the cold boundary of their distribution. Ann. Bot. 109, 473–480. https://doi.org/10.1093/aob/mcr299.

Landis, T.D., Nisley, R.G., 1990. The Container Tree Nursery Manual (No. 674). US Department of Agriculture, Forest Service.

Maltoni, A., Mariotti, B., Tani, A., Jacobs, D.F., 2010. Relation of *Fraxinus excelsior* seedling morphology to growth and root proliferation during field establishment. Scand. J. For. Res. 25, 60–67. https://doi.org/10.1080/02827581.2010.485805.

Marčiulynienė, D., Davydenko, K., Stenlid, J., Shabunin, D., Cleary, M., 2018. *Fraxinus excelsior* seed is not a probable introduction pathway for *Hymenoscyphus fraxineus*. For. Pathol. 48, e12392. https://doi.org/10.1111/efp.12392.

McNabb, K., Pike, C.C., 2019. A Nursery Guide for the Production of Bareroot Hardwood Seedlings. Agriculture Handbook 733. U.S. Department of Agriculture, Forest Service. Available at: https://rngr.net/publications/a-nursery-guide-for-the-production-of-bareroot-hardwood-seedlings.

Moraldi, M., 2023. Compendio di Vivaistica Forestale. ARSIAL Regione Lazio, Roma (Italy).

O'Reilly, C., Harper, C., Keane, M., 2002. Influence of physiological condition at the time of lifting on the cold storage tolerance and field performance of ash and sycamore. Forestry 75, 1–12. https://doi.org/10.1093/FORESTRY/75.1.1.

O'Reilly, C., Mortazavi, M., Keane, M., 2003. Effect of physiological status on the cold storage tolerance and field performance of ash. Oak Sycamore in Ireland. Phyt. - Ann. Rei Bot. 43, 335–350.

Peltier, J.P., Marigo, G., 1998. Drought tolerance of *Fraxinus excelsior*. Ecologie 29 (1/2), 399–402.

Piotto, B., Noi, D., 2001. Propagazione per seme di alberi e arbusti della flora mediterranea, ed. ANPA Agenzia Nazionale per la Protezione dell'Ambiente Dipartimento Prevenzione e Risanamento Ambientali.

Sambeek, J.W.V., Preece, J.E., 2007. In vitro propagation of *Fraxinus* species. In: Jain, S.M., Häggman, H. (Eds.), Protocols for Micropropagation of Woody Trees and Fruits. Springer, Dordrecht. https://doi.org/10.1007/978-1-4020-6352-7_17.

Suszka, B., Muller, C., Bonnet-Masimbert, M., 1994. Graines des feuillus forestiers. De la récolte au semis. INRA, Paris.

Takos, I., 1998. Seeds of woody plants (in Greek). http://apps.inforest.gr/sporoi.php.

Tani, A., Maltoni, A., Mariotti, B., 2007. Quality of Seedlings based on a Field Study. Field Performance. Sherwood - for. Ed Alberi Oggi, pp. 5–9.

Tani, A., Maltoni, A., Mariotti, B., 2008. Field performance: risultati su semenzali di frassino. Sherwood 145, 37–42.

Van Der Linde, S., Perez-Sierra, A., Needham, R.H., Combes, M., McCartan, S.A., 2021. Identification, detection and eradication of *Hymenoscyphus fraxineus* from ash (*Fraxinus excelsior*) seeds. Forestry 94, 745–756. https://doi.org/10.1093/FORESTRY/CPAB017.

Willoughby, I., Stokes, V., Kerr, G., 2009. Side shelter on lowland sites can benefit early growth of ash (*Fraxinus excelsior* L.) and sycamore (*Acer pseudoplatanus* L.). Forestry 82, 199–210. https://doi.org/10.1093/forestry/cpn054.

Zocca, A., 1999. La propagazione di alberi e arbusti, Edagricole. ed. Bologna (Italy).

Fraxinus ornus L.

Luna Morcillo[1], Daniel Kurjak[2], Cengiz Yücedağ[3], Nebi Bilir[4] and Alberto Vilagrosa[1,5]
[1]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain; [2]Faculty of Forestry, Technical University in Zvolen, Zvolen, Slovakia; [3]Burdur Mehmet Akif Ersoy University, Faculty of Engineering Architecture, Department of Landscape Architecture, Istiklal Campus, Burdur, Türkiye; [4]Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye; [5]Department of Ecology, University of Alicante, Alicante, Spain

Propagation

Seed propagation

Seeds of *Fraxinus ornus* L. (flowering ash) typically mature in October, but they can remain on the tree until January. The optimal time for seed collection is debated. Some authors suggest that seeds should be harvested while still green, as soon as they are fully developed but before they completely dry on the tree (McMillan-Browse, 1983). Others recommend collecting the seeds once they show a uniform brown color (del Campo et al., 2013). The species has high interannual variation in fruit production (i.e., a mast seeding habit), with some years yielding almost no production (García-Fayos et al., 2001). Harvesting can be done manually or by carefully stripping the seeds with a net spread on the ground. The seeds have a samara (i.e., winged achene) with a high moisture content. Therefore, seeds should either be sown immediately in a cold frame or dried in a shaded area until their moisture content reaches 5%, after which they can be tightly stored at −4°C to +4°C. Some studies recommend maintaining a moisture content of around 8–10% and storing the seeds at −4°C, which can preserve their viability for up to 10 years (Piotto and Piccini, 2000).

Similar to other *Fraxinus* species, the seeds of *F. ornus* show physiological dormancy. To overcome this dormancy, seeds are typically sown after a stratification period at about 20°C, followed by a second cold stratification period at 1 to 4°C. The duration of these stratification periods varies depending on the source (del Campo et al., 2013). In the nursery, sowing can be done in autumn without prior treatment, with germination occurring the following spring (Takos and Efthimiou, 2003). Alternating cold and warm temperatures, 5 to 10°C and 20 to 25°C, respectively may enhance germination (Piotto and Piccini, 2000). Yücedağ and Gezer (2007) reported high germination (75%) after a cold-wet stratification period of 1.5 months at +4°C in sand for 3 months. Piotto and Piccini (2000) cautioned that a constant temperature of 20°C could induce secondary dormancy, making late sowing inadvisable. Average seed length and width of *F. ornus* seeds are 19.4 and ~4 mm, respectively (Yücedağ et al., 2011; Bilir, 2019). Other main seed characteristics of *F. ornus* can be found in Table 11.14.

Table 11.14 Characteristics of seed lots of *Fraxinus ornus*.

Purity (%)	No. seeds/kg	Germination capacity (%)
90–98 (94)	27,000–55,500 (39,700)	60–80 (73)

Mean values in brackets.
Gradi (1989); Catalán (1991); García Fayos (2001); Piotto and Di Noi (2001); Yücedağ and Gezer (2007); Yilmaz and Tonguç (2013); Moraldi (2024).

Vegetative propagation

Fraxinus ornus is not usually propagated by vegetative methods. Although there are no reliable methods for rooting green cuttings, ash cultivars can be propagated by budding, grafting, and possibly layering (Hartmann et al., 1997). In vitro propagation through axillary shoot micropropagation, adventitious shoot organogenesis, or somatic embryogenesis has been proved in ash species (Sambeek and Preece, 2007). Arrillaga et al. (1992) developed a specific in vitro propagation protocol for juvenile and adult specimens of manna ash, using nodal segments and stem apices. Regarding acclimation and field performance, after in vitro conditions and the gradual exposure to reduced relative humidity over 2–4 weeks, survival of over 65% was reported for microplants (Arrillaga et al., 1992).

Seedling cultivation

Container plants

Fraxinus ornus can be produced in the nursery in one growing season; one- (1 + 0) or two-year-old (2 + 0) seedlings are the most common stock types. Given its tolerance to sunlight, the complete nursery cultivation process, which lasts 25–30 weeks from sowing to the final hardening phase, can be conducted outdoors under full sun, or under a maximum of 20% shade (del Campo et al., 2013). The recommended cultivation density for this species ranges from 250 to 400 plants/m^2 (Chirino et al., 2008).

The nursery period usually starts in early spring, ensuring frost is avoided. Two or three seeds are placed per container cell to ensure full tray occupancy. Germination can take several months. The most common container volumes range from 230 to 300 cm^3. Given the strong taproot of *F. ornus*, it is advisable to used containers of at least 15–18 cm in height to allow proper root development (Chirino et al., 2008). The growing medium must meet the general specifications for growing forest plants (Landis and Morgan, 2009). A mixture of blonde peat and coconut coir, with 5–10% vermiculite, is commonly used. *F. ornus* is insensitive to pH variations and can tolerate a growing medium with irrigation of pH of 6 or 7. Salinity should be below 1.5 10^3 mS/cm (del Campo et al., 2013).

Maintaining good moisture conditions in the substrate is crucial for obtaining quality seedlings. A thin layer (about 0.5 cm thick) of sand or perlite on top of each container cell helps retain moisture, which is especially important during germination. Fertilization should begin early in the cultivation process, with medium to high rates applied. If slow-release fertilizers are used, it is advisable to use a 15-8-12 N–P–K compound, at a dose of 2.5–3.5 kg/m^3. If fertigation is the chosen method, weekly applications of macro and micronutrients, like those proposed by Landis and Nisley (1990) for each cultivation phase, is appropriate.

No significant pests have been documented for *F. ornus*, so phytosanitary treatments in nurseries are mainly preventive or routine. Although some ash species are susceptible to *Hymenoscyphus pseudoalbidus*, a fungus causing widespread dieback of common ash (*F. excelsior* L.) and narrow-leaved ash (*F. angustifolia* Vahl) in Europe, *F. ornus* does not appear to be a natural host for the pathogen, as demonstrated by artificial inoculations on seedlings (Caudullo and de Rigo, 2016). Preventively, biweekly fungicide treatments with common active ingredients are recommended. Weeds can be effectively controlled by weeding every 6–8 weeks.

To our knowledge, there are no specific references regarding optimal morphological seedling quality parameters that promote superior field performance. However, we have gathered some data from experiments conducted across the species' distribution (Table 11.15).

Bareroot plant production

Bareroot cultivation of *F. ornus* is generally not recommended (del Campo et al., 2013), as the Mediterranean climate conditions under which this species is typically planted are

Table 11.15 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container and bareroot seedlings of *Fraxinus ornus*. The root collar diameter is measured immediately above the cotyledon insertion point. Observed values are mean values ± standard error.

Attributes	Container	Bareroot
Shoot height (cm)	12.5 ± 2.5	38.0 ± 0.81
Root collar diameter (cm)	0.44 ± 0.06	1.90 ± 0.49
Shoot dry weight (g)	1.00 ± 0.39	0.24 ± 0.12
Root dry weight (g)	2.13 ± 0.45	0.53 ± 0.26
Plant dry weight (g)	3.13 ± 0.82	0.77 ± 0.12
Shoot/Root dry weight ratio	0.43 ± 0.13	0.48 ± 0.35
Shoot [N] (mg/g)	12.2 ± 3.1	–
Shoot [P] (mg/g)	1.2 ± 0.2	–
Shoot [K] (mg/g)	7.8 ± 2.3	–

Radoglou and Raftoyannis (2001); Yücedağ and Gezer (2007); Bilir (2012); del Campo et al. (2013); Granados (2019).

unsuitable for bareroot stock. Experimental studies conducted with bareroot seedlings (Rahman et al., 2015; Yücedağ and Gezer, 2007), concluded that one-year-old bareroot seedlings did not meet the required standards. In comparison, container-grown seedlings of the same age and provenance exhibited superior traits, including greater root collar diameter and a higher number of roots than bareroot seedlings.

Field establishment

Fraxinus ornus has been mainly used in gardening and less frequently in reforestation projects. However, as with many species considered accessory or secondary in forest restoration, its use in reforestation has increased significantly since the 1990s (del Campo et al., 2013). Soil preparation is the same to that for other similar species (Gavinet et al., 2015; Granados et al., 2016). In Mediterranean areas, where water scarcity and shallow soils are common, planting holes of at least $0.4 \times 0.4 \times 0.4$ m are recommended, which can be smaller on wetter sites (Vilagrosa et al., 2008). Holes can be manually or mechanically excavated, but we recommend mechanical site preparation, preferably using an excavator or a spider excavator on steep slopes to minimize soil erosion.

Field performance of container *F. ornus* seedlings has been excellent, with nearly full survival rates in the first year after planting (del Campo et al., 2008; Bilir, 2022; Aparicio, 2010). In contrast, field performance of bareroot plants under drought conditions is poor, with survival rates of 45% eight months after outplanting (Rahman et al., 2015). Over time, however, mortality rates can vary significantly, depending on the suitability of the planting site. Some studies reported survival rates above 80% for the first 5 years after planting (Bilir and Seyedi, 2015; Tekocak and Bilir, 2015; Granados et al., 2016). Other trials conducted in Spain under unfavorable seasonal conditions, reported survival rates around 70% after 10 years (Morcillo et al., 2022). Conversely, minimum survival rates as low as 10% were reported 10 years after outplanting, following a sharp decline from the 84% survival achieved in the first year during the establishment period (del Campo et al., 2020). In experiments assessing seedling performance under tree cover in Aleppo pine forests (*Pinus halepensis* Miller), *F. ornus* demonstrated high shade-tolerance with higher survival rates compared with other species such as *Quercus pubescens* Willd., *Arbutus unedo* L., or *Ceratonia siliqua* L. that were negatively affected by high shade conditions (Gavinet et al., 2015).

Field growth rate of *F. ornus* presents intermediate values (del Campo et al., 2013), with a more pronounced development of the stem and aerial parts during spring (Flors, 2008). Results are high variable in literature, depending on the suitability of the planting site. Bilir (2019 and unpublished data) reported after 7 years, growth rates of 170 and 2.53 cm in height and diameter, respectively, in a plantation conducted under favorable conditions. Other authors reported heights after 10 growing seasons above 130 cm (Muzzi and Fabbri, 2007). However, much lower values have also been reported

between the third and the 10th year, such as those net growth rates of 7.9 and 1.2 cm, for height and diameter respectively, obtained by Granados et al. (2016) and Morcillo et al. (2022), or those that reported 0.8 cm of net growth in diameter, and even negative values in height (del Campo et al., 2020). The lowest values slightly improved when the plantation was implemented under the canopy of Aleppo pine forests, specially under a density of 800- to 1400 trees/ha (Morcillo et al., 2020).

The shade-tolerance of *F. ornus* was highlighted by Gavinet et al. (2015) when comparing its relative growth rate to other species (i.e., *Q. ilex, Q. pubescens, A. unedo*) and also in comparison with reforestation of *A. unedo* and *Sorbus domestica* L. under Mediterranean climate in the south of France (Helluy et al., 2021). Due to its demonstrated shade-tolerance, the use of tree shelters may enhance *F. ornus* seedling survival and growth by reducing desiccation and protecting from herbivory as previously observed in *F. angustifolia* (Klossas et al., 2013). However, the benefits of tree shelters may decrease as seedlings mature, and careful management will be required when removing shelters to avoid negative effects on the developing trees (Mechergui and Pardos, 2022).

First year irrigation is highly recommended during summer. Field experiments showed that higher survival rate of watered seedlings during the dry summer months was positively related to moisture availability (75% and 45% survival, respectively) and their greater root biomass, quality index, and number and length of first order lateral roots in relation to stressed plants without irrigation (Rahman et al., 2015). It should be noted that hermaphroditic individuals may be more drought-tolerant than male individuals (Verdú, 2004). However, seedlings sired by hermaphrodites showed reduced growth compared with those sired by males (Verdú et al., 2004), which may contribute to variability in plant production quality.

Chapter References

Aparicio, A.I., 2010. Estudio de una repoblación forestal en La Muela de Cortes (Valencia): análisis y evaluación de los resultados al año de plantación. Trabajo Profesional fin de Carrera. ETSIA. Universidad Politécnica de, Valencia.

Arrillaga, I., Lerma, V., Segura, J., 1992. Micropropagation of juvenile and mature flowering ash. J. Amer. Soc. Hort. Sci. 117, 346–350.

Bilir, N., 2012. Variation of Seedling Morphology in Manna ash (*Fraxinus ornus* L.). Seed Orchards and Breeding Theory Conference. May, Antalya, pp. 107–113.

Bilir, N., 2019. Morphological variation of seedling and seed characteristics in Taurus flowering ash. J. Bart. For. Facul. 21 (2), 525–533.

Bilir, N., 2022. Heritability of some seed characteristics in Manna ash. J. MAKU FEBED 13 (1), 129–133.

Bilir, N., Seyedi, N., 2015. Fourth year results in variations of Manna ash (*Fraxinus ornus* L.) families. RRBB 9, 436–440.

Catalán, G., 1991. Semillas de Łarboles y arbustos forestales. In: Instituto Nacional para la ConservaciŁon de la Ł Madrid, Spain.

Caudullo, G., de Rigo, D., 2016. *Fraxinus ornus* in Europe: distribution, habitat, usage, and threats. In: San-Miguel-Ayanz, J., de Rigo, D., Caudullo, G., Houston Durrant, T., Mauri, A. (Eds.), European Atlas of Forest Tree Species. Publ. Off. EU, Luxembourg e01435d+.

Chirino, E., Vilagrosa, A., Hernández, E.I., Matos, A., Vallejo, V.R., 2008. Effects of a deep container on morpho-functional characteristics and root colonization in *Quercus suber* L. seedlings for reforestation in Mediterranean climate. For. Ecol. Manage. 256 (4), 779–785.

del Campo, A., Aguilella, A., Lidón, A., Segura, G., 2008. Influencia del tipo y dosis de hidrogel en las propiedades hidrofísicas de tres suelos forestales de distinta textura. C. Soc. Esp. Cienc. For. 25, 137–143.

del Campo, A., Verdú, M., Prada-Sáez, M.A., 2013. *Fraxinus ornus* L. Producción y manejo de semillas y plantas forestales, vol 1, pp. 558–570.

Flors, J., 2008. Estudio del efecto de distintos tratamientos culturales (tubos protectores e hidrogeles) y de fertilización potásica en el establecimiento de brinzales forestales. Trabajo Profesional Fin de Carrera. ETSIA. Universidad Politécnica de, Valencia.

FRAXIGEN, 2005. Ash species in Europe: biological characteristics and practical guidelines for sustainable use. In: Oxford Forestry Institute, Department of Plant Sciences. University of Oxford, Oxford.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal de la Comunidad Valenciana. Conselleria de Medi Ambient, Generalitat Valenciana, Alboraya, Valencia, Spain. ISBN: 84-482-2934-7.

Gavinet, J., Vilagrosa, A., Chirino, E., Granados, M.E., Vallejo, V.R., Prévosto, B., 2015. Hardwood seedling establishment below Aleppo pine depends on thinning intensity in two Mediterranean sites. Ann. For. Sci 72, 999–1008.

Gradi, A., 1989. Vivaistica Forestale. Edagricole, Bologna, Italy.

Granados, M.E., Vilagrosa, A., Chirino, E., Vallejo, V.R., 2016. Reforestation with resprouter species to increase diversity and resilience in Mediterranean pine forests. For. Ecol. Manage. 362, 231–240.

Granados, M.E., 2019. Restauración de Bosques de *Pinus halepensis* con especies rebrotadoras para aumentar su resiliencia y capacidad de recuperación después de incendios forestales. PhD. University of Barcelona, Spain.

Hartmann, H.T., Kester, D.E., Davies Jr., F.T., Geneve, R.L., 1997. Plant Propagation - Principles and Practices, sixth ed. Prentice Hall, Upper Saddle River, NJ. 770 p.

Helluy, M., Gavinet, J., Prévosto, B., Fernandez, C., 2021. Influence of light, water stress and shrub cover on sapling survival and height growth: the case of *A. unedo, F. ornus* and *S. domestica* under Mediterranean climate. Eur. J. For. Res. 140, 635–647.

Klossas, G., Kyriazopoulos, A.P., Koukoura, Z., 2013. Post-planting treatments and shading effects in a *Fraxinus angustifolia* Vahl. silvopastoral system. Ann. For. Res. 56, 179–186.

Landis, T.D., Nisley, R.G., 1990. The Container Tree Nursery Manual (No. 674). US Department of Agriculture, Forest Service.

Landis, T.D., Morgan, N., 2009. Growing media alternatives for forest and native plant nurseries. In: Dumroese, R.K., Riley, L.E. (Eds.), tech. coords. National Proceedings: Forest and Conservation Nursery Associations-2008. Fort Collins, CO: US Department of Agriculture, Forest Service, Rocky Mountain Research Station, USA, pp. 26–31. Proc. RMRS-P-58.

McMillan-Browse, P.D.A., 1983. Hardy Woody Plants from Seed. CABI Datasets, 1979/1980. Grower Books, London, UK, p. 163.

Mechergui, T., Pardos, M., 2022. Tree shelters: a promising tool for environmental and livestock management. In: Natural Resources Conservation and Advances for Sustainability. Elsevier, pp. 309–325.

Moraldi, M., 2024. Vivaistica forestale - Conoscerla per praticarla con successo. Compagnia delle Foreste, Arezzo, 221pp.

Morcillo, L., Turrión, D., Soliveres, S., Chirino, E., Vallejo, V.R., Vilagrosa, A., 2022. Moderate pine cover maximizes 10-year survival and growth in late-successional species of contrasting functional strategies. For. Ecol. Manage. 509, 120098.

Muzzi, E., Fabbri, T., 2007. Revegetation of mineral clay soils: shrub and tree species compared. Land Degrad. Dev. 18, 441–451.

Piotto, B., Piccini, C., 2000. Dormienza, germinazione e conservazione dei semi dei frassini spontanei in Italia. Sherwood 52, 19–23.

Radoglou, K., Raftoyannis, Y., 2001. Effects of desiccation and freezing on vitality and field performance of broadleaved tree species. Ann. For. Sci 58, 59–68.

Rahman, M.S., Tsitsoni, T., Tsakaldimi, M., Ganatsas, P., 2015. Field performance of *Fraxinus ornus* bare-root plants to drought stress. In: Ivetić, V., Stankovid, D. (Eds.), Proceedings: International Conference Reforestation Challenges. 03-06 June 2015, Belgrade, Serbia. Reforesta, pp. 164–174.

Sambeek, J.V., Preece, J.E., 2007. In vitro propagation of *Fraxinus* species. In: Protocols for Micropropagation of Woody Trees and Fruits. Springer Netherlands, Dordrecht, pp. 179–192.

Takos, I.A., Efthimiou, G.S., 2003. Germination results on dormant seeds of fifteen tree species autumn sown in a Northern Greek nursery. Silvae Genet. 52, 67–71.

Tekocak, S., Bilir, N., 2015. Morphological variations in Manna ash (*Fraxinus ornus* L.) families: fifth year field test. App. Cell Bio. 4, 88–93.

Verdú, M., 2004. Physiological and reproductive differences between hermaphrodites and males in the androdioecious plant *Fraxinus ornus*. Oikos 105 (2), 239–246.

Verdú, M., Montilla, A.I., Pannell, J.R., 2004. Paternal effects on functional gender account for cryptic dioecy in a perennial plant. Proceed. Royal Soc. London. Series B: Biol. Sci. 271 (1552), 2017–2023.

Vilagrosa, A., Chirino, E., Bautista, S., Urgeche, A.M., Alloza, J.A., Vallejo, V.R., 2008. Proyecto de demostración de lucha contra la desertificación: Regeneración y plan de manejo de zona semiáridas degradadas en el término municipal de Albatera. Alicante. Cuad. Soc. Esp. Cienc. For. 28, 317–322.

Yilmaz, M., Tonguç, F., 2013. Dormancy level and dormancy-breaking pretreatments in seeds of *Fraxinus ornus* subsp. cilicica, an endemic to Turkey. Propag. Ornam. Plants 13 (1), 40–45.

Yücedağ, C., Gezer, A., 2007. The effects of different stratification periods on seed germination and transplanting on some morphological characteristics of flowering ash (*Fraxinus ornus* L.) seedlings. Tr. J. For. 1, 20–27.

Yücedağ, C., Gezer, A., Fakir, H., 2011. Variation of *Fraxinus ornus* subsp. cilicica (Lingelsh) Yalt in the lakes District of Turkey. Sci. Res. Essays 6 (22), 4788–4791.

Ilex aquifolium L.

Anastazija Dimitrova[1] and Juan Luis Nicolás Peragón[2]
[1]Ss. Cyril and Methodius University in Skopje, Hans Em Faculty of Forest Sciences, Landscape Architecture and Environmental Engineering, Skopje, North Macedonia; [2]Centro Nacional de Recursos Genéticos Forestales "El Serranillo", MITECO, Guadalajara, Spain

Propagation

Seed propagation

The fruits of *Ilex aquifolium* (holly) ripen in the fall, between mid-September and November, which is indicated by their color change from green to light red (Bonner, 2008; Nicolás Peragón and García González, 2012). Fruits can be harvested from mid-autumn to early spring, either by hand or by shaking the trees and collecting the fruits from sheets spread on the ground. If temporary storage is needed before seed extraction, overheating should be avoided. Seeds are extracted by macerating the fruits in water and subsequent blending, which does not damage the seeds. Aborted seeds and pulp remain floating on the water surface and can be easily removed (Nicolás Peragón and García González, 2012). Alternatively, seeds can be fermented in warm water and the pulp removed by hand-rubbing over a screen (Bonner, 2008). If the seeds are stratified immediately, they do not need to be dried. *I. aquifolium* has orthodox seeds that require airtight storage once cleaned and dried. Seed moisture content should be between 10% and 14%, with storage temperatures of 2–4°C. Under these conditions, seeds keep their germination capacity for 3–4 years (Bonner, 2008; Nicolás Peragón and García González, 2012). The quantitative characteristics of *Ilex aquifolium* seeds are presented in Table 11.16.

Germination is slow and irregular due to physical dormancy imposed by a woody seed cover and physiological dormancy from an underdeveloped embryo. Therefore, both dormancies must be removed, and embryo development needs to be stimulated to germinate seeds. Even after dormancy is broken, seeds may remain in a quiescence state for up to 3 years until exposed to suitable germination conditions (Arrieta and Suárez, 2004).

The seeds can be sown immediately after collection, but germination should not be expected until the second or third spring (Bonner, 2008). Many pregermination treatments have been evaluated, but they have proven only partially effective. Examples of these treatments include hot water, sulfuric acid, moist heat, temperature alternation between 3°C and 20°C in 16/8 h cycles for at least 28 days, warm stratification in peat for 12 months, cold stratification for 3 months, and warm stratification for 60 days at 20–30°C followed by cold stratification at 5°C. In vitro cultivation of isolated embryos has also been tested (Nicolás Peragón and García González, 2012; Pérez and Lancha, 2001).

Table 11.16 Characteristics of seed lots of *Ilex aquifolium*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
9–70 (40)	95–100 (98)	21,000–60,000 (40,500)	43–96 (70)

Mean values in brackets.
Nicolás Peragón and García González (2012).

Vegetative propagation

Vegetative propagation is possible using cuttings, grafting, micropropagation, and air layering (Nicolás Peragón and García González, 2012; Obeso, 1998). Cuttings should be taken from lateral and thin branches between late autumn to early spring. Avoid branches which grow horizontally. Cuttings need to be collected early in the morning when fully hydrated (swollen and firm) and stored in black polyethylene bags at 4°C to minimize drying. Cuttings should be processed within 24 h, following these steps: leaf removal (leaving only four to five apical leaves), a clean diagonal cut at the base, and two superficial opposite cuts (0.5 cm wide) along the last basal 5–10 cm. Rooting can be stimulated by treating the base of the cutting with 1% indole-3-acetic acid powder (Nicolás Peragón and García González, 2012).

Grafting techniques for *I. aquifolium* include split, English double split, and lateral (side) grafting. In vitro micropropagation has been done using buds from adult trees, inducing axillary sprouting with auxins (NAA and IBA) and cytokinins (BA and kinetin). The highest multiplication rates been achieved with 6-benzylaminopurine (Majada et al., 2000; Morte et al., 1991; Nicolás Peragón and García González, 2012). In addition, rooting has been attempted by dipping shoots in concentrated naphthaleneacetic acid and cultivated in half-diluted Murashige and Skoog's medium (Nicolás Peragón and García González, 2012).

Seedling cultivation

Container plants

Rooted cuttings are more commonly used for propagation due to the high dormancy of *I. aquifolium* seeds that complicates abundant and homogenous germination (Bonner, 2008). Cultivation of cuttings starts in a greenhouse in winter, where cuttings are rooted in a mixture of peat and perlite (1:1) at 20–25°C (minimum 12°C) with high air relative humidity (>80%). Trays are covered with plastic, and micro-spray irrigation is applied for 6 s every 15 min. Fungicides such as Captan (50%, 2 g/L) should be applied every 15 days. Rooting takes around 2 months (Nicolás Peragón and García González, 2012). Once rooted, cuttings require progressive acclimation for one additional month in a greenhouse and then are moved outdoors in spring to a shaded, wind-protected area. Moderate shading is especially important for the first growing season.

Fertilization should start early in spring and fertilized peat supplemented with 70–100 mg N by fertigation has resulted in high quality seedlings (Nicolás Peragón and García González, 2012). During cultivation, optimal watering is important. Container seedlings left outdoors in winter must be protected from strong frosts, as plug freezing can lead to mortality (Nicolás Peragón and García González, 2012).

If seeds are used to cultivate seedlings, stratified seeds should be sown in trays covered with polyethylene film and placed in a greenhouse under temperatures between 15–27°C. Emergence may be uneven, and emerged seedlings can be transplanted into final containers. Seed trays facilitate efficient management of plants (Nicolás Peragón and García González, 2012). Cultivation should be performed in containers with volumes of at least 300 mL, at densities between 200 and 300 plants/m^2. Only containers designed to prevent root spiraling should be used. For older seedlings (2 + 0 and 3 + 0), the container volume must be at least 400 mL (Nicolás Peragón and García González, 2012).

Bareroot plants

I. aquifolium can also be grown as bareroot stock using cuttings. They can be planted in rows spaced 15–20 cm. When using seeds they should be sown at a depth of 3–6 mm, with the seedbed mulched with needles or straw during the winter (Catalán, 1993). Common stock types are 1 + 0, 1 + 1, and 1 + 2 with average height between 10 and 40 cm (Logorelli, 2006). Depending on their use, bareroot plants may need three to 5 years to grow. Sowing and transplanting to a different bed can be done both in autumn and spring. Regardless of whether the plants are from seeds or cuttings, they need to be protected from wind and drought in the first year of their development. No particular pests and diseases have been noted for *I. aquifolium* during cultivation (Logorelli, 2006).

Field establishment

As *Ilex aquifolium* is not commonly used in forest plantations, knowledge of field establishment practices is limited. In Spain, it is mostly used as secondary species in afforestation or to diversify monospecific pine plantations. Planting densities are usually <300 plants/ha. In some cases, it has been planted as the main species at densities of around 1100 plants/ha (Nicolás Peragón and García González, 2012). Young plants need to be protected from herbivores, usually with some type of fencing or mesh-type tree shelters. Certain varieties of *I. aquifolium* have been shown to have a higher rate of survival in the field after transplanting. Using plants with large plugs could



Figure 11.6 Container *Ilex aquifolium* in the field. *(Pedro Villar-Salvador.)*

increase field survival (Fare et al., 1985). In southern Europe, under Mediterranean climates, soil preparation should be deep (>60 cm), preferably using a backhoe or subsoiling. For successful establishment, *I. aquifolium* requires high environmental humidity and abundant rainfall (minimum 600 mm); it does not tolerate dry seasons exceeding 1 month (Nicolás Peragón and García González, 2012). Some irrigation might be necessary during the summer on dry sites. Weeds need to be removed during the first 2 years to facilitate survival. If plants propagated from cuttings are used, it is recommended to keep the sex ratio of one male per 20 females, especially when planting in hedges (Appleton et al., 2009) (Fig. 11.6).

Chapter References

Appleton, B.L., Spivey, A.D., French, S.C., 2009. Virginia cut holly production: orchard layout and planting. Virginia Cooperat. Extens. 430–467. http://hdl.handle.net/10919/48115.

Arrieta, S., Suárez, F., 2004. Germination and seed bank depletion of holly (*Ilex aquifolium* L.) in four microhabitat types. Seed Sci. Res. 14, 305–313. https://doi.org/10.1079/SSR2004180.

Bonner, F.T., 2008. *Ilex* L. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual. US Department of Agriculture Forest Service, vol 727. Agriculture Handbook, Washington, DC.

Catalán, G., 1993. Semillas de árboles y arbustos forestales, fourth ed. ICONA, Madrid.

Fare, D.C., Gilliam, C.H., Ponder, H.G., 1985. Root distribution of two field-grown *Ilex*. Hortscience 20, 1129–1130.

Logorelli, C., 2006. Il genere *Ilex* e la seua specie aquifolium. Torsanlorenzo 8, 13–15.

Majada, J.P., Sánchez-Tamés, R., Revilla, M.A., Casares, A., 2000. Micropropagation of *Ilex aquifolium* L. Vitro Cellul. Developm. Biol. Plant 36, 521–526. https://doi.org/10.1007/s11627-000-0093-4.

Morte, M.A., Olmos, E., Hellin, E., Piqueras, A., 1991. Micropropagation of holly (*Ilex aquifolium*). Acta Horticult. 289, 139–140. https://doi.org/10.17660/ActaHortic.1991.289.33.

Nicolás Peragón, J.L., García González, D., 2012. *Ilex aquifolium* L. In: Producción y manejo de semillas y plantas forestales. Organismo Autónomo Parques Nacionales, Madrid.

Obeso, J.R., 1998. Effects of defoliation and girdling on fruit production in *Ilex aquifolium*. Funct. Ecol. 12, 486–491. https://doi.org/10.1046/j.1365-2435.1998.00216.x.

Pérez, J.C.C., Lancha, A.S., 2001. Manual Para la Identificación y Reproducción de Semillas de Especies Vegetales Autóctonas de Andalucía. In: Consejería de Medio Ambiente. Junta de Andalucía, Sevilla. Spain.

Juglans regia L.

Barbara Mariotti
Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy

Propagation

Seed propagation

Fruit production of *Juglans regia* L. may begin early, around 6 years, depending on environmental conditions, earlier in clones or cultivars selected for fruit production (Aletà Soler and Vilanova Subirats, 2012). The fruit is a dehiscent drupe with a green smooth involucre that ripens from August to October. The seed (nut) has a woody hard and thick endocarp, and the dispersal is mainly zoochorous (Castro et al., 2017; Zhang et al., 2017). The nut consists of shells joined by a suture seam and the shell is divided into four cavities. Nuts are harvested in the fall by shaking the branches or collecting fruits fallen naturally to the ground, in this case the period may extend into early winter. Harvesting is done manually and can be facilitated using nets or tarpaulins; the elimination of ground vegetation before harvesting can facilitate the collection. *J. regia* fruits are harvested when the external involucre splits, as soon as possible to avoid damaging the seed (Aletà Soler and Vilanova Subirats, 2012).

Seed extraction may be completed manually or using a shelling or husking machine (Aletà Soler and Vilanova Subirats, 2012). Seeds are preserved during the year following harvest; after natural drying, they should be kept at temperatures below 15°C with a maximum humidity of 60% (Aletà Soler and Vilanova Subirats, 2012). The seeds are suborthodox; to store them for a longer period, they should be dried to a moisture content of approximately 12% and kept at a temperature between 3 and 4°C (Aletà Soler and Vilanova Subirats, 2012). A suggested method to increase germination consists of dipping the seed in water for 2 weeks, replacing the water three times (Vahdati and Hoseini, 2005); otherwise, cracking, along with treatment by gibberellic acid (750 ppm), followed by chilling stratification has been successfully tested (Lamichhane et al., 2020). Seed purity is generally close to 100%; other seed characteristics are reported in Table 11.17.

Freshly collected seeds of *J. regia* can be sown directly. If stored, they should be soaked for 48 h before sowing or stratified for 1 month in humid cold sand (Aletà Soler and Vilanova Subirats, 2012). Germination is hypogeal and recently emerged seedlings

Table 11.17 Characteristics of seed lots of *Juglans regia*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
70–90 (85)	97–100 (99)	72–172 (121)	60–83 (71)

Mean values in brackets.
Aletà Soler and Vilanova Subirats (2012); Betti and Cappanelli (2009); Catalán (1991); Moraldi (2023); Navarro Cerrillo and Galvez (2001); Piotto (1992).

4–5 cm tall, have the first true leaves of an intense reddish color (Aletà Soler and Vilanova Subirats, 2012). The ISTA standards refer only briefly to this species in their guidelines for germination evaluation methods, specifically a tetrazolium test.

Vegetative propagation

J. regia is an important nut crop species in Europe and Asia; thus, genetic improvement for fruit production has promoted the use of vegetative reproduction to regenerate specific clones or other kinds of selected material. However, this species is not easily propagated vegetatively; the factors determining the success of different propagation techniques have been reviewed by Vahdati et al. (2022). Grafting and micropropagation are the most commonly used techniques.

To optimize grafting success, temperature should be maintained between 26 and 27°C and relative humidity between 80% and 95%; such conditions are crucial for callus formation at the graft section. Grafting success can be often hindered, however, due to problems in the root system caused by heavy irrigation or rainfall and low transpiration. An inadequate callus formation between the scion and rootstock is a primary reason for graft failure. Studies indicate that highly turgid cells are more likely to form calluses compared with wilting cells (Vahdati et al., 2022). As a result, the best environment for grafting this species is the greenhouse with the hot callus bench grafting procedure, that is, by a hot-callusing pipe that directs or localizes heat to the area of the graft union. Grafted trees are then subjected to controlled, elevated temperature and humidity. This technique is also useful to avoid breaking dormancy of the scion buds. Grafted plants are maintained at 25°C for 33 days.

Micropropagation can use different plant organs, such as zygotic embryos, shoots, branches, or adult plant scions (Jain and Häggman, 2007). Caulinary apex and axillary buds have produced plants with a good root system (Aletà Soler and Vilanova Subirats, 2012). The primary challenge with in vitro techniques is the presence of endogenous bacterial contamination and the exudation of phenolic compounds (Jain and Häggman, 2007; Vahdati et al., 2022). Using renewed material from consecutive pruning or grafting provides the opportunity to cultivate shoot tips with good growth (Jain and Häggman, 2007). Using annual growth shoot tips, meristems, and tips derived from epicormic sprouts has been reported as the most promising alternatives for transitioning explants from field conditions to in vitro environments (Jain and Häggman, 2007). A protocol for micropropagation of mature tissues of *Juglans regia* was accurately described by Jain and Häggman (2007), and the main factors affecting the success of this type of vegetative propagation were accurately reported by Vahdati et al. (2022), who provided a table summarizing several in-vitro rooting procedures including doses of plant growth regulators and dark (preinduction) and light (root expression) phases.

Despite the success of different phases of in vitro production and the good vegetative state of the obtained plants, the acclimatization of such plants at the nursery level is still the main limitation for forest nursery production (Aletà Soler and Vilanova Subirats, 2012). Propagation by cuttings has not produced satisfactory results yet, except in specific cases using root cuttings (Aletà Soler and Vilanova Subirats, 2012).

Seedling cultivation

Container plants

Seeds can be sown into small volume alveolate seeding trays and the seedlings transplanted into larger containers when they are about 5 cm tall. An alternative is to seed the nuts directly into the final cultivation containers if germination of the seed lot is high. Pruning the radicle before sowing is recommended if the nut has already germinated. Place the trays on an elevated support, such as a grid, which favors root air pruning, and, consequently, root-system fibrosity (Aletà Soler and Vilanova Subirats, 2012). The recommended container volume may vary according to the kind of desired stock type, from 0.7 to 6 L for 1–3 years of cultivation (1 + 0, 1 + 1, 2 + 0). In specific cases, such as for productive plantations, the branches can be pruned (Aletà Soler and Vilanova Subirats, 2012). Using a larger container can favor the growth of the taproot, which needs larger space for a proper development compared with other broadleaf species (Mariotti et al., 2015b). Very large containers (from 5 to 15 L) were successfully tested to produce 2-year-old seedlings for productive high quality timber plantations. This size was chosen to produce semiqualified seedlings in terms of height and branching and to reduce the rotation period (Mariotti et al., 2015a).

Grafted seedlings cultivated in containers should be fertilized with ammonium nitrate (NH_4NO_3) at a rate of 2–4 g N per container to promote the development of the photosynthetic apparatus (Akova et al., 2020). In 1-year-old seedlings cultivated for ornamental purposes, the use of humic acid at 4 mL/L and the bioregulator biohorm (ascorbic acid, citric acid, fulvic acid, free ammonium acid 20%, molybdenum 4%, and cytokinin) at 6 mL/L promoted the growth of all morphological traits (Taha, 2017). Plant growth-promoting rhizobacteria (i.e., *Bacillus cereus* L90) has been used to promote water stress resistance of seedlings (Liu et al., 2023).

Seedling height should range between 50 and 60–80 cm. In Spain, *J. regia* seedlings must be taller than 30 cm, while plants produced in vitro must be taller than 20 cm (Aletà Soler and Vilanova Subirats, 2012). If the seedlings are used for high quality timber plantations, the terminal bud should be lignified, the stem should be straight and have good apical dominance (Calvo and D'Ambrosi, 1995; Mariotti et al., 2015). Table 11.18 shows the morphological ranges together with the recommended values for 1 + 0 container *Juglans regia* plants (Fig. 11.7).

Table 11.18 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Juglans regia*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	49–107	>50	11–129	>40
Root collar diameter (cm)	1.05–1.77	>1.5	1.5–3.1	>1.5
Shoot dry weight (g)	12–147		1.5–31	
Root dry weight (g)	12–50		12.8–172	
Plant dry weight (g)	26–85		14.3–203	
Shoot/root dry weight ratio	0.36–1.56			

Akova et al. (2020); Calvo and D'Ambrosi (1995); Mariotti et al. (2015); Tani et al. (2007), Aletà Soler and Vilanova Subirats (2012).



Figure 11.7 *Juglans regia* seedlings after one growing season in a single pot. *(Barbara Mariotti.)*

Bareroot plants

In bareroot nurseries, sowing in the seedbeds should be superficial, placing the nut with the suture plane parallel to the soil surface at about 2 cm, covering the seeds with a thin layer of sand or nursery soil. Fall-winter sowings with untreated seeds are common, otherwise spring sowings with previously stratified seed are used. When pregermination treatments are carried out, it is advisable to sow when the shell valves start to open (Aletà Soler and Vilanova Subirats, 2012).

Weeding or using plastic mulch to prevent herbaceous competition is beneficial in the early stages of growth, especially for selected materials such as hybrid progenies (Aletà Soler and Vilanova Subirats, 2012). Protecting seeds from predation by rodents and birds until the seedlings reach a height of 10 cm is common practice; rodents and birds may take the nuts even after germination. The leaf concentration of photosynthetic pigments and consequently photosynthesis and plant vigor depend on soil fertility, which can be increased by fertilizer. Popović et al. (2015) observed that a high concentration of photosynthetic pigments was promoted by the microbiological preparation Bactofil B 10.

Bareroot seedlings can remain in the nursery field for 1–2 years. 1 + 0 seedlings should reach a minimum height of 30–40 cm, while two-year-old seedlings, which are on average 60–80 cm tall, should be transplanted after the first year to be 1 + 1 plants, although 2 + 0 seedlings are also marketed. *J. regia* develops a thick and deep taproot from the first growing season (Wani et al., 2016). Thus, it is crucial to avoid severe damage to the taproot during the lifting phase. Studies on the effects of different pricking-out practices have shown that removing the apex of the tap-root at germination significantly increases plant survival (Ciccarese, 1998, Aletà Soler and Vilanova Subirats, 2012).

Seedling quality grading based on size has been proposed by Calvo and D'Ambrosi (1995) and Tani et al. (2007). For productive plantations, 1 + 0 seedlings should have a minimum height of 30 cm, a root collar diameter greater than 0.5 cm, and height/diameter (H/d) ratio between 60 and 80 (Table 11.18). Seedlings that are less than 20 cm tall, and have a diameter <0.4 cm, or H/d ratio <20 or >90 should be discarded. Seedlings ranging within these thresholds can be used for other reforestation projects. However, such standards have not been updated but updated guidelines for productive plantations of valuable broadleaves, such as *J. regia*, requires larger seedlings (Buresti Lattes and Mori, 2016), at least a minimum height of 50–60 cm (Mariotti et al., 2015b) (Fig. 11.8).

Field establishment

J. regia has mainly been planted in the EU for timber production in afforestation of agricultural lands, mainly supported by public subsidies. Its use in traditional reforestation or forest restoration projects is uncommon. Growing practices useful for fruit production are not covered here.

J. regia does not tolerate waterlogging, making areas prone to periodic flooding unsuitable for this species. The species is also sensitive to late frosts, hence the selection of appropriate planting sites is crucial (Pelleri et al., 2021). A detailed table with the main ecological limits and site parameters for selecting suitable sites for productive plantations is provided by Pelleri et al. (2021). *J. regia* is also sensitive to competition for soil moisture, which can reduce both aerial and root biomass by up to 60% (Aletà Soler and



Figure 11.8 Outplanted 1-year old container seedlings of *Juglans regia* after one growing season in a field. *(Barbara Mariotti.)*

Vilanova Subirats, 2012). Therefore, managing weed and shrub competition is essential to reduce competition (Aletà Soler and Vilanova Subirats, 2012). Effective techniques include mechanized removal with chain or hammer mowers, chemical treatments where applicable, and manual stripping in grasslands.

J. regia has a robust taproot when young. As the plant grows, it develops early lateral roots that expand into a wide (up to 12–14 m) and shallow root network (Becquey, 1997). Therefore, soil preparation is critical for optimal root development. Deep cultivation can provide well aerated soil and break up the plow pan on previously cultivated land to facilitate root growth. Cross subsoiling is recommended for moderate slopes to improve soil structure and water infiltration (Aletà Soler and Vilanova Subirats, 2012). In addition, the removal of large root residues from previous plantings is important to reduce potential spread of *Armillaria mellea* (Aletà Soler and Vilanova Subirats, 2012).

The amount of liming needed to increase soil pH to the level required by the crop has been summarized in a table by Fernández-Moya et al. (2019), with recommendations according to the site pH. Composted manure should be applied at least 2 months before planting or placed at the bottom of the hole without direct contact with the roots (Becquey, 1997; Aletà Soler and Vilanova Subirats, 2006). However, fertilization should be based on soil analysis, as *J. regia* requires high N and K and moderate P (Fernández-Moya et al., 2019). The application of 100–150 kg/ha of P_2O_5/and 150–200 kg/ha of K_2O before the establishment of the plantation is recommended for productive plantations (Fernández-Moya et al., 2019), while Becquey (1997) recommends even higher values, around 150–200 kg/ha P_2O_5 and 300 kg/ha K_2O. Excessive N can hinder

growth, however, especially in wood production, so fertilization should be evaluated during the rotation cycle according to growth (Aletà Soler and Vilanova Subirats, 2012).

Due to the deep and large taproot, *J regia* it is generally planted as bareroot seedlings (Fernández-Moya et al., 2019), using 1- or 2-year-old seedlings. Innovative containers, with a minimum volume of 3 L, have provided very promising results in productive plantations (Mariotti et al., 2018; Pelleri et al., 2021). Direct seeding may be an option due to the high germinability of the seed. To protect the nuts from predation by rodents and other wildlife, the recommendation is to sow two or three nuts in a 30–40 cm plastic shelter and select the best seedling at the end of the growing season (Pelleri et al., 2021).

Planting is usually done manually, with holes prepared by hand or with an auger of more than 40 cm in diameter. Backhoes are rarely used (Aletà Soler and Vilanova Subirats, 2012; Becquey, 1997). The ideal time for planting is mid or early autumn for in vitro material. In regions with severe frost or waterlogged soils, planting should be postponed until late winter or early spring (Aletà Soler and Vilanova Subirats, 2012). Before planting, the roots of bareroot seedlings can be pruned to remove damaged or poorly shaped roots and to refresh the cut of the taproot and stimulate fibrosity. Deep planting, where the root collar is buried 5–10 cm below the soil surface, can delay plant growth by three to 4 years in heavy soils (Becquey, 1997).

J. regia generally grows well when isolated, so planting densities should be moderate. Recommended planting densities range from 100 to 300 trees/ha (Aletà Soler and Vilanova Subirats, 2012). In productive plantations, the trees are usually planted at their final spacing, which means that no thinning is planned. The spacing is typically wide, ranging from 7 × 10 m to 12 × 12 m between plants. In intensively managed plantations, *J. regia* can be planted at final spacings of 70–120 trees/ha (Aletà Soler and Vilanova Subirats, 2012). On low quality sites or if lower quality seedlings are used, narrower spacing may be chosen to allow future selection of the best trees, but not less than 5 × 5 m (Aletà Soler and Vilanova Subirats, 2012). Wildlife can cause significant damage to newly established plantations. To mitigate this, plants should be protected with shelter tubes at least 60 cm tall (Fernández-Moya et al., 2019). The shelters should have holes to facilitate air circulation around the plants. In addition, support stakes can be used to ensure that the trees grow straight and upright (Fernández-Moya et al., 2019). Further details on the establishment and management of *J. regia* plantations, including postplanting practices such as pruning and thinning, can be found in Pelleri et al. (2021) and in Fernández-Moya et al. (2019).

Chapter References

Akova, V., Dimitrova, N., Nacheva, L., Staneva, I., Vasilev, À., Gandev, S., 2020. Impact of nitrogen fertilization on growth and photosynthetic activity of walnut planting material (*Juglans regia* L.), cultivated in containers. Sci. Pap. Series B. Horticult. 64 (1), 15–20.

Aletà Soler, N., Vilanova Subirats, A., 2012. *Juglans* spp. In: Pemán Garcia, J., Navarro Cerrillo, R., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y manejo de semillas y plantas forestales. Organismo Autonomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio ambiente, Madrid, Spain, pp. 606–631.

Becquey, J., 1997. Les Noyers à bois. Institute pour le Développment Forestier (IDF), France.

Betti, A., Cappanelli, D., 2009. Le sementi Forestali. Manuale operativo per la produzione del germoplasma. In: Corpo forestale dello Stato - Centro Nazionale per lo Studio e la Conservazione della Biodiversità Forestale. Pieve Santo Stefano, Arezzo (Italy).

Buresti Lattes, E., Mori, P. (Eds.), 2016. Design, Implementation and Management of Naturalistic Permanent Polycyclic Tree Farms. Compagnia delle Foreste Srl.

Castro, J., Molina-Morales, M., Leverkus, A.B., Martínez-Baroja, L., Pérez-Camacho, L., Villar-Salvador, P., Rebollo, S., Rey-Benayas, J.M., 2017. Effective nut dispersal by magpies (*Pica pica* L.) in a Mediterranean agroecosystem. Oecologia 184 (1), 183–192. https://doi.org/10.1007/s00442-017-3848-x.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. Instituto Nacional para la Conservación de la Naturaleza. Ministerio de Agricultura, Pesca y Alimentación, Madrid, Spain.

Ciccarese, L., 1998. Establishment of *Juglans regia*. A four-year field comparison of direct seeding-containerised and bareroot nursery stock [Latium]. Monti Boschi 49 (3).

Fernández-Moya, J., Urbán-Martínez, I., Pelleri, F., Castro, G., Bergante, S., Giorcelli, A., Sansone, D., 2019. Silvicultural guide to managing walnut plantations for timber production. In: H2020 EU Project "Second Generation of Planted Hardwood Forests in the EU-Woodnat.".

Jain, S.M., Häggman, H. (Eds.), 2007. Protocols for Micropropagation of Woody Trees and Fruits. Springer Science & Business Media. https://doi.org/10.1007/978-1-4020-6352-7.

Lamichhane, S., Thapa, R., Thapa, P., Ahamad, K., 2020. Effect of different pre-sowing treatments on germination of Persian walnut (*Juglans regia* L.) in Rukum (East) District, Nepal. Tropical Agroecosyst. 2, 1165–1171. https://doi.org/10.26480/taec.01.2021.57.61.

Liu, F., Ma, H., Liu, B., Du, Z., Ma, B., Jing, D., 2023. Effects of plant growth-promoting rhizobacteria on the physioecological characteristics and growth of walnut seedlings under drought stress. Agronomy 13, 290. https://doi.org/10.3390/agronomy13020290.

Mariotti, B., Maltoni, A., Chiarabaglio, P.M., Giorcelli, A., Jacobs, D.F., Tognetti, R., Tani, A., 2015a. Can the use of large, alternative nursery containers aid in field establishment of *Juglans regia* and *Quercus robur* seedlings? New For. 46, 773–794.

Mariotti, B., Maltoni, A., Jacobs, D.F., Tani, A., 2015b. Container effects on growth and biomass allocation in *Quercus robur* and *Juglans regia* seedlings. Scand. J. For. Res. 30, 401–415. https://doi.org/10.1080/02827581.2015.1023352.

Moraldi, M., 2023. Compendio di Vivaistica Forestale. ARSIAL Regione Lazio, Roma (Italy).

Navarro Cerrillo, R.M., Galvez, C., 2001. Manual para la identificación] reproducción de semillas de especies Vegetales autóctonas de Andalucía. Consejería de Medio Ambiente. Junta de Andalucía, Córdoba, Spain.

Pelleri, F., Bidini, C., Manetti, M., 2021. La coltivazione del noce da legno (*Juglans regia* L.) (in Italian). I tempi della terra 10, 81–97.

Piotto, B., 1992. Semi di alberi e arbusti in Italia: come e quando seminarli. Società Agricola e Forestale (Grupo ENCC), Roma.

Popovic, V., Cirkovic-Mitrovic, T., Lucic, A., Rakonjac, L., 2015. Effect of fertilizers on concentration of photosynthetic pigments in leaves of one-year-old seedlings of walnut (Juglans regia L.). J. Agri. For. 61, 243–247. https://doi.org/10.17707/AgricultForest.61.4.28.

Taha, S.M., 2017. Effect of fertilization by biohorme and humic acid on (*Juglans regia* L.) seedling. J. Kirkuk. Univ. Agr. Sci. 8 (2), 1–12.

Tani, A., Adduci, M., Barbarotti, S., Maltoni, A., Mariotti, B., 2007. Comparison of *Juglans regia* L. bareroot nursery stocks for plantations: morphological characterization. Forest@ - Rivista di Selvicoltura ed Ecologia Forestale 4, 227–234. https://doi.org/10.3832/efor0454-0040227.

Vahdati, K., Hoseini, S.H., 2005. Introducing an innovative procedure for large commercial seed lots stratification in Persian walnut. Acta Hortic. 705, 355–357. https://doi.org/10.17660/ActaHortic.2005.705.49.

Vahdati, K., Sadeghi-Majd, R., Sestras, A.F., Licea-Moreno, R.J., Peixe, A., Sestras, R.E., 2022. Clonal propagation of walnuts (*Juglans* spp.): a review on evolution from traditional techniques to application of biotechnology. Plants 11 (22), 3040.

Wani, M.S., Hussain, A., Ganie, S.A., Munshi, A.H., Lal, E.P., Gupta, R.C., 2016. *Juglans regia*—a review. Int. J. Latest Res. Sci. Technol. 5, 90–97.

Zhang, H., Chu, W., Zhang, Z., 2017. Cultivated walnut trees showed earlier but not final advantage over its wild relatives in competing for seed dispersers. Integr. Zool. 12 (1), 12–25. https://doi.org/10.1111/1749-4877.12242.

Juniperus communis L.

Nebi Bilir
Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye

Propagation

Seed propagation

Juniperus communis (common juniper) can start producing berries at around 20 years, but in natural stands production usually is delayed until individuals are 50 years old (Gultekin, 2007). Berry maturation takes 18–19 months, with ripening beginning in October (Gultekin, 2007; Benito Matías et al., 2012). The optimal harvesting time falls between October and November. The berries have a width of 7–11 mm and contain one to three seeds (Gultekin, 2007; McCartan and Gosling, 2013). Berry harvesting is done manually by shaking branches. Harvested berries can be dried outdoors under sunlight for a few days. Seed extraction involves fragmenting the dried berries with oak ashes (a byproduct of burning oak wood) using a barley-breaking machine to separate seeds from other materials. To remove oil, resin, and chemicals that may hinder germination, the seeds must undergo the following treatments. First, they are immersed in a 5%–10% solution of oak wood ash for 1–3 days, followed by immersion in a 5000 mg/L citric acid solution for 1 day. The solution volume for both treatments must be at least 20 times the volume of the seeds. Afterward, they are rinsed in tap water under outdoor conditions (Gultekin, 2007). To remove empty seeds, they can be immersed in a 5000 mg/L NaCl solution for 5 days. Floating seeds must be discarded. Finally, rinsed seeds should be dried in the shade for 5 days (Gultekin and Bayav, 2005). This process yields approximately 35–60% filled seeds (Gultekin, 2014).

Seed dormancy in *J. communis* can be broken through a warm-cold stratification regime, starting with 15°C for 14 weeks and then 3°C for 12 weeks (Tylkowski, 2009). Germination is usually low, reaching around 40–50% in the best cases under nursery conditions (Gultekin and Bayav, 2005). Additional germination, up to 36%, can occur during the second and third springs following sowing (García, 2001). Germination rates vary depending on the showing date, decreasing from 62.5% in July to 0% in March (Gultekin and Bayav, 2005). Higher germination has been observed in soils with high organic matter and high silt + clay content (De Frenne et al., 2020). Cleaning seeds before sowing significantly enhances germination compared with sowing whole berries (Broome, 2003). This author evaluated several seed pretreatment methods to enhance germination. For all tested pretreatments, no germination occurred until the second spring after sowing. The highest germination rates (50–60% of viable, cleaned seeds) were achieved through a cool-moist stratification treatment at 0°C for 30–50 weeks or by outdoor burial for 60 weeks. Exposure to freezing temperatures of −13°C for 1 week during the winter following sowing reduced germination, with less than 30%

of viable seeds germinating in both treatments that included freezing. A warm stratification period at 20°C for 13 weeks did not improve germination. In another experiment using cleaned seeds, preliminary results suggest that treatment with a 1% citric acid solution or concentrated sulfuric acid, followed by 12 weeks in cool (4°C), moist conditions, significantly improved germination. The long cold period is likely necessary for embryo maturation, while acid treatments break the seed coat. After sowing, germination may take up to 12 months, although shorter times have been observed.

The weight of 1000 seeds ranges from 15.6 to 19.4 g, with an average of 17.1 g. Seed characteristics vary widely among populations (Houle and Babeux, 1994; García et al., 2000; McCartan and Gosling, 2013; De Frenne et al., 2020), as well as among individuals and years (Tylkowski, 2009). Some details of seed lots of *J. communis* are shown in Table 11.21.

Vegetative propagation

Vegetative multiplication is the main method for propagating *J. communis* in ornamental nurseries. Cultivars are multiplied by rooting cuttings under controlled temperature and humidity conditions. Although this technique is not used in forestry plant production, in marginal populations, which tend to have a low production of viable seeds, vegetative propagation could be suitable method for obtaining plants (Broome, 2003; Benito Matías et al., 2012). Cuttings from female plants root more easily than those from male plants. High indolyl-3-butyric acid concentration inhibits rooting of female cuttings (Houle and Babeux, 1994).

For propagation by cuttings, 10-cm-long hardwood cuttings taken in spring (February or March) are recommended, as opposed to softwood cuttings taken in autumn, which are more susceptible to fungal attack (Broome, 2003). Cuttings should be taken from the tips of the branches and should include the previous year's growth. They should be inserted into trays with a 1:1:1 mixture of peat, bark, and perlite without rooting hormone powder. Cuttings should be kept in a mist house with a 95–100% relative humidity for 12 weeks, with rooting typically starting after 6–8 weeks. After an additional 12 weeks, misting should be gradually reduced, so that by 16 weeks, plants can acclimate to ambient greenhouse conditions. In October, rooted plants should be potted and allowed to grow for another year before being planted out (Broome, 2003).

Table 11.21 Characteristics of seed lots of *Juniperus communis*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
0.4–16.0 (8.2)	90–100 (95)	56,000–120,000 (97,000)	7–75 (~45)

Mean values in brackets.
Piotto and Di Noi (2001); Gultekin (2007); Benito Matías et al. (2012).

Seedling cultivation

Cultivation of *J. communis* plants from seed is challenging, mainly due to its low and variable germination timing (Broome, 2003).

Container plants

According to experience in Türkiye, one- or two-year-old seedling can be produced in 2.8 L containers (11 × 30 cm). Larger 6 L containers (16 × 30 cm) are preferred for growing 1 + 1 or 1 + 2 seedlings. Sowing six pretreated seeds per cell or pot is recommended; however, pregerminating seeds in trays and transferring recently emerged seedlings to final cultivation containers is highly advisable. The growing medium usually consists in a mixture of 70:30 sand and organic matter with a pH of 6.0–7.5 (Gultekin, 2014).

In Spain, *J. communis* is mostly cultivated from seeds in 200–300 mL forest containers at cultivation densities of <400 seedlings/m^2 (Benito Matías et al., 2012). The most common stock type used in forest plantings is 2 + 0 container. Nurseries often use fertilized growing media, mostly blonde peat or coir mixed with vermiculite (80-20), or a mixture of peat, coconut fiber, and vermiculite (40-40-20). When nutrients are supplied via irrigation, recommended concentrations are 110 mg/L N, 28 mg/L P, and 120 mg/L K per irrigation event, with an optimal yearly nutrient supply per plant of 100–150 mg N, 30–40 mg P, and 75–140 mg K. If using slow-release fertilizer, Benito Matías et al. (2012) recommend a dose of 3 g/L of an NPK 17-5-12 formulation. Systemic fungicides should be applied during preemergence and emergence stages to prevent damping-off.

Seedling morphological attributes are shown in Table 11.20 (Gultekin, 2007; Anonymous, 1988). For 1 + 0 container seedlings, those with shoot heights below 6 cm and root collar diameters under 3 mm, are not recommended for plantings (Gultekin, 2007). In the United Kingdom, Broome (2003) recommends that plants cultivated from seeds

Table 11.20 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 seedlings of *Juniperus communis*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	7–15	>7	7–15	>7
Root collar diameter (cm)	0.11–0.2	>0.11	0.11–0.2	>0.11
Shoot/Root dry weight ratio	2–3	3>	2–3	3>

Anonymous (1988); Gultekin (2007).

should be grown in containers until they are a minimum of 20 cm tall before outplanting, which takes about 3–4 years. Plants obtained from cuttings and cultivated in 1.5 L containers can be outplanted when they are approximately 2 years old and 10 cm tall (Broome, 2003).

Bareroot plants

Pretreated seeds are sown at a 3–5 mm depth between August and October. The seeding rate in seed beds is 40–60 g/m^2 in seven rows, yielding 250–300 seedlings/m^2 (Gultekin, 2007). Per kilogram of seeds, 5000–10,000 seedlings can be cultivated. For forest plantings, 1 + 0 seedlings with a minimum shoot height of 7 cm and a root collar diameter of at least 0.1 cm are preferred (Gultekin, 2007) (Table 11.22). However, seedlings exceeding 15 cm height and 2 mm root-collar diameter are considered the highest quality; seedlings higher than 7 cm and 1.1 mm root-collar diameter are suitable for planting (Gultekin, 2007).

Field establishment

The most common method for establishing *J. communis* is planting nursery-cultivated plants. Direct sowing of berries (Broome, 2003) or seeds in the field is not recommended. According to experience in other *Juniperus* species such as *J. thurifera*, which coexists with *J. communis* in the Iberian Peninsula and France, we recommend subsoiling or mechanical holes (Nicolás Peragón et al., 2012).

Competition with other plants is the main issue for *J. communis* establishment, especially for plants <20 cm tall. An annual herbicide application, such as glyphosate at a rate of 0.05 L/m^2 around the plant's root zone, ensures good field performance (Broome, 2003). Alternatively, managers can also consider mechanical removal of weeds.

The planting season significantly affects the survival of plants propagated from cuttings. Plants derived from cuttings harvested in February and March and planted in May of the following year perform better than those from cuttings obtained in September and planted in July of the next year. According to Broome (2003), weather conditions at and after planting influence survival as much as the cutting's history.

Juveniles are frequently predated. When fencing is not an option, protective shelters help control browsing by rabbits, hares, and sheep. Short shelters (0.6 m tall) are recommended (Broome, 2003). In dry environments, shelters should be ventilated and allow relatively high light transmission (Benito Matías et al., 2012), as *J. communis* does not tolerate strong shading. Shelters must be removed before they collapse and disintegrate, which can occur after about five or more growing seasons (Broome, 2003).

Field fertilization does not increase the survival of *J. communis*, although growth improvement has been observed on poor sites (Broome, 2003). On dry sites, irrigation



Figure 11.9 Outplanted 2 + 0 Juniperus communis seedling after 1 year in field. *(Pedro Villar-Salvador.)*

is recommended, supplying 10–20 L per plant up to three times during the dry season (Benito Matías et al., 2012) (Fig. 11.9).

Chapter References

Anonymous, 1988. Seedling Quality Classification of Coniferous. Turkish Standard Institute Press, Ankara.

Benito Matías, L.F., Villar-Salvador, P., García Viñas, J.I., Gastón González, A., Prada Saéz, M.A., 2012. *Juniperus communis*. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de semillas y plantas forestales, Organismo Autónomo de Parques Nacionales, vol I. Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, Spain, pp. 632–646.

Broome, A., 2003. Growing juniper: propagation and establishment practices. Forest. Comisión Informat. Note 50, 1–12.

De Frenne, P., Gruwez, R., Hommel, P.W.F.M., De Schrijver, A., Huiskes, R.P.J., De Waal, R.W., Vangansbeke, P., Verheyen, K., 2020. Effects of heathland management on seedling recruitment of common juniper (*Juniperus communis*). Plant Ecol. Evolut. 153 (2), 188–198. https://doi.org/10.5091/plecevo.2020.1656.

García, D., 2001. Effects of seed dispersal on *Juniperus communis* recruitment on a Mediterranean mountain. J. Veg. Sci. 12 (6), 839–848. https://doi.org/10.2307/3236872.

Garcia, D., Zamora, R., Gomez, J.M., Jordano, P., Hodar, J.A., 2000. Geographical variation in seed production, predation and abortion in *Juniperus communis* throughout its range in Europe. J. Ecol. 88 (3), 436446. http://www.jstor.org/stable/2648450.

Gultekin, H.C., Bayav, A., 2005. The effects of different sowing times on germination percentage of Mountain juniper (*Juniperus communis* L. subsp. *nana* Syme), Small fruited juniper (*Juniperus oxycedrus* L. subsp. oxycedrus), Savin juniper (*Juniperus sabina* L.) seeds. Artvin Forest. Facul. J. Kafkas Univers. 6 (1–2), 102–112.

Gultekin, H.C., 2007. Nursery Practices of Turkish *Junipers*. General Directorate of Afforestation press, Ankara, Turkey.

Gultekin, H.C., 2014. Nursery Practices of Some Forest Tree Species. Research Institute of Poplar and Fast Growing Forest Tree Species (Izmit, Turkey).

Houle, G., Babeux, P., 1994. Variations in rooting ability of cuttings and in seed characteristics of five populations of *Juniperus communis* var. *depressa* from subarctic Quebec. Can. J. Bot. 72, 493–498. https://doi.org/10.1139/b94-066.

McCartan, S.A., Gosling, P.G., 2013. Guidelines for seed collection and stratification of common juniper (*Juniperus communis* L.). Tree Plant. Notes U.S. Dep. Agric. For. Serv. 56 (1), 25–29.

Nicolás Peragón, J., Benito Matías, L.F., García González, D., 2012. *Juniperus thurifera*. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de semillas y plantas forestales, vol I. Organismo Autónomo Parques Nacionales, Madrid, Spain, pp. 687–706.

Piotto, B., Di Noi, A., 2001. Propagazione per seme di alberi e arbusti della flora merditerranea. In: ANPA, Dipartimento Prevenzione e Risanamento Ambientale, Roma, Italy, p. 212.

Tylkowski, T., 2009. Improving seed germination and seedling emergence in the *Juniperus communis*. Dendrobiology 61, 47–53.

Juniperus excelsa M. Bieb.

Cengiz Yücedağ[1] and Georgi Hinkov[2]
[1]Burdur Mehmet Akif Ersoy University, Faculty of Engineering Architecture, Department of Landscape Architecture, Istiklal Campus, Burdur, Türkiye; [2]Forest Research Institute, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

The berry-like cones of *J. excelsa* are harvested manually from November to March (Gültekin, 2014). The diameter of these cones typically ranges between 5.4 and 14.4 mm, with an average value of 9.4 mm. Before seed extraction, the cones undergo sun drying and are fragmented using a barley crushing machine. The fragmented material is then sieved to separate cone scales from seeds. Given that a high proportion of seeds are empty (Table 11.19) they can be removed from seed lots by immersing them in a 25,000 ppm NaCl solution, which isolates the sound seeds (Yücedağ et al., 2010). The width and length of the seeds vary from 1.7 to 2.4 mm and from 4.9 to 6.5 mm, respectively with average values of 2.7 for width and 4.6 mm for length. Notably, the number of sound seeds is significantly and negatively correlated with the elevation of populations ($r = -0.77$) (Yücedağ, 2008). The average characteristics of seed and cone lots of *J. excelsa* are detailed in Table 11.19.

Seeds show both physical and physiological dormancy, needing specific treatments to break these dormancies. To address these limitations, seeds must undergo a 15-day immersion in water containing 20% oak wood ash, with the solution renewed every 5 days. Subsequently, the seeds should be dried for 5 days under shady conditions. Finally, the seeds must be immersed in a 5000-ppm citric acid solution for 15 days to remove oil, resin, and chemical materials present on the seed coats (Yücedağ et al., 2010).

For optimal germination, seeds need pretreatment involving warm stratification at 20°C for 30 days followed by cold stratification at 10°C for 30 days (Gültekin et al., 2006). This treatment results in the highest seed germination (66%) from seeds with a diameter of less than 2.5 mm. The typical dormancy of this species can significantly delay seedling emergence in nurseries if seeds are not pretreated. For instance, in a continental

Table 11.19 The average and range values of cone and seed lots of *Juniperus excelsa*. Seed viability is the proportion of live seeds per seed lot, determined by cutting method.

Proportion of sound seeds per cone (%)	Purity (%)	Seed viability (%)	No. seeds/kg	Germination potential (%)
18–94 (71)	95–100 (98)	9–11 (10)	17,544–31,250 (23,810)	5–66 (35)

Mean values in brackets.
Gültekin et al. (2003); Demir (2004); Mazur et al. (2004); Gültekin et al. (2006); Yücedağ (2008); Douaihy et al. (2011); Yücedağ and Gailing (2013b); Khajjak et al. (2016); Khajjak et al. (2017); Gülcü et al. (2019); Nikolova et al. (2020); Khan et al. (2022).

climate site in Bulgaria, only 5% of the seeds sown in November 2021 resulted in emerged seedlings 18 months later (Todorov and Georgieva, 2023).

Vegetative propagation

According to Iliev et al. (2015), woody cuttings harvested between September and October are typically utilized for rooting purposes, but those harvested in March and April yield a higher rooting capacity. Rooting can be enhanced by combining indole-3-acetic acid (IAA) and indole-3-butyric acid (IBA) hormones with fungicides such as benomyl. The optimal concentrations of these hormones are 5000 ppm IAA, 3000 ppm IBA, and 5000 ppm naphthaleneacetic acid (NAA) (Tektas et al., 2017).

Keskin (1991) indicated that rooting is positively correlated with the dose of IBA, while no correlation was observed with IAA. In addition, employing a growing medium comprised of sand (grain size ranging from 0.02 to 0.2 mm) supplemented with 200 ppm IBA under white light conditions, resulted in a rooting percentage of 37.3% (Khoshnevis et al., 2008).

Seedling cultivation

Container plants

Polythene bags, measuring 13 × 25 cm (volume 2 L) (Yücedağ and Gailing, 2013b) or 11 × 30 cm (volume 2.5 L) (Gülcü et al., 2010), are proper for the cultivation of container seedlings (Fig. 11.9). Seedlings are often cultivated in a growing medium comprising a mixture of soil and humus (1:1) (Gürlevik et al., 2014). The recommended sowing depth is 2 mm (Yücedağ and Gailing, 2013b). Sowing germinated seeds in containers after stratification is more cost-effective in nursery settings. Transplanting germinated seeds to the final cultivation container is ideally conducted in winter or early spring



Figure 11.9 One-year-old containerized (plastic bags) seedlings of *J. excelsa* in Eğirdir forest nursery in Türkiye. *(Cengiz Yücedağ.)*

Table 11.20 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container and bareroot seedlings of *J. excelsa*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Seedling height (cm)	16.8–33.8 (25.3)	≥12	11.9–22.0 (17.0)	≥12
Root collar diameter (cm)	0.19–0.26 (0.23)	≥0.2	0.14–0.28 (0.21)	≥0.2
Shoot dry weight (g)	0.5–1.3 (0.9)	Unknown	1.0–1.1 (1.1)	Unknown
Root dry weight (g)	0.4–0.9 (0.7)	Unknown	0.4–0.5 (0.5)	Unknown
Shoot/Root dry weight ratio	1.3–1.9 (1.6)	Unknown	1.4–3.0 (2.2)	Unknown

Recommended values are based on TS 2265/February standards (Anonymous, 1988). For container plants: Gülcü et al. (2010); Deligöz (2012); Yücedağ and Gailing (2013b). For bareroot plants: Gülcü and Gültekin (2005b); Eser and Gülcü (2019).

(Gültekin, 2007). Seed provenance has been identified as critical for the growth of *J. excelsa* seedlings with Tota and Söğüt being the fastest and slowest growing provenances, respectively (Gülcü and Gültekin, 2005b).

Irrigation generally has little effect on most seedling attributes. Over-irrigation may result in nutrient loss and does not necessarily produce larger or healthier seedlings. Thus, limited irrigation (9 mm water applied for 20 min) combined with a weekly fertilization using a water soluble 20-20-20 (N–P–K) compound fertilizer is a suitable option for the cultivation of container *J. excelsa* seedlings (Gürlevik et al., 2014). Table 11.20 presents observed and recommended values for high-quality container and bareroot seedlings of *J. excelsa*.

Bareroot plants

Nursery cultivation typically starts between November and March, with optimal sowing depth of 2 mm (Gülcü and Gültekin, 2005a). The recommended cultivation density is between 150 and 300 seedlings/m^2 with best results from 200 to 250 seedlings/m^2 (Eser and Gülcü, 2019; Fig. 11.10). For late winter or early spring sowings, mulching after sowing is recommended. Specifically, a mulch layer of 8 cm thickness using *Cedrus libani* A. Rich. cone scales is recommended, with the thickness reduced to 1 cm after germination (Gülcü and Gültekin, 2005a). Until the second week of May following germination, the recently emerged seedlings should be covered with a hessian cloth placed at a height of 50 cm to protect seedlings from birds and late spring frosts (Gültekin, 2014; Fig. 11.11).

The root collar diameter and height of seedlings are primarily influenced by growing density and fertilizer dose, respectively. Root pruning enhances the quality, root growth, and survival of *J. excelsa* seedlings (Darroudi et al., 2015). For this species, a nitrogen

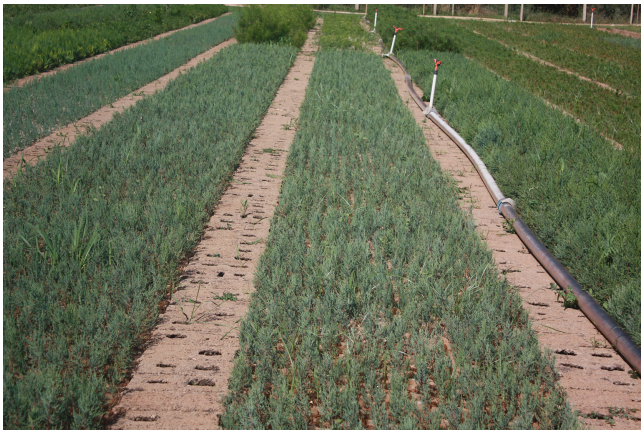


Figure 11.10 Seeding on seedbeds in Eğirdir forest nursery in Türkiye. *(Cengiz Yücedağ.)*



Figure 11.11 Early emerged *J. excelsa* seedlings with the hessian cloth cover removed in Eğirdir forest nursery in Türkiye. *(Cengiz Yücedağ.)*

fertilization rate of at least 10 g/m^2 (using ammonium sulfate) is necessary to produce high-quality seedlings. Increasing N fertilization to 20 g/m^2 results only in a small increase in growth while substantially increasing nursery costs. Consequently, we recommend a cost-effectiveness nitrogen fertilization rate of 10 g/m^2 (Eser and Gülcü, 2019).

Field establishment

Juniperus excelsa is an evergreen tree in the Cupressaceae family, distributed from South Crimea to the western Tien Shan and western Himalayas, and from Oman, Cyprus, Israel to North Macedonia, Bulgaria, Kosovo, and western Greece (Euforgen, 2023).

In Türkiye, it comprises approximately 82% of juniper forests (OGM, 2006), particularly in various regions of Anatolia, notably the Taurus and Anti-Taurus Mountains (Yaltırık, 1993; Yücedağ and Gailing, 2013a; Yücedağ et al., 2021a). The species is highly resistant to drought, frost, and poor soils (Douaihy et al., 2012; Yücedağ and Gailing, 2013a). Renowned for its durable timber (Pirani et al., 2011), landscape value (Zencirkıran, 2009), and medicinal properties (Nabi et al., 2017; Parvizi et al., 2018; Khanavi et al., 2019), it plays a crucial role in afforestation efforts, particularly in arid and semiarid lands of its distribution range (Yücedağ et al., 2021b).

When seeded directly in the field, the highest seed germination occurs at the canopy's edge and in the northern direction although the highest seedling survival occurs in the south side (Khoshnevis et al., 2019). Despite the higher germination and early survival under the canopy's edge, it is recommended to sow seeds outside of the tree canopy and in the northern direction, as seedling long-term survival under maternal trees is very low.

Gültekin et al. (2005) reported outplanting survival rates of 69–73% for 1 + 0 bareroot *J. excelsa* in the Lakes District of Türkiye. Container 1 + 0 seedlings had higher survival rates than bareroot 1 + 0 seedlings. While 1 + 1 and 1 + 0 container plants showed the highest survival, 2 + 0 seedlings cultivated in pots seemed to be the least successful (Örtel et al., 2020). The annual mean minimum temperature and soil texture at the populations' origin were the best predictors of growth of container seedlings after 10 years, accounting for 49% of the variation in growth (Yücedağ et al., 2021a). We recommend using 1 + 0 and 1 + 1 container seedlings in afforestation projects. If these are unavailable, then 1 + 0 bareroot seedlings can be planted. The optimal planting window for bareroot seedlings is just before they start growing in spring, typically between February and March, depending on weather conditions (Gültekin and Gültekin, 2007).

Mechanized soil preparation such as ripping with a bulldozer, is recommended over manual soil preparation (Örtel et al., 2020). In the first year following mechanized soil preparation, weed issues typically are minimal. Hoeing may be necessary in the first year, particularly in clay soils prone to cracking, while in subsequent years, it may not be required. Deep tillage in the first year can increase seedling establishment, especially in extremely arid regions (General Directorate of Forestry, 2006; Gültekin and Gültekin, 2007).

The *Ligustrum* witches' broom phytoplasma can be a significant disease affecting *Juniperus excelsa* (Çağlar et al., 2024).

Chapter References

Anonymous, 1988. TS 2265/February. Coniferous Tree Seedlings Standards. Turkish Standardization Institute, Ankara. (in Turkish).

Çağlar, B.K., Şimşek, E., Elbeaino, T., 2024. Ligustrum witches broom phytoplasma caused witches' broom disease for *Juniperus excelsa* Bieb. Turkey. J. Plant Dis. Protect. 131, 515–522.

Darroudi, H., Khosrojerdi, E., Shahabi, M., 2015. Effect of root pruning on physiology and morphology of Greek juniper seedlings (*Juniperus excelsa* M. Bieb.). J. For. Wood Prod. 68, 685–697.

Deligöz, A., 2012. Morphological and physiological differences between bareroot and container *Juniperus excelsa* seedlings. Turk. J. Agric. For. 36, 619–628.
Demir, P., 2004. A Study on the Practical Techniques to Offer Solutions for Germination Problems of Some Juniper Species Being Used in Afforestation in Turkey. Master Thesis. Ankara University, Ankara. (in Turkish).
Douaihy, B., Vendramin, G.G., Boratyński, A., Machon, N., Bou Dagher-Kharrat, M.B., 2011. High genetic diversity with moderate differentiation in *Juniperus excelsa* from Lebanon and the eastern Mediterranean region. AoB Plants 2011, plr003.
Douaihy, B., Sobierajska, K., Jasinska, A.K., Boratynska, K., Ok, T., Romo, A., Machon, A., Didukh, Y., Bou Dagher-Kharrat, M., Boratynski, A., 2012. Morphological versus molecular markers to describe variability in *Juniperus excelsa* subsp. *excelsa* (Cupressaceae). AoB Plants 2012, pls013.
Eser, Y., Gülcü, S., 2019. The effects of growing density and fertilization on morphological seedling characteristics of Crimean juniper (*Juniperus excelsa* Bieb.). Turk. J. For. 20, 15–19.
Euforgen, 2023. Map of *Juniperus excelsa*. https://www.euforgen.org/species/juniperus-excelsa/. (Accessed 13 June 2023).
General Directorate of Forestry, 2006. Action Plan for the Rehabilitation of Juniper Forests (2006–2015). Ministry of Environmental and Forestry, General Directorate of Forestry, Ankara, Turkey. (in Turkish).
Gülcü, S., Demir, S., Dirlik, S., 2019. Variations in cone and seed traits of Crimean juniper (*Juniperus excelsa* Bieb.) populations. Turk. J. For. 20, 187–194 (in Turkish).
Gülcü, S., Gültekin, H.C., 2005a. Determination of proper sowing techniques for Crimean juniper (*Juniperus excelsa* Bieb.) and small fruited juniper (*Juniperus oxycedrus* L.). J. Forestry Facul. Süleyman Demirel Univers. 1, 37–48 (in Turkish).
Gülcü, S., Gültekin, H.C., 2005b. Comparison of seed sources of Crimean juniper (*Juniperus excelsa* Bieb.) in the Lakes District in terms of morphological seedling quality criteria. J. Artvin Forest. Facul. Kafkas Univers. 6, 121–128 (in Turkish).
Gülcü, S., Gültekin, H.C., Çelik, S., Eser, Y., Gürlevik, N., 2010. The effects of different pot length and growing media on seedling quality of Crimean juniper (*Juniperus excelsa* Bieb.). Afr. J. Biotechnol. 9, 2101–2107.
Gültekin, H.C., 2007. Ecology and Silviculture Techniques of Turkish Juniper (*Juniperus* L.) Species, vol 27. TMMOB Chamber of Forestry Engineers Publication Nr 27, p. 136 (in Turkish).
Gültekin, H.C., 2014. Sapling production techniques of important forest trees (conifers, deciduous trees). Popl. Fast-Grow. Fore. Trees Res. Inst. Direct. Nr 271, p. 337 (in Turkish).
Gültekin, H.C., Çetin, M.E., Datumani, A., Ateş, M., Altınsoy, S., Divrik, A., Gültekin, Ü.G., Yiğci, M., Divrik, H., Sarılı, Z., 2005. Some studies on afforestation and artificial regeneration of Juniperus L. in Göller Region. Batı Akdeniz Ormancılık Araştırma Enstitüsü Dergisi 6, 42–60 (in Turkish).
Gültekin, H.C., Gülcü, S., Gültekin, Ü.G., Divrik, A., 2003. Studies on determination the effects of some practicable classification methods on seed germination of Crimean juniper (*Juniperus excelsa* Bieb.) before sowing process. J. Artvin Fores. Facult. Kafkas Univers. 1–2, 111–120 (in Turkish).
Gültekin, H.C., Gülcü, S., Gültekin, Ü.G., 2006. Studies on determination of separating empty seeds and the effects of seed classification on germination of Crimean juniper (*Juniperus excelsa* Bieb.). J. Fores. Facul. İstan. Univers. 56, 197–212 (in Turkish).
Gültekin, H.C., Gültekin, Ü.G., 2007. Silviculture techniques of juniper (*Juniperus* L.) species in Turkey. J. East. Medit. For. Res. Direct. 13, 69–111 (in Turkish).
Gürlevik, N., Deligöz, A., Yıldız, D., 2014. Effects of irrigation and fertilization on the growth of juniper seedlings. Aust. J. For. Sci. 131, 171–190.
Iliev, N., Milev, M., Alexandrov, P., Petkova, K., 2015. Production of Seedlings from Tree and Shrub Species. Publishing House at Forestry University, Sofia, 333 p. (in Bulgarian).
Keskin, S., 1991. Studies on the Possibilities of Rooting of Foetid-Odour Juniper and Crimean Juniper Cuttings, vol 233. Forestry Research Institute Publications, Technical Bulletin Series Nr, 30 p. (in Turkish).
Khajjak, M.H., Raza, A.M., Luqman, M., Yousafzai, A., Naudhani, S., Ali, G.M., Saeed, M., 2016. Evaluation of seed viability of *Juniperus excelsa* (M. Bieb.) found in Balochistan by tetrazolium assay. J. Appl. Emer. Sci. 6, 31–35.
Khajjak, M.H., Saeed, M., Shahwani, M.N., Ahmed, S.S., Shaheen, G., Yousafzai, A., Durrani, N.A., 2017. Seed and cone biometry of *Juniperus excelsa* from three provenances in Balochistan. Int. J. Biosci. 10, 345–355.

Khan, M.W., Tareen, A.K., Tareen, M., Sultan, I.N., 2022. Cone and seed characteristics of *Juniperus excelsa* (*M. Bieb*) in Balochistan province, Pakistan. Pure Appl. Biol. 11, 814–822.

Khanavi, M., Enayati, A., Ardekani, M.R.S., Akbarzadeh, T., Razkenari, E.K., Eftekhari, M., 2019. Cytotoxic activity of *Juniperus excelsa* M. Bieb. leaves essential oil in breast cancer cell lines. Res. J. Pharmacogn. 6, 1–7.

Khoshnevis, M., Korori, S.A.A., Teimouri, M., Matinizadeh, M., Rahmani, A., Shirvany, A., 2008. The effect of different treatments on rooting of *Juniperus excelsa* cutting. Res. Inst. Fores. Rangelands Iran 16, 158–167.

Khoshnevis, M., Teimouri, M., Hallaj, M.H.S., Matinizadeh, M., Shirvany, A., 2019. The effect of canopy and its geographic orientation on seeds germination and survival of *Juniperus excelsa* seedlings. Iran J. For. 11, 363–371.

Mazur, M., Boratynska, K., Marcysiak, K., Didukh, Y., Romo, A., Kosinski, P., Boratynski, A., 2004. Low level of inter-population differentiation in *Juniperus excelsa* M. Bieb. (Cupressaceae). Dendrobiology 52, 39–46.

Nabi, S., Kaleemullah, Kahraman, A.-Y.M.S.A., Tahira, B., Hajira, B., Rasool, A., Muhammad, A., 2017. A review on *Juniperus excelsa*: description, distribution and ecology, ethnobotany and biological activities. Indo Am. J. Pharm. Sci. 4, 636–644.

Nikolova, A., Tsvetanova, Z., Koyuva, L., 2020. Technical Manual with Information on Characteristics, Extraction, Processing, Sowing and Storage of Seeds of Target Tree Species under Project: LIFE16-NAT/BG/000817 "Restoration and Improvement of the Nature Protection Status of Priority Forest Habitats from the NATURA 2000 Network in Bulgaria", 88 p. (in Bulgarian).

OGM, 2006. Action Plan for the Rehabilitation of Juniper Forests (2006–2015). https://www.ogm.gov.tr/tr/e-kutuphane-sitesi/Yayinlar/Ard%C4%B1%C3%A7%20Ormanlar%C4%B1n%C4%B1n%20Rehabilitasyonu%20Eylem%20Plan%C4%B1.pdf.

Örtel, E., Kavgacı, A., Çalıkoğlu, M., Türkkan, M., Gültekin, H.C., Çobanoğlu, A., 2020. Artificial regeneration of Crimean juniper (*Juniperus excelsa*). Turk. J. For. Res. 7, 22–30 (in Turkish).

Parvizi, M.M., Khazaii, M.M., Moein, m., Zarshenas, M.M., 2018. *Juniperus excelsa* M.Bieb; a medicinal plant with various pharmacological activities. Trends Pharmaceut. Sci. 4, 1–8.

Pirani, A., Moazzeni, H., Mirinejad, S., Naghibi, F., Mosaddegh, M., 2011. Ethnobotany of *Juniperus excelsa* M. Bieb. (Cupressaceae) in Iran. Ethnobot. Res. Appl. 9, 335–341.

Tektas, İ., Türkoğlu, N., Çavuşoğlu, Ş., 2017. Effects of auxin doses on rooting of *Juniperus* L. Prog. Nutr. 19, 130–136.

Todorov, T., Georgieva, L., 2023. Personal communication about *Juniperus excelsa* at Lokorsko nursery, Sofia, Bulgaria.

Yaltırık, F., 1993. Dendrology Course Book I - Gymnospermae (Angiosperms). Istanbul University Forestry Faculty Publications, Istanbul (in Turkish).

Yücedağ, C., 2008. Studies on Genetic Variation of Seed and Seedlings of Some Crimean Juniper (*Juniperus excelsa* Bieb.) Populations in Turkey's Lakes District. Master Thesis. Süleyman Demirel University, Isparta. (in Turkish).

Yücedağ, C., Çiçek, N., Gailing, O., 2021a. Local adaptation at a small geographic scale observed in *Juniperus excelsa* populations in southern Turkey. iForest 14, 531–539.

Yücedağ, C., Ayan, S., Farhat, P., Özel, H.B., 2021b. *Juniperus* L. for restoration of degraded forest lands in Turkey. SEEFOR 12, 71–81.

Yücedağ, C., Gailing, O., 2013a. Genetic variation and differentiation in *Juniperus excelsa* M. Bieb. populations in Turkey. Trees 27, 547–554.

Yücedağ, C., Gailing, O., 2013b. Differences among *Juniperus excelsa* populations as revealed at morphological traits. Dendrobiology 70, 73–81.

Yücedağ, C., Gezer, A., Orhan, H., 2010. The genetic variation in Crimean juniper populations from the Lakes District of Turkey. Rom Biotechnol. Lett. 15, 5487–5492.

Zencirkıran, M., 2009. Determination of native woody landscape plants in Bursa and Uludağ. Afr. J. Biotechnol. 8, 5737–5746.

Juniperus oxycedrus L.

Nebi Bilir[1] **and Alberto Vilagrosa**[2]
[1]Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye; [2]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain

Propagation

Seed propagation

Juniperus oxycedrus (cade or prickly juniper) start berry production when they reach an age of 20–50 years in natural stands (Gultekin, 2007). Berry diameter ranges from 7 to 12 mm, with maturation taking 17–18 months. Each berry contains two to three seeds (Juan et al., 2003; Gultekin, 2007) (Fig. 11.13).

Deficient pollination and site conditions contribute to low seed cone production and seed viability in both subspecies (Ortiz et al., 1998). Optimal berry harvesting time is from September to November. Berries can be harvested manually or by gently shaking the branches to the ground. Harvested berries can be dried outdoors under sunlight for a few days. Seed extraction involves fragmenting the dried berries with oak ashes using a barley-breaking machine to separate seeds from other materials. To remove oil, resin, and chemicals that may hinder germination, the seeds must undergo the following treatments. First, they are immersed in a 10–20% solution of oak wood ash at room temperature for 3 days, followed by immersion in a 5000 mg/L citric acid solution at 4°C for 5 days. The solution volume for both treatments must be at least 20 times the volume of the seeds. Afterward, they are rinsed in tap water under outdoor conditions for 15 days (Gultekin and Bayav, 2005). To remove empty seeds, they can be immersed in a



Figure 11.13 Seeds of *Juniperus oxycedrus*. *(Nebi Bilir.)*

10,000 mg/L NaCl solution for 5 days. Floating seeds must be discarded. This process yields 60–80% filled seeds (Gultekin, 2007, 2014). Seeds can be stored for 10 years at 4°C.

Juniperus oxycedrus shows significant differences in fruit and seed characteristics across subspecies and sites (Juan et al., 2006; Gultekin, 2007; Vilagrosa et al., 2012). Some details of seed lots of *J. oxycedrus* are given in Table 11.23.

Published literature indicates that some kind of pretreatment is necessary to enhance germination. In most cases, cold stratification or a combination of cold and warm stratification is recommended. Piotto and Di Noi (2003) recommend sowing in the fall immediately after seed extraction without any pretreatment. However, for early spring sowing, Piotto and Di Noi (2003) recommend a warm stratification for 2–3 months followed by 3–4 months of cold stratification for the subspecies *J. oxycedrus* subsp. *macrocarpa*. Alternatively, warm stratification can be replaced by soaking seeds in sulfuric acid for 30 min.

Aouadj et al. (2022) showed that germination can be significantly improved by mechanical scarification with a scalpel (92% germination), cold stratification at 5°C for 30 days (86% germination), or a treatment with a concentrated (95%) sulfuric acid for 60 min (82% germination). Untreated seeds did not germinate. However, Juan et al. (2006) found that chemical scarification with different acids did not enhance seedling emergence compared with untreated seeds. These authors also observed that seeds from immature cones had higher germination rates than those from mature cones.

Under nursery conditions, maximum germination typically ranges from 60% to 80% (Gultekin, 2007) with significant differences depending on sowing time. Specifically, Gultekin and Bayav (2005) observed that seeds sown in summer, especially in August, or early fall show high germination in May of the next year, while those seeded in winter or early spring (March) show poor germination.

Vegetative propagation

Vegetative propagation is not commonly used for forest restoration. However, cuttings that include a woody section from the previous year's growth can be collected in summer or autumn for propagation (Cervelli, 2005).

Table 11.23 Characteristics of seed lots of *Juniperus oxycedrus*. Some germination values come from tetrazolium tests and consequently denotes germination potential.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
4–24 (15)	88–100 (97)	10,000–78,000 (37,000)	56–91 (73)

Mean values in brackets.
Juan et al. (2003); Gultekin and Bayav (2005); Gultekin (2007); Vilagrosa et al. (2012).

Seeds with the testa removed, when sown in a mixture of natural soil and peat under greenhouse conditions, do not germinate. Under in vitro conditions (1/3 strength Murashige and Skoog medium with 3% sucrose, with or without 0.5 g/L GA_3), germination is low (12%). However, isolated embryos achieved approximately 50% germination on the same medium (Cantos et al., 1998). Acclimation of in vitro plantlets in soil under greenhouse conditions was successful, with an 80% survival rate and an average height of 20 cm after 1 year (Cantos et al., 1998). The efficiency of explant decontamination varied by season, with the highest success observed in summer (57.3% survival) compared with spring and winter (Bertsouklis et al., 2023). Optimal in vitro performance was achieved in summer using Rugini Olive Medium (OM) supplemented with 0.1 mg/L 6-(γ,γ-Dimethylallylamino)purine (2iP), resulting in a 72% shoot formation rate, with an average of 3.1 shoots per explant reaching 1.4 cm in length (Bertsouklis et al., 2023).

Seedling cultivation

Container plants

Information on the container production of *J. oxycedrus* plants is scarce and few trials with this species are known. Here, we summarize the experience in Türkiye and Spain.

In Türkiye, one- and two-year-old seedlings are grown in 11 × 30 cm plastic bags, while 16 × 30 cm plastic bags are preferred for producing 1 + 1 or 1+2-year-old seedlings. Each bag is sown with six seeds. The recommended growing medium consists of 70–75% sand and 25–30% organic matter, with a pH of 6.0–7.5 (Gultekin, 2014).

In Spain, cultivation typically begins in large seed trays within greenhouses. As seedlings emerge, they are transplanted into their final containers. Timely transplantation is crucial to prevent root deformation. Like other *Juniperus* species, nursery growth is slow, so two or three-year-old seedlings are commonly used for forest plantings. These seedlings show advantages such as moderate size and a well-developed root system, resulting in a more compact plug (Vilagrosa et al., 2012).

The growing media typically consists of a mixture of amended and fertilized blonde peat combined with coir, perlite, vermiculite, or other materials such as composted bark. Compost from sewage sludge and dried olive pomace has also been used successfully as a substrate (Caravaca et al., 2006). Container sizes should be similar to those used for other Mediterranean species, ranging from 200 to 400 cm^3. Large volumes (300–400 cm^3) are recommended for plants cultivated for two or more years (Fig. 11.14).

Drought hardening improves fine root structure, potentially enhancing resource uptake efficiency, particularly for water and nutrients (Fonseca, 1999; Cortina et al., 2004). However, hardened seedlings do not show significantly higher field survival (Fonseca, 1999). The inoculation of mycorrhizal fungi from the genus *Glomus* has demonstrated positive effects on the growth and nutritional status of *J. oxycedrus* seedlings in nurseries



Figure 11.14 Cultivation of 2 + 0 container (plastic bags) *Juniperus oxycedrus* seedlings. *(Source: Nebi Bilir.)*

(Caravaca et al., 2006). Morphological characteristics of container seedlings are presented in Table 11.23.

Bareroot plants

Seeding density ranges from 30 to 50 g/m^2, with seeds distributed in seven rows per seedbed and sown at a depth of 4–6 mm between August and October. The final seedling density varies between 250 and 350 seedlings/m^2 (Gultekin, 2007). Each kilogram of seeds can produce between 5000 and 10,000 seedlings. In high-quality seed lots, germination rates can reach 81% when seeds undergo warm, wet stratification at 20–25°C for 60 days, followed by cold and wet stratification at 0–4°C for 15 days, and an additional wet stratification at 4–8°C for 60 days (Gultekin and Gultekin, 2003a, b).

Low temperature significantly influences germination in *Juniperus oxycedrus*, with germination initiating at 0–4°C and peaking at 5–10°C (Gultekin and Gultekin, 2003a, b). For forestry plantations, 1 + 0 bare-root seedlings are preferred (Gultekin, 2007). In southern Türkiye, the highest seedling performance was observed at a density of 189 seedlings/m^2 (17 × 3 cm spacing) among tested densities ranging from 63 to 600 seedlings/m^2 (Alim and Kavgaci, 2017). Morphological characteristics of *J. oxycedrus* bareroot seedlings are presented in Table 11.24.

Field establishment

Juniperus oxycedrus is usually planted as a secondary species with other commonly used species in forest restoration such as oaks and pines, at low densities not exceeding 400 individuals/ha. Direct seeding is not recommended due to the species' low and erratic germination rates.

In degraded semiarid areas of southeastern Spain, *J. oxycedrus* shows modest survival and growth in particularly dry years. However, under average rainfall years, survival

Table 11.24 Observed values for morphological attributes of 1 + 0 (bareroot) and 2 + 0 (container) plants as well as recommended values of high-quality seedlings of *Juniperus oxycedrus*.

	Container seedlings		Bareroot seedlings	
	2 + 0		1 + 0	
Attributes	Observed values	Recommended values	Observed values	Recommended values
Shoot height (cm)	8–48 (37)	>15	11.5–13.2	>7
Root collar diameter (cm)	0.16–0.65 (0.4)	>0.2	0.19–0.31	>0.2
Shoot dry weight (g)	1.80–4.70 (3.25)	–	0.6–1.4	–
Root dry weight (g)	1.30–3.3 (2.3)	–	0.4–1.1	–
Plant dry weight (g)	3.3–7.6 (5.5)	–	0.9–2.4	–
Shoot/root dry weight ratio	1.2–2.0 (1.6)	<3	1.2–1.5	<3

Anonymous (1988); Fonseca (1999); Caravaca et al. (2006); Gultekin (2007); Alim and Kavgaci (2017), and unpublished data by Vilagrosa.

ranges from 60% to 70% (Chirino et al., 2009; Padilla et al., 2009), comparable to those of *Juniperus phoenicea*, *Pinus halepensis*, and *Ceratonia siliqua*, and higher than *Chamaerops humilis* and *Quercus coccifera*. In semiarid environments, outplanting survival tends to be higher in northern exposed areas and in small stands with scattered pines. Under dry, more mesic conditions, *J. oxycedrus* show similar outplanting survival than other shrubs such as *Rhamnus lycioides*, *Rhamnus alaternus*, or even *Quercus ilex* with survival rates around 60%–70% after 5 years (Duchicela Moreira, 2024).

The most common soil preparation method involves planting in 40 × 40 × 40 cm holes (Fonseca, 1999). However, when feasible, more intensive and deeper mechanical preparations, such as 60 × 60 × 60 cm holes or 80 × 40 × 40 cm banks with microbasins (Saquete Ferrándiz et al., 2006) are recommended. The application of approximately 4 kg of sewage sludge per planting hole and the use of mesh shelters against herbivores can increase survival (Vilagrosa et al., 2012). For dry and semiarid regions, planting is recommended in late autumn.

Chapter References

Alim, E., Kavgaci, E., 2017. The effects of cultivation density on morphological characteristics of prickly juniper (*Juniperus oxycedrus* L. subsp. *oxycedrus*) seedlings in Eğirdir Forest Nursery. J. For. Res. 4, 1–11. https://doi.org/10.17568/ogmoad.309242.

Aouadj, S.A., Zouidi, M., Allam, A., Brahmi, M., Djebbouri, M., Nasrallah, Y., Hasnaoui, O., Nouar, B., Khatir, H., 2022. Preliminary study of the pre-germinative treatments of *Juniperus oxycedrus* L. and

Pistacia lentiscus L. in the Saida region (Western Algeria). Biodivers. Res. Conserv. 67, 13–20. https://doi.org/10.2478/biorc-2022-0011.

Anonymous, 1988. Seedling Quality Classification of Coniferous. Turkish Standard Institute Press, Ankara.

Bertsouklis, K., Paraskevopolou, A.T., Petraki, E., 2023. In vitro regeneration from adult node explants of *Juniperus oxycedrus*. Not. Bot. Horti Agrobot. Cluj-Napoca 51 (1), 13062. https://doi.org/10.15835/nbha51113062.

Cantos, M., Cuerva, J., Zárate, R., Troncoso, A., 1998. Embryo rescue and development of *Juniperus oxycedrus* subsp. *oxycedrus* and *macrocarpa*. Seed Sci. Technol. 26, 193–198.

Caravaca, F., Aiguacil, M.D., Diaz, G., Marin, P., Roladan, A., 2006. Growth and nitrate reductase activity in *Juniperus oxycedrus* subjected to organic amendments and inoculation with arbuscular mycorrhizae. J. Plant Nutr. Soil Sci. 169, 501–505.

Cervelli, C., 2005. Le specie arbustive della macchia mediterranea. Un patrimonio da valorizzare. Colla Siciliana Foreste 26. Azienda regionale Foreste Demaniali, Palermo, Italy.

Chirino, E., Vilagrosa, A., Cortina, J., Valdecantos, A., Fuentes, D., Trubat, R., Luis, V.C., Puértolas, J., Bautista, S., Peñuelas, J.L., Vallejo, V.R., 2009. Ecological restoration in degraded drylands: the need to improve the seedling quality and site conditions in the field. In: Grossberg, S.P. (Ed.), Forest Management. Nova Science Publishers, Inc, pp. 85–158.

Cortina, J., Bellot, J., Vilagrosa, A., Caturla, R.N., Maestre, F., Rubio, E., Ortiz de Urbina, J.J., Bonet, A., 2004. Restauración en semiárido. In: Vallejo, V.R., Alloza, J.A. (Eds.), Avances en el estudio de la gestión del monte mediterráneo. Fundación CEAM, Generalitat Valenciana - Bancaja, Valencia. Spain, pp. 345–406.

Duchicela Moreira, K.L., 2024. Solapamiento entre espacios mineros y zonas de alto valor para la biodiversidad y potencial de la restauración ecológica: un caso de restauración minera en España. Master Science Thesis. University of Alicante, Spain.

Fonseca, A.D., 1999. Manipulación de las características morfo-estructurales de plantones de especies forestales mediterráneas producidas en vivero. Master Science Thesis. CIHEAM-IAMZ, Zaragoza, Spain.

Gultekin, H.C., Gultekin, U.G., 2003a. Improvement of seed characteristics and stratification and effects of some chemicals on germination of *J. excelsa, J. foetidissima, J. oxycedrus*. J. For. Hunting 2, 33–40 (in Turkish).

Gultekin, H.C., Gultekin, U.G., 2003b. A research on seed-break dormancy of *J. excelsa, J. oxycedrus and J. phoenicea*. J. Daph. 2, 16–18 (in Turkish).

Gultekin, H.C., Bayav, A., 2005. The effects of different sowing times on germination percentage of Mountain juniper (*Juniperus communis* L. subsp. *nana* Syme), Small fruited juniper (*Juniperus oxycedrus* L. subsp. oxycedrus), Savin juniper (*Juniperus sabina* L.) seeds. Forest. Facul. J. 6 (1–2), 102–112.

Gultekin, H.C., 2007. Nursery Practices of Turkish *Junipers*. General Directorate of Afforestation of Türkiye press, Ankara, Türkiye.

Gultekin, H.C., 2014. Nursery Practices of Some Forest Tree Species. Research Institute of Poplar and Fast Growing Forest Tree Species. Izmit, Türkiye.

Juan, R., Pastor, J., Fernández, I., Diosdado, J.C., 2003. Relationship between mature cone traits and seed viability in *Juniperus oxycedrus* L. subsp. *macrocarpa* (Sm.) Ball (*Cupressaceae*). Acta Biologica Cracoviensia. Series Botanica 45, 69–78.

Juan, R., Pastor, J., Fernández, I., Diosdado, J.C., 2006. Seedling emergence in the endangered *Juniperus oxycedrus* L. subsp. *macrocarpa* (Sm.) Ball in Southwest Spain. Acta Biolog. Cracovien. Series Botan. 48, 49–58.

Ortiz, P.L., Arista, M., Talavera, S., 1998. Low reproductive success in two subspecies of *Juniperus oxycedrus* L. Int. J. Plant Sci. 159, 843–847. https://www.jstor.org/stable/2475156.

Padilla, F.M., Ortega, R., Sánchez, J., Pugnaire, F.I., 2009. Rethinking species selection for restoration of arid shrublands. Basic Appl. Ecol. 10 (7), 640–647.

Piotto, B., di Noi, A., 2003. Seed propagation of mediterranean trees and shrubs. In: APAT - Agency for the Protection of the Environment and for Technical Services. Roma, Italy.

Saquete Ferrándiz, A., Lledó, M.J., Escarré, A., Ripoll, M.A., de Simón, E., Lledó Solbes, M.J., Escarré Esteve, A., Ripoll Morales, M.A., De-Simón Navarrete, E., 2006. Effects of site preparation with micro-basins on *Pinus halepensis* Mill. afforestations in a semiarid ombroclimate. Ann. For. Sci. 63 (1), 15–22. https://doi.org/10.1051/forest.

Vilagrosa, Carmona, A., García Viñas, J.I., González, G.,A., Prada, Sáez, M.A., 2012. *Juniperus oxycedrus* L. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales, pp. 647–663.

Larix decidua Mill.

Kateřina Houšková[1], Daniel Kurjak[2] and Peter Petrík[3, 4]
[1]Faculty of Forestry and Wood Technology, Mendel University in Brno, Brno, Czech Republic; [2]Faculty of Forestry, Technical University in Zvolen, Zvolen, Slovakia; [3]Institute of Forest Botany and Forest Zoology, Technical University of Dresden (TUD), Tharandt, Germany; [4]Karlsruhe Institute of Technology (KIT), Institute of Meteorology and Climate Research—Atmospheric Environmental Research (IMK-IFU), Garmisch-Partenkirchen, Germany

Propagation

Seed propagation

Larix decidua Mill. (European larch) is an important managed species and is the only *Larix* species native to Europe (CABI Compendium, 2022). It flowers from March to April (Hoffman et al., 2005) and in May in higher elevations, simultaneously with the growth of needles (Kantor et al., 1975). If growing without competition, flowering begins at the age of 15–20 years, while trees growing in stands start flowering at the age of 40–50 years (Hoffman et al., 2005). Flowering at low elevation locations is frequent and abundant with peaks every 3–5 years. In mountain areas flowering peaks occur every 6–10 years. Cones are collected from December to March (Vincent, 1965). They can be collected from standing or felled trees in natural population or by harvesting cones in seed orchards (Fig. 11.15) established for seed production (Palátová, 2008). *L. decidua* seeds are also collected from the snow by hand (Shearer, 2008) or in late winter from canvas spread on the ground and trees are shaken to release the seeds (Rudolf, 1974).

After collecting, the cones are stored in dry, ventilated, cool rooms in temporary warehouses. Depending on their moisture, cones need to be turned over regularly. A hectoliter (hL) of undried fresh cones weighs 25–35 kg (Gordon, 1992). If cone weight/hL is more than 35 kg the thickness of the cone layer should be less than 25 cm and cones should be stirred once every 2 days. If the weight/hL is between 30 and 35 kg the thickness of the cone layer should be no more than 40 cm and cones



Figure 11.15 Seed orchard of *L. decidua* at the University forest Enterprise Masaryk forest in Křtiny, Czech Republic. *(Kateřina Houšková.)*

should be stirred once every 4 days. Finally, if the hL weight is less than 30 kg the layer should be less than 50 cm and cones must be mixed once a week (Vincent, 1965).

Larix decidua seeds can be extracted from the cones by a combination of hot air and mechanical methods (Chrz et al., 1951). Vincent (1965) recommended extraction only when cone water content fell below 15%. If the cones are insufficiently dried the edges of the scales are difficult to abrade during mechanical extraction, leading to longer extraction times and increasing seed damage. In some countries such as in the Czech Republic hot-air seed extraction is performed in multistory ovens with floors separated by hinged perforated grates (Palátová, 2008). Fresh cones are first placed on the upper floor, where they are predried for about 8 h at 30°C. Then they are moved to the middle floor with an average temperature of 40°C. Finally, cones are moved to the lowest floor and placed for 15 h in a rotating drum made of wire mesh, in which the seeds are released from the open cones. The freed seeds fall through the perforated drum to the floor. With this extraction system, only about 60% of the seeds are recovered, mainly due to the anatomical structure and shape of the scales. In the past, several crushers were used to loosen the seeds remaining in the cones, but seeds were mechanically damaged. At present, a special shaker is used to release seeds from cones. Cones tumble in the drum and their scales rub against each other releasing the remaining seeds. With proper adjustment, the percentage of mechanically damaged seeds should not exceed 1% (Palátová, 2008).

Extracted seeds must be free of impurities, partially dewinged, and cleaned (Vincent, 1965). The wings are firmly attached to the seed and their complete removal can damage the seed. Therefore, only the part of the wing that exceeds the circumference of the seed is broken off, and this is done under dry conditions with the help of thumb or brush wing removers (Palátová, 2008). To remove impurities such as dust, wing fragments, and empty or undeveloped seeds, some cleaner machines use pneumatic methods. Since the weight of full and empty seeds is small, we recommend removing empty seeds by floating in water. Full seeds absorb water, increase their weight and sink, while empty seeds float and can be separated. Palátová (2008) considered that seeds need from 20 min to 24 h to soak and sink. In the first 20 min, only cone fragments and mechanically damaged seeds settle; after 24 h even empty seeds begin to settle. Solutions with density between that of full and empty seeds are also used for seed sorting by flotation. For instance, seeds can be floated in alcohol (90% to absolute) for a maximum of 5 min, with full seeds sinking and the empty seeds floating (Palátová, 2008). Considering the necessity of mechanical extraction of *L. decidua* seeds and the possibly significant proportion of mechanically damaged seeds, following the Prevac method to remove empty, damaged seeds is recommended (Lestander and Bergsten, 1985).

Larix decidua seeds are orthodox and remain viable for at least 3 years (CABI Compendium, 2022). Table 11.25 presents the average characteristics of *L. decidua* seed lots. The ideal seed storage conditions are in airtight containers at a temperature around

Table 11.25 Seed characteristics of *Larix decidua*.

Cone to seed yield (%)	Purity (%)	No. seeds/kg	Germination capacity (%)
1.2–7.2 (4.5)	70–94 (82)	154,000–229,000 (194,200)	35–40 (43)

Mean values in brackets.
Gordon (1992); Hoffman et al. (2005); Shearer (2008).

0–4°C (Hoffman et al., 2005). Suitable seed moisture for storage is 6–8% for long-term storage (10 years). However, it is important to ensure a stable storage temperature (Hoffman et al., 2005) as fluctuations of even ±1°C can negatively affect the quality of the seeds. Machaníček (1980) recommended determining the time of storage according to seed quality based on the accelerated aging test (stress test), where germination capacity is tested after exposing the seeds to a temperature of 40 °C and a relative humidity of 100% for 96 h. If germination is reduced only by 1%, the seeds can be stored for a long time. If germination decreases by 3%, seeds can be stored for a maximum of 5 years, and if the decrease is >3%, seeds should be sown immediately.

L. decidua seeds are not dormant, and no presowing treatment is necessary (Bezecný et al., 1992). However, to homogenize germination, Chrz et al. (1951) recommended soaking the seeds in water before sowing, mainly after long-term storage. Hrabí (1989) recommended soaking the seeds for 24 h in water at 17–20°C and then drying them for 24 h. This will not only speed germination but also will remove empty seeds (Palátová, 2008). Although it is not necessary, cold stratification in a moist medium for 1 month can also speed and homogenize germination (Plants for a Future, n.d).

Vegetative propagation

L. decidua is readily propagated by seed, but seed supply is scarce, due to irregular flowering and a low percentage of filled seed (Kosinski, 1986; Owens, 1995), so vegetative propagation could be an option. Vegetative propagation by rooting softwood or hardwood stem cuttings is possible (Lehotský, 1964; Mason, 1989; Weisgerber and Sindelar, 1992; Li and Wyckoff, 1994). Tissue culture is also possible, either by somatic embryogenesis or in vitro organogenesis (Lelu et al., 1994; Benkrima and Aderkas, 1995, Chalupa, 2000). These techniques are quite recent and not yet widely adopted but are gaining increasing significance as breeding programs for European and other fast-growing *Larix* species intensify globally. Grafting is also used in horticulture (Vidakovic, 1991) and to establish clonal seed orchards. Grafting can be carried out in spring (March) or in late summer (August) on seedlings no older than 2 years (Sheat, 1957). The most successful techniques for *L. decidua* are side and splice grafting (Lehotský, 1964).

Seedling cultivation

Nursery growth of *L. decidua* is fast with 2-year-old bareroot plants and 1-year-old container seedlings the most planted stock types (Mauer et al., 2013). Observed and recommended dimensions of 1-year-old container seedlings are presented in Table 11.26. Bezecný et al. (1992) recommended older, 3-year-old transplants on sites with strong weed competition such as clearcuts after disturbances. Average seed yield of *L. decidua* planting material in the Czech Republic is 33,000 seedlings and 18,900 transplants/kg of seeds (Cafourek and Kotrla, 2022). This agrees with the data from Slovakia (Šmelková, 2009), though the final yield is influenced by several factors such as seed quality, cultivation technology, seed age, soil preparation method, and sowing date. Aldhous and Mason (1994) stated that the minimum dimensions for *L. decidua* material suitable for planting are 20 cm tall and 4 mm stem collar diameter for both container and bareroot cultivation (Table 11.26).

Container plants

L. decidua container plants used in forest plantations are typically cultivated in one growing season (Mauer et al., 2013; Fig. 11.16). It is possible to cultivate seedlings with shoots up to 50 cm tall in one growing season (Table 11.26). Sowing in the nursery must be carried out in early spring (the optimum time in Central Europe is mid-March) and the sown containers must be placed in greenhouses during seedling emergence. Exceptionally, 1-year-old and several-year-old bareroot and container seedlings can be transplanted into containers to obtain older container plants (Mauer et al., 2013; Table 11.27; Fig. 11.16). To avoid root deformation, seedlings must be cultivated in containers of the correct size: for seedlings up to 25 cm tall, the upper container diameter and height of 4 × 12 (min. 10) cm is recommended; for plants up to 50 cm tall, diameter

Table 11.26 Observed morphological and physiological attributes and recommended values of high-quality 1-year-old seedlings of *Larix decidua*.

Attributes	Observed values	Recommended values
Shoot height (cm)	15–50	>20
Root collar diameter (cm)	0.19–0.34	>0.3
Root:shoot ratio	0.1–1.3	>0.5
Length of tap root (cm)	13–20.6	>10
Leaf [N] (mg/g)	14.9–31.1	>19.5
Leaf [P] (mg/g)	0.3–2.3	>1.4
Leaf [K] (mg/g)	0.8–5.2	>5
Leaf [Ca] (mg/g)	1.2–9.4	>3
Leaf [Mg] (mg/g)	0.4–2.3	>1

Aldhous and Mason (1994); Pâques (1994); Cornelissen et al. (1997); Kloeppel et al. (1998); Mason et al. (2004); Dauer et al. (2007); Leski et al. (2008); Jurásek et al. (2012); Modrzyński et al. (2015); Fajardo and Siefert (2016); Repáč et al. (2017); Plesa et al. (2019).

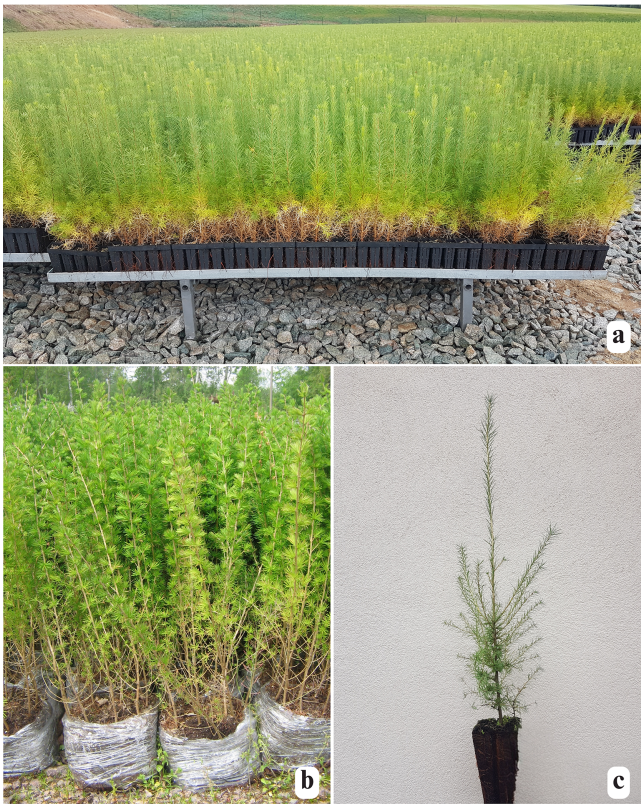


Figure 11.16 (a): 1-year-old *Larix decidua* container plants at the holding area (LESCUS Cetkovice company, Czech Republic). (b): 2-year-old *L. decidua* container plants (Lesoškolky company, Czech Republic). (c): 2-year-old *L. decidua* container plant (Dyk's nursery of the University forest Enterprise Masaryk forest in Křtiny, Czech Republic). *(Kateřina Houšková.)*

and height of 8 × 18 (min. 15) cm.; for plants up to 80 cm tall, diameter and height of 12 × 18 (min. 15) cm. For larger plants up to 120 cm tall, an upper container diameter and height of 15 × 30 (min. 26) cm is recommended and with anti-spiraling structures (vertical ribs and lateral perforations) to prevent root deformations (Jurásek et al., 2012). Specific containers may be required; for example, in the Czech Republic, only tested containers may be chosen from a catalog published by the Forestry and Game Management Research Institute, which is updated annually (Jurásek et al., 2006).

The optimum growing medium is 50% loam or sandy loam soil, 20–30% organic material (compost or pine bark), and 20–30% sand or perlite (Dirr and Heuser, 2009). The growing medium pH is particularly important for *L. decidua* and should range from pH 4 to 5 (Aldhous and Mason, 1994). Irrigation and fertilizing rates are not as specific as for other conifers. However, due to its high growth rate it is necessary to apply slightly higher doses to ensure proper growth. Fertilization can begin soon after seedling emergence. In early spring, seedlings demand high amounts of N, P, K, Ca and Mg, but the maximum demand is at the end of summer. Therefore, these macronutrients are

supplied through slow-release fertilizers (Mauer et al., 2013). Usually 1.5 kg of slow-release fertilizer with an action period of 3–4 months and 1.5 kg of slow-release fertilizer with an action period of 5–6 months are supplied to each 1 m^3 of growing medium before filling the containers. A typical formulation of slow-release fertilizer is N = 16%, P_2O_5 = 8%, K_2O = 12%, MgO = 2.2%, B = 0.01%, Cu = 0.015%, Fe = 0.3%, Mo = 0.01%, Zn = 0.012%.

Bareroot plants

Seeds are usually sown in spring, when the soil temperature exceeds 10°C. Autumn sowing (September and later) is uncommon (Mauer et al., 2013). Seeds are covered with a 0.5 cm layer of material, usually sand (Dušek et al., 1970). Broadcast sowing at a rate of 30–60 g/m^2 using seed lots with 80% germination capacity is recommended for sowing directly into soil beds (Dušek et al., 1970). A rate of 55 g/m^2 can be used for sowing into prepared substrates (Bezecný et al., 1992). Broadcast sowing can yield up to 500 seedlings/m^2 in beds and 200 seedlings/m^2 when cultivation lasts 2 years (MLVH, 1977; Aldhous and Mason, 1994). For 147 cm-wide beds, the sowing rate should result in 450 1-year-old seedlings/m of bed for spring transplanting, 375 seedlings for summer transplanting (2-year-old seedlings), or 300 seedlings for undercutting (Bezecný et al., 1992).

One-year-old bareroot seedlings are rarely used for forest regeneration due to their small size. Consequently, *L. decidua* bareroot plants are usually cultivated for 2 years. However, in some places such as the United Kingdom 1-year-old bareroot stock can be used (Aldhous and Mason, 1994). A primary tap root usually develops during the first year of growth (Köstler et al., 1968) and seedlings are commonly undercut at the beginning of their second growing season (Mauer et al., 2013). Dušek et al. (1970) recommended undercutting at a depth of 6–8 cm. Mauer et al. (2013) recommended the depth be determined based on the root size of a sample of lifted seedlings. The diameter of the root at the point of undercutting should not exceed 6 mm, with up to 1/3 of the root system removable, and at least 3 cm of the tap root excised to encourage root multiplication at the cut point. Without undercutting, seedlings develop disproportionately large shoots compared with their root systems.

In addition to undercutting, seedlings can also be transplanted, but these plants typically show reduced grow and consequently are smaller. As L. *decidua* sprouts early in spring, undercutting or transplanting must be completed before sprouting and once the soil temperature exceeds 5°C (Mauer et al., 2013). Therefore, *L. decidua* is often the first tree to be processed in nurseries. The recommended row spacing for transplanting is 21 cm, with 7.5–10 cm between seedlings in the row for one additional year of cultivation, or 12.5 cm for two additional years (Dušek et al., 1970).

Fertilization of bareroot *L. decidua* follows the nutrient schedule of container-grown stock, adjusted by soil fertility. *L. decidua* shows a strong response to N, so typically application rates should be reduced by 50%–66% by omitting one application of the top dressing (Aldhous and Mason, 1994). The recommended annual dose of pure N or N-based fertilizers is 100 kg/ha (MLVH, 1977), while for pure K it ranges 35–75 kg/ha depending on the maturity of the plants (Aldhous and Mason, 1994). Deficiencies lead to reduced growth, weak and stunted plants, and visible symptoms on the assimilation organs, such as changes needle yellowing (Mauer et al., 2013). P deficiency, however, is specifically manifested in *L. decidua* and *Picea abies* by a blue-green to gray–green color of the needles (Mauer et al., 2013).

L. decidua does not require any special irrigation; the timing and amount should be adjusted based on crop needs. It is advisable to implement protective measures against abiotic and biotic factors either preventively or as needed (Mauer et al., 2013). Drought stress can be easily mitigated with operative irrigation. The negative effects of heatwaves can be reduced by implementing basal or overhead shading (Helgerson, 1990) and additional irrigation due to high evaporative demand. Regular monitoring and early detection of biotic stressors is critical for timely mitigation action. Infected plants should be removed and disposed properly to prevent the spread of pathogens. Fungicides can be applied to manage Larch needle cast disease (*Mycosphaerella laricina, Lachnellula willkommii*) in nurseries (Jalkanen 2016). Insecticides can be used to mitigate larch sawfly (*Pristiphora erichsonii*) or aphids (Holmes and MacQuarrie, 2016). Moreover, biological control agents such as parasitoid wasps or biopesticides (e.g., *Bacillus thuringiensis*, *Beauveria bassiana*) can be implemented for integrated pest management of *L. decidua* (Bravo et al., 2011; Li et al., 2013; Skrzecz et al., 2024).

Field establishment

L. decidua occurs naturally in four discontinuous areas of Europe: the Alps, Carpathian Mountains, Sudeten Mountains, and parts of Poland. It is found both in pure and mixed stands in Austria, Czech Republic, Slovakia, Slovenia, southeastern France, southern Germany, Poland, northern Italy, Romania, Switzerland, and northwestern Ukraine (Ostenfeld and Larsen, 1930; Weisgerber and Sindelar, 1992; Schmidt and McDonald, 1995). Natural regeneration is successful in clearcut areas (CABI Compendium, 2022) where seeds come from the edge of the stand or from reserved trees (Bezecný et al., 1992). *L. decidua* can also regenerate under shelterwood systems on nutrient-rich and moist habitats, although seedlings must be released quickly by final felling (Bezecný et al., 1992). It is a suitable species for forest regeneration in challenging areas, including preparatory stands for afforestation of agricultural land, postfire areas, peat bogs, and waste dumps (Mauer et al., 2013).

Planting seedlings is the most common regeneration method (Bezecný et al., 1992). From a silviculture point of view, *L. decidua* is different from other conifers due to its early, rapid, and sustained growth in height, diameter, and volume. This high growth rate, combined with their competitiveness, requires intensive management (Cook, 1969). *L. decidua* is often used for replanting in cases of high mortality in previously established plantations (Mauer et al., 2013).

Successful plantation establishment requires preparation of the site to control competing vegetation (CABI Compendium, 2022). Mauer (2009) suggested keeping the soil weed-free until planting by maintaining a sufficient canopy of a regenerated stand. In the case of weeding, chemical (herbicide) or mechanical intervention (soil scarification) is recommended. Planting can be conducted in spring or autumn, and even in winter in mild climates (Mauer et al., 2013). Spring planting can be done when the soil thaws and soil temperature exceeds 5°C, soil moisture is between 50% and 70%, and frost risk is low. Because L. *decidua* sprouts early, dormant seedlings should be planted early, before other forest trees. Planting sprouted seedlings can result in poor field performance. Autumn planting, from mid-October until the beginning of frost season, is recommended only for dormant, bareroot plants with lignified terminal shoots and completed growth (Mauer et al., 2013). As seedlings are leafless, they will not transpire, which reduces transplanting shock.

The most effective planting technique is hole planting. For container stock, specialized planters such as a hollow or half-hollow spade can be used depending on the shape and size of the plug (Mauer et al., 2013). On light soils, slit planting with a planting bar can be used (Bezecný et al., 1992).

The plantation density of *L. decidua* varies depending on the management objectives (CABI Compendium, 2022). Bachasson (1982) recommended 2500 plants/ha, which is also the minimum density requirement for planting in the Czech Republic (Decree No. 456/2021Coll.). Wider initial spacing is advantageous as it allows thinning to be delayed until the trees reach a commercial size (CABI Compendium, 2022). Carter and Selin (1987) concluded that an initial stocking density of 1000 plants/ha (approximately a 3 × 3 m spacing) was optimal for most objectives. This spacing results in lower planting costs, shorter rotations, and large branch and stem diameter. French foresters recommend a low planting density of 1600 plants/ha and expect to develop a broadleaved subcanopy later (Parde, 1957). Generally, lower planting densities are acceptable for container stock compared with bareroot stock and for older or larger plants (Mauer et al., 2013).

Reforestation by direct seeding is rarely practiced. Ančák (1960) recommended sowing in spring, although occasionally it can be done in autumn. Tichý (1949) recommended sowing in non-weedy areas or after site preparation, in square patches with sides of 30–60 cm, spaced 1.2–1.3 m (1.5 m in mountain regions). The soil should be loosened in autumn before spring sowing. In the spring, these areas should be slightly loosened

again, and a pinch of seed (10–12 seeds) sown and covered with a thin layer of loose humus (approx. 2 cm). Tichý also recommended sowing around stumps but noted that sowing in clearcut areas was frequently unsuccessful, as additional sowing in case of initial failure was impossible. Therefore, sowing should be limited to specific cases like low accessibility areas where transportation of seedlings would be very challenging and expensive.

Pure L. *decidua* stands do not provide good soil cover, an issue only partially mitigated by the annual needle fall (Bezecný et al., 1992). Parde (1957), however, recommended *L. decidua* monocultures, arguing that assistance from other species to improve soil conditions is not required, and that hardwood species are incompatible with successful *L. decidua* cultivation. On fertile sites, however, Parde (1957) suggested introducing a subcanopy to maintain good soil quality such as the broadleaf *Fagus sylvatica* or the conifer *Abies alba*. *L. decidua* requires a free growing crown, which is best achieved in a mixture with other woody species (Bezecný et al., 1992). As a result, *L. decidua* is commonly found in mixed stands, either in group-like formations or individually mixed with other trees, particularly spruce and pine. Tichý (1949) recommended against planting *L. decidua* near *Picea abies*, as it suffers from shading and root competition at later stages. In contrast, Bezecný et al. (1992) recommended mixtures of *L. decidua* and *Picea abies*, which can be transformed later into pure *L. decidua* stands. The highest-quality stands are two-story, with *L. decidua* forming the upper layer, and *F. sylvatica* or *Tilia* spp. in the lower layer (Bezecný et al., 1992). *L. decidua* is sometimes used as a secondary species in admixture with other species due to its deep and extensive root system, which provides good anchorage and contributes to stand stability (Kacálek et al., 2017).

Weeding is only necessary during the first two to 3 years, until the seedlings overtop the surrounding shrubs (Cook, 1969). To avoid using herbicides, Mauer (2009) recommended mowing, trampling, mulching, or hand pulling weeds, depending on the weed species. Watering with home prepared biocides, such as a water solution from *Urtica dioica* macerate, is usually ineffective. Ungulates damage both terminal and lateral shoots, primarily by dislodging them with their antlers. The appropriate protection method, whether chemical, biological, or mechanical—is selected based on the species of game and its population density. The most reliable form of protection is to enclose the plantation with fencing (Mauer, 2009).

Fertilization of L. *decidua* plantations is not a common practice. However, Möhl et al. (2019) demonstrated a positive effect of fertilization on growth in the Swiss Alps (2083–2225 masl); NPK applied at a rate of 15 kg/ha N increased annual increment by 10–20% compared with unfertilized plots. A higher dose of 30 kg/ha N did not produce additional benefits.

Chapter References

Aldhous, J.R., Mason, V.L. (Eds.), 1994. Forest Nursery Practice. Forestry Commission 111. HMSO, UK, 268 p.

Ančák, J., 1960. Forest seed management, forest nurseries and forest establishment (in Slovak). Poverenictvo podohospodárstva, 168 p.

Bachasson, B., 1982. The larch in Haute-Savoie (in French). Rev. For. Fr. 34, 15–30.

Benkrima, L., von Aderkas, P., 1995. In vitro embryogenesis in larch. In: Schmidt, W.C., McDonald, K.J. (Eds.), Ecology and Management of *Larix* Forests: A Look Ahead. Proceedings International Symposium, Whitefish, Montana, USA, October 5-9, 1992. US Department of Agriculture Forest Service, Intermountain Research Station, 412416. General Technical Report INT-GTR-319.

Bezecný, P., Lipovský, I., Sumara, J., Šrámek, V., 1992. Silviculture (in Czech). Zemědělské nakladatelství Brázda, 367 p.

Bravo, A., Likitvivatanavong, S., Gill, S.S., Soberón, M., 2011. Bacillus thuringiensis: a story of a successful bioinsecticide. Insect Biochem. Mol. Biol. 41, 423–431. https://doi.org/10.1016/j.ibmb.2011.02.006.

CABI Compendium, 2022. *Larix decidua* (Common Larch). CABI International. https://doi.org/10.1079/cabicompendium.29967.

Cafourek, J., Kotrla, P., 2022. Average Yield of Planting Stock of Forest Woody Secies in the Czech Republic (Seedlings and Transplants) (in Czech). Ministry of Agriculture (Czech Republic), 6 p. https://eagri.cz/public/web/file/707507/Vypestnost_pro_PRV_8._6._1.pdf.

Carter, K.K., Selin, L.O., 1987. Larch plantation management in the northeast. N. J. Appl. For. 4, 18–20. https://doi.org/10.1093/njaf/4.1.18.

Chalupa, V., 2000. Growth of forest trees grown in vitro from organ cultures and from somatic embryos (in Czech). Lesn. Pr. 79, 498–501.

Chrz, V., Kasalický, E., et al., 1951. Almanac of forest seed production (in Czech). Československé státní lesy. Prague 65 p.

MLVH, 1977. Instructions for Forest Nurseries [Original in Czech: Instrukce Pro Lesní Školky Státních Organizací Lesního Hospodářství]. Ministry of Forestry and Water Management of Czechoslovakia, Forestry and Game Management Research Institute, 27 p.

Cook, D.B., 1969. Planted Larch in New York. Albany, New York (Published by the author).

Cornelissen, J.H.C., Werger, M.J.A., Castro-Díez, P., van Rheenen, J.W.A., Rowland, A.P., 1997. Foliar nutrients in relation to growth, allocation and leaf traits in seedlings of a wide range of woody plant species and types. Oecologia 111, 460–469. https://doi.org/10.1007/s004420050259.

Dauer, J.M., Chorover, J., Chadwick, O.A., Oleksyn, J., Tjoelker, M.G., Hobbie, S.E., Reich, P.B., Eissenstat, D.M., 2007. Controls over leaf and litter calcium concentrations among temperate trees. Biogeochemistry 86, 175–187. https://doi.org/10.1007/s10533-007-9153-8.

Decree No. 456/2021 Coll, 2021. On the details of the transfer of forest reproductive material, on the records of the origin of reproductive material, and on the details of forest stand regeneration and afforestation of land designated for the fulfilment of forest functions. Collection of Laws of the Czech Republic (in Czech). https://www.zakonyprolidi.cz/cs/2021-456.

Dirr, M., Heuser, C.W., 2009. The Reference Manual of Woody Plant Propagation: From Seed to Tissue Culture, second ed. Timber Press, Portland, OR.

Dušek, V., Kotyza, F., et al., 1970. Modern Forest Nursery (in Czech). Prague. Státní zemědělské nakladatelství 480 p.

Fajardo, A., Siefert, A., 2016. Phenological variation of leaf functional traits within species. Oecologia 180, 951–959. https://doi.org/10.1007/s00442-016-3545-1.

Gordon, A.B., 1992. Seed manual for forest trees. London. Forestry Commiss. 83, 86–97.

Helgerson, O.T., 1990. Heat damage in tree seedlings and its prevention. New Forest 3, 333–358. https://doi.org/10.1007/BF00030044.

Hoffman, J., Chválová, K., Palátová, E., 2005. Forest Seed Management in Slovakia (In Slovak). PEREX K+K, Bratislava, 195 p.

Holmes, S.B., MacQuarrie, C.J.K., 2016. Chemical control in forest pest management. Can. Entomol. 148, S270–S295. https://doi.org/10.4039/tce.2015.71.

Hrabí, L., 1989. Pre-sowing treatment of European larch (*Larix decidua* LAM. DC.) by soaking in water (in Czech). Lesnictvi 35, 131–142.
Jalkanen, R., 2016. Synthesis and new observations on needle pathogens of larch in northern Finland. Forests 7, 25. https://doi.org/10.3390/f7010025.
Jurásek, A., Nárovcová, J., Nárovec, V., 2006. A guide to containerized planting stock of forest woody species. Kostelec nad Černými lesy: Lesnická práce 56.
Jurásek, A., Nárovcová, J., Nárovec, V., Mauer, O., 2012. Forest reproductive materiál (in Czech). Czech Technical Standard. Úřad pro technickou normalizaci, metrologii a státní zkušebnictví 23.
Kacálek, D., Mauer, O., Podrázský, V., Slodičák, M., Houšková, K., Špulák, O., Souček, J., Novák, J., Jurásek, A., Leugner, J., Dušek, D., 2017. Soil improving and stabilising functions of forest trees (in Czech). In: Kostelec mad Černými lesy: Lesnická práce s.r.o, 300 p.
Kantor, J., 1975. Forest establishment and breeding of forest tree species (in Czech). Prague. Státní zemědělské nakladatelství 526 p.
Kloeppel, B.D., Treichel, I.W., Kharuk, S., Gower, S.T., 1998. Foliar carbon isotope discrimination in *Larix* species and sympatric evergreen conifers: a global comparison. Oecologia 114, 153–159. https://doi.org/10.1007/s004420050431v.
Kosinski, G., 1986. Megagametogenesis, fertilization, and embryo development in *Larix decidua*. Can. J. For. Res. 16, 1301–1309. https://doi.org/10.1139/x86-230.
Köstler, J.N., Brückner, E., Biebelriether, H., 1968. Die Wurzeln der Waldbäume (in German). Verlag Paul Parey, Hamburg und Berlin, 282 p.
Lehotský, L., 1964. Grafting and vegetative propagation of forest woody species (in Slovak). Slovenské vydavatelstvo podohospodárskej literatúry v Bratislave 130.
Lelu, M.A., Bastien, C., Klimaszewska, K., Ward, C., Charest, P.J., 1994. An improved method for somatic plantlet production in hybrid larch (*Larix x leptoeuropaea*): Part 1. Somatic embryo maturation. Plant Cell Tissue Organ Cult. 36, 107–115. https://doi.org/10.1007/BF00048322.
Leski, T., Rudawska, M., Aučina, A., 2008. The ectomycorrhizal status of European larch (*Larix decidua* Mill.) seedlings from bare-root forest nurseries. For. Ecol. Manag. 256, 2136–2144. https://doi.org/10.1016/j.foreco.2008.08.004.
Lestander, T., Bergsten, U., 1985. PREVAC-a method for the removal of mechanically damaged seeds (in Swedish). Sverige Skogsvårdsförbunds. Tidskrift 1, 35–42.
Li, B.L., Wyckoff, G.W., 1994. Breeding strategies for *Larix decidua, L. leptolepis* and their hybrids in the United States. For. Genet. 1, 65–72.
Li, T., Sheng, M.-L., Sun, S.-P., Luo, Y.-Q., 2013. Parasitoids of larch sawfly, Pristiphora erichsonii (Hartig) (Hymenoptera: Tenthredinidae) in Changbai Mountains. Journal of Natural History 48, 123131. https://doi.org/10.1080/00222933.2013.791930.
Machaníček, J., 1980. Research into Factors Affecting Long-Term Seed Storage (Spruce, Pine, Larch) (In Czech). Forestry and Game Management Research Institut, 52 p.
Mason, W.L., 1989. Vegetative propagation of hybrid larch (*Larix x eurolepis* Henry) using winter cuttings. Forestry 62, 189–198.
Mason, W., Edwards, C., Hale, S., 2004. Survival and early seedling growth of conifers with different shade tolerance in a Sitka spruce spacing trial and relationship to understorey light climate. Silva Fenn. 38, 357–370. https://doi.org/10.14214/sf.404.
Mauer, O., 2009. Forest Establishment I. Study Material. Mendel University in Brno, 174 p.
Mauer, O., et al., 2013. Planting Stock Production (In Czech). Mendel University of Agriculture and Forestry in Brno, 206 p.
Modrzyński, J., Chmura, D.J., Tjoelker, M.G., 2015. Seedling growth and biomass allocation in relation to leaf habit and shade tolerance among 10 temperate tree species. Tree Physiol. 35, 879–893. https://doi.org/10.1093/treephys/tpv053.
Möhl, P., Mörsdorf, M.A., Dawes, M.A., Hagedorn, F., Bebi, P., Viglietti, D., Freppaz, M., Wipf, S., Körner, C., Thomas, F.M., Rixen, C., 2019. Twelve years of low nutrient input stimulates growth of trees and dwarf shrubs in the treeline ecotone. J. Ecol. 107, 768–780. https://doi.org/10.1111/1365-2745.13073.

Ostenfeld, C.H., Larsen, C.S., 1930. The Species of the Genus *Larix* and Their Geographical Distribution. Bianco Lunos Bogtrykke, Copenhagen, Denmark, 106 p.
Owens, J.N., 1995. Reproductive biology of larch. In: Schmidt, W.C., McDonald, K.J. (Eds.), Ecology and Management of *Larix* Forests: A Look Ahead. Proceedings International Symposium, Whitefish, Montana, USA, October 5–9, 1992. General Technical Report INT-GTR-319, US Department of Agriculture Forest Service, Intermountain Research Station, pp. 97–109.
Palátová, E., 2008. Forest Establishment I, Forest Seed Management (In Czech). Mendel University of Agriculture and Forestry in Brno, 121 p.
Pâques, L.E., 1994. Relationship between foliar nutrient concentrations and growth of hybrid larch (*Larix* × *eurolepsis* Henry). For. Ecol. Manag. 63, 153–167. https://doi.org/10.1016/0378-1127(94)90108-2.
Parde, J., 1957. A plea for the Larch (in French). Rev. For. Fr. 9, 634–651.
Plants for A Future, n.d. *Larix decidua* - database search results, October 2025, Lostwithiel, Cornwall, UK. https://pfaf.org/user/Plant.aspx?LatinName=Larix+decidua.
Plesa, I.M., Al Hassan, M., González-Orenga, S., Sestras, A.F., Vicente, O., Prohens, J., Boscaiu, M., Sestras, R.E., 2019. Responses to drought in seedlings of European larch (*Larix decidua* mill.) from several *Carpathian provenances*. Forests 10, 511. https://doi.org/10.3390/f10060511.
Repáč, I., Parobeková, Z., Sendecký, M., 2017. Reforestation in Slovakia: history, current practice and perspectives. Reforesta 3, 53–88. https://doi.org/10.21750/REFOR.3.07.31.
Rudolf, P.O., 1974. *Larix* Mill., larch. In: Schopmeyer, C.S. (Ed.), Seeds of Woody Plants in the United States. US Department of Agriculture Forest Service, Washington, DC, pp. 478–485. Agriculture Handbook 450.
Schmidt, W.C., McDonald, K.J., 1995. Ecology and Management of *Larix* Forests: A Look Ahead. Proceedings International Symposium, Whitefish, Montana, USA, October 5-9, 1992. General Technical Report INT-GTR-319. US Department of Agriculture Forest Service, Intermountain Research Station, p. 521.
Shearer, R.C., 2008. *Larix* P. Mill. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual. US Department of Agriculture Forest Service, Agriculture Handbook 727, Washington, DC, pp. 637–647.
Sheat, W.G., 1957. Propagation of Trees, Shrubs and Conifers. MacMillian & CO LTD, London, 479 p.
Skrzecz, I., Sierpińska, A., Tumialis, D., 2024. Entomopathogens in the integrated management of forest insects: from science to practice. Pest Management Science 80, 2503–2514. https://doi.org/10.1002/ps.7871.
Šmelková, Ľ., 2009. Forest establishment (in Slovak). Zvolen. Technical University in Zvolen, 338 p.
Tichý, J., 1949. Cultivation of larch (in Czech). Česká akademie zemědělská 42.
Vidakovic, M., 1991. Conifers: Morphology and Variation. CAB Int, Wallingford, UK, 754 p.
Vincent, G., 1965. Forest Seed Management (In Czech). Státní zemědělské nakladatelství, Prague, 329 p.
Weisgerber, H., Sindelar, J., 1992. IUFRO's role in coniferous tree improvement. History, results, and future trends of research and international cooperation with European larch (*Larix decidua* Mill.). Silvae Genet. 41, 150–161.

Olea europaea L. *var. sylvestris Brot*

Luna Morcillo[1], Antonio del Campo[2] and Alberto Vilagrosa[1]
[1]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain; [2]Research Group in Forest Science and Technology (Re-ForeST), Universitat Politècnica de València, Valencia, Spain

Propagation

Seed propagation

The seeds *of Olea europaea* var. *sylvestris* (hereafter, *O. europaea)* start developing in July and usually end by September, when the fruit is viable. Physiological maturation, nevertheless, may extend until mid-winter. The fruit's maturity state will condition germination rates. Due to the inhibitory effect of the mature pulp on the seed (Martin, 2008), seeds harvested and stratified in September to October usually have maximum germination rates in comparison with physiologically ripe fruits. Early harvesting also prevents fruit predation by frugivorous birds (Herrera, 1998).

Seeds should be collected from many individuals, from different populations, to ensure high genetic diversity. Olives are harvested from the tree using the traditional methods of hand-pole beating and vibrator pickers, or a mixture of beating and vibration. The time between harvesting and processing should be minimized to prevent the fruit from drying out. Small-seeded fruits tend to germinate more easily.

The type of basic material for this species corresponds in most cases to seed sources and stands, which provide forest reproductive material of the identified category (according to EU regulation, 2023). However, given the coexistence of natural populations of wild olive trees with cultivated and naturalized populations of olive trees in the Mediterranean landscapes and the close genetic makeup of the genotypes, the selection of seed sources and stands for seed sourcing can be challenging. This normative aspect is one of the pitfalls that have prevented the wild olive tree from broader use in forest restoration programs.

After fruit collection, the seed is extracted by pulp maceration, gently crushed with a blender avoiding seed damage, and separating the seeds from the pulp using a sieve and pressurized water. Selection of viable seeds ends by soaking in a sodium chloride solution (250–300 g/L), washed and dried or together with a gravimetric separation by winnowing or on a densimetric table. The orthodox character of *O. europaea* seeds (i.e., long-lived seeds) allows them to be stored with 6%–8% moisture in containers airtight at 4–5°C for several years, without losing its germination capacity (García-Fayos et al., 2001). The clean seed represent 15%–35% of the total weight of the olive fruit.

The pregermination treatments recommended for this species must overcome both the mechanical resistance of the endocarp and the physiological dormancy of the embryo. To overcome endocarp resistance, mechanical scarification could be applied to break the endocarp, or chemical scarification by soaking the seeds in concentrated

sulfuric acid for 24–30 h and then washing it for 1–2 h (Martin, 2008). Comparing the effect of chemical and mechanical scarification treatments, Rostami and Shasavar (2009) found the highest germination percentage after 97% sulfuric acid solution treatments for 6h in several commercial varieties of *O. europaea*. After scarification, the seed could be sown, but germination is significantly improved when the scarification is combined with stratification (after soaking in water for 24 h) in sand or moist vermiculite at 15°C, for 1 month and in darkness. This combination reduces the levels of abscisic acid responsible for dormancy (Martin, 2008).

Different pregermination treatments have been gathered from the literature: (i) cleaning the seed with NaOH, followed by washing in running water for 2 h and germination at a constant temperature of 13°C for 48 days. (ii) Washing for 24 h and germination at 15°C in darkness or half-light for 2 months. (iii) Cold stratification for 1 week. (iv) In controlled propagation work, the structures of the seed coat, endocarp and testa have been eliminated, achieving adequate germination at 13°C in light or darkness (Navarro-Cerrillo and Gálvez, 2001). Other trials demonstrate higher germination rates after pregermination chemical with procedures such as Gibberellic acid (GA_3) 500 ppm for 12 h dip, HCl 1 N for 12 h dip or NaOH 1 N for 12 h dip (Lal et al., 2015).

Due to the difficulty and time that pretreatment involves, it is widely accepted to evaluate the seed lots by means of colorimetric assays, which allows faster knowledge of the quality of the seed lot. The high variability in the characteristics of *O. europaea* seed lots is shown in Table 11.27.

There exists some controversy regarding the effect of seed size on germination. Some authors found a positive correlation (Rey et al., 2004), while other found no relationship (Rinaldi, 2004). Seedlings from bigger-size seeds, nevertheless, show a higher proportion of root biomass, which increase seedling establishment in dry sites (del Campo and Navarro-Cerrillo, 2013).

Vegetative propagation

Olea europaea can be vegetatively propagated using branch cuttings, grafting, and layering (Lambardi et al., 2023). For rooting cuttings, the recommended procedure is using a mixture of peat-perlite at 10–15°C ambient temperature and 25°C basal temperature.

Table 11.27 Characteristics of seed lots of *Olea europaea* var. *sylvestris*.

Purity (%)	No. seeds/kg	Germination capacity (%)
95–100 (99)	1,000–9,500 (4,530)	5–97 (67)

Mean values in brackets.
García-Fayos (2001); Navarro-Cerrillo and Gálvez (2001); Piotto and Di Noi (2001); Louro and Pinto (2011); Picher-Morelló et al. (2013); Herrero-Sierra and Nicolás-Peragón (2013).

Some hormonal treatments to stimulate rooting have been tested: (i) 2.500 ppm AIB + 2.500 ppm ANA, recording 90% rooting. (ii) 2.500 ppm AIB + 2.500 ppm ANA + 25% capture, recording 80% rooting; (iii) 5000 ppm of AIB + 25% of Captan, registering 40% rooting (Dirr and Heuser, 1987).

It is recommended to start from leafy branch cuttings, since it promotes callusing and root development (Suárez et al., 1999). Trials with the African subspecies (i.e., subsp. *cuspidata*) resulted in 70%–90% rooting using only indole-butyric acid (IBA) at doses of 20–40 μg per cutting, which were 2–3 mm in diameter and 10 cm in length (Negash, 2003). Micropropagation techniques have been greatly developed in past decades (Rugini et al., 2005; Lambardi et al., 2013). The in vitro culture of isolated embryos without influence of the endocarp entails minimization of inhibitory effects, with 100% germination in just 10 days compared with the 300 days required to achieve 40% germination in conventional sowing (Cantos et al., 2001). However, the development of these plants can be slowed down (Acebedo et al., 1997). Good results have also been reported with mature or immature embryos (Orinos and Mitrakos, 1991; Rugini et al., 2005).

Propagation material from mother plants (cuttings, explants, etc.) must be assured to be free of infections such as that of the bacterium *Pseudomonas savastanoi* which is a common knot disease on olives that can constraint several stages of plant propagation and can subsequently be spread from nursery plants (Marchi et al., 2009).

Seedling cultivation

Container plants

Cultivation of *O. europaea* is commonly done in container nurseries using seeds. Sowing depth should be 2–3 times the diameter of the seed. It is not recommended to sow directly in the definitive container, except with pregerminated seeds, given the irregular germination rates of this species (Table 11.28). Seeds without stratification sown in winter will begin to germinate in spring, although very irregularly, and may extend germination up to 3 months. Therefore, the most common practice is to stratify, sow then in a seedbed reducing thereby the germination period to 1 month, and subsequently transplanting the germinated seeds to the definitive container after 1 or 1.5 months, when the recently emerged seedling has three true leaves or when the extraction from the cell would be safe. This method can occasionally result in root deformation issues (Navarro-Cerrillo et al., 1998) and adverse transplant shock effects, which require an extended recovery period for the plants. Transplanting wild olive seedlings from the seedbed to the forest container trays can cause a considerable slowdown in growth due to the transplant stress. Thus, it is highly recommended that sowing to be carried out in a small volume cell (<75 cm^3) and once the seedling has colonized the reduced container transplant it to de final container. This practice, together with high nitrogen fertilization

Table 11.28 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Olea europaea* var. *sylvestris*. The root collar diameter is measured immediately above the cotyledon insertion point. Observed values are mean values ± standard error.

	Container seedlings	
Attributes	**Observed values**	**Recommended values**
Shoot height (cm)	41.2 ± 9.8	20–60
Root collar diameter (cm)	0.45 ± 0.1	0.3–0.56
Shoot dry weight (g)	3.5 ± 1.3	2–5
Root dry weight (g)	1.7 ± 0.7	1.1–2.3
Plant dry weight (g)		3.1–7.3
Shoot/Root dry weight ratio	2.2 ± 0.5	1.4–3.8
Leaf[N] (mg/g)	22.8 ± 4.7	14–28
Leaf [P] (mg/g)	1.3 ± 0.7	1.2–1.4
Leaf [K] (mg/g)	10.1 ± 3.7	8–12

Navarro-Cerrillo et al. (1998); del Campo (2002); del Campo and Segura (2009).

after transplant, has led to high growth rates in the early and rapid growth phases of nursery cultivation (del Campo, 2002). In seedbeds, a mixture of peat:coconut coir: perlite (6:3:1, by volume, or 5:4:1) is commonly used as growing medium, covering the surface with vermiculite. It is important to provide the plant with adequate environmental conditions, shading and high environmental humidity to successfully pass this phase (Del Campo and Navarro-Cerrillo, 2013).

Plant cultivation in containers is carried out during one or two vegetative cycles, with highly variable cultivation programs depending on the nursery location (Navarro-Cerrillo et al., 1998). Minimizing transplant shock (i.e., germination and transplanting of seedlings from the seedbed) improves efficiency and allows obtaining plants of suitable morphologic characteristics. The size of the containers used in cultivation ranges between 150 and 300 cm^3, with densities ranging between 247 and 387 plants/m^2. 1-year-old seedlings for forest restoration should be grown preferably in containers of 200–300 cm^3 (6 x 6 × 18 cm) to obtain a high-quality seedling of 30–50 cm height with a well-developed root system (Table 11.28).

O. europaea has no special growing media requirements so it is commonly cultivated with conventional mixtures based on organic components such as blonde and black peat or coconut coir as main components (>75% by volume) and some inorganic components such as perlite, vermiculite, or sand (<25% by volume).

Regarding fertilization, the supply of N and K is especially important in *O. europaea* to guarantee proper seedling development and provide adequate nutritional concentrations in the leaf. The fertilization of the wild olive does not seem so dependent on the high dose of phosphorus. In fact, most nurseries producing *O. europaea*, tend to

incorporate a slow-release fertilizer as an additive in the substrate formulation, ensuring that the product maintains its nutrient supply throughout the vegetative period (Di Vaio et al., 2021). A dose of 2.5–3.5 kg/m^3, of an N-P-K 15-8-15 fertilizer (8–9 months release), is appropriate for the species. In case of extending the nursery period, a certain control is recommended to ensure nutritional requirements throughout the period. In fertigation programs, Navarro-Cerrillo et al. (1998) recommend an initial base fertilization with fertilized peat, followed by fertilizations rich in N and K. Fertilization may start with a Starter type fertilizer (9-45-15) and a concentration in the solution of 50–90 ppm of N with a weekly or biweekly frequency. During the active growth phase, a Grower type fertilizer (20-20-20) is advisable and a concentration in the solution of 150–200 ppm of N and a frequency of 2–3 times per week, which may vary depending on the of plant development state. Finally, once the proper height for the lot is reached, the N supply should be considerably reduced by switching to a Finisher type fertilizer with weekly or biweekly applications (Landis et al., 1989). Wild olive tree typically shows higher range of foliar concentrations of nutrients than other common species, which is due to the high degree of leaf storage. Hardening or drought preconditioning are not recommended when the plant is not intended for cold areas (Del Campo and Navarro-Cerrillo, 2013). In summary, both the transplant method and nitrogen fertilization can be considered the most effective tools to control plant development in the nursery and its quality attributes (del Campo, 2002).

Finally, several works reported better field and nursery performance in *O. europaea* mycorrhized individuals by the arbuscular mycorrhizal fungi of the genus *Glomus* (Caravaca et al., 2002; Estaún et al., 2003; Dag et al., 2009). Mycorrhizal plants have better growth, greater vigor, greater assimilation rates of nutrients, such as phosphorus and potassium, and a better comprehensive response in arid and semiarid conditions. To incorporate inoculation in the nursery simple techniques can be applied. Dag et al. (2009), for instance, inoculated *Glomus mosseae* and *Glomus intraradices* through the suspension of spores, hyphae, infected roots and substrate, which was applied on vermiculite, which participated in the substrate in 10% of its volume.

Bareroot plants

Bareroot production in *O. europaea* is not very common due to the purposes of the plant production, although many transplanted large trees have been reported from the field to nurseries for gardening (Sánchez and Arroyo, 2003).

Field establishment

Its unique functional traits (physiological, reproductive, and morphological) regarding drought and fire resistance make *O. europaea* var. *sylvestris* one of the most suitable species for generalized use in forest restoration programs (Kassout et al., 2022). Planting is the

only viable reforestation method for *O. europaea*. Planting holes and soil preparation can be manually or mechanically performed using crawler backhoes or walking excavators to minimize soil erosion. Planting should be conducted in autumn or winter, according to the period of precipitation. Delays that could compromise the survival of the plantations must be avoided, particularly in degraded areas with very shallow soils. Despite root activity of *O. europaea* during the winter is not very pronounced (Sardá et al., 2005), it is enough to advance into the establishment process, especially in warm locations, so it is recommended to advance planting as much as possible. In contrast, *O. europaea* reforestations should be avoided in areas with high risk of frost, due to the great sensitivity of the species to low temperatures.

Field performance of *O. europaea* under favorable conditions has demonstrated being successful, both in survival and growth, as reported by numerous works (Del Campo and Navarro-Cerrillo, 2004; Domínguez et al., 2004; Vilagrosa et al., 2008; Alameda and Villar, 2009; Kribeche et al., 2012; Morcillo and Bautista, 2022) where the species usually resulted in survival rates above the average (when compared to other species), with values between 70% and 100% survival. In trial plantations with cooccurring species such as holm oak and carob tree (del Campo, 2002, Del Campo and Navarro-Cerrillo, 2004), this species showed a greater capacity for early posttransplant development, surviving at higher rates and growing faster, allowing for good correlations of the Root Growth Potential test with establishment success. The authors also reported that the lack of water preconditioning can favor survival if the weather subsequent to planting is favorable.

Although *O. europaea* is considered a suitable candidate to adapt to predicted climate change scenarios (Ruiz de la Torre, 2006; Navarro-Cerrillo et al., 2009), in tests under very adverse ecological conditions and in unfavorable areas coinciding with years of very low precipitation, mortality rates can sharply increase up to 80% (Padilla-Ruiz et al., 2004; Sánchez et al., 2004; Morcillo and Bautista, 2022).

In very limiting establishment conditions, especially in areas with high variability in precipitation as occur in arid and semiarid Mediterranean areas, some authors recommend applying establishment irrigation if the conditions at the time of planting are unfavorable, and maintenance irrigation during the first summer, in a variable number, between 2 and 4, depending on the ease and economy (Padilla-Ruiz et al., 2004). In this regard, several irrigation methods have been experimentally tested to optimize irrigation doses in semiarid ecosystems. Sánchez et al. (2004) probed the efficiency of micro-irrigation at 25 cm depth (5 mm diameter PVC-tube connected to a network of pipes that linked to a water tank. Irrigation doses of 1.5–3 L per plant applied before and during summer resulted in close to 100% survival of *O. europaea* seedlings, while the control without irrigation resulted in 3% survival. Morcillo and Bautista applied punctual doses of 1.5 L per plant in emergency irrigation application, during the following 3 years after outplanting. In this case the treatments consisted of 50 mm diameter PVC-tubes installed 30–40 cm depth, at the base of the planting hole (Fig. 11.17A).

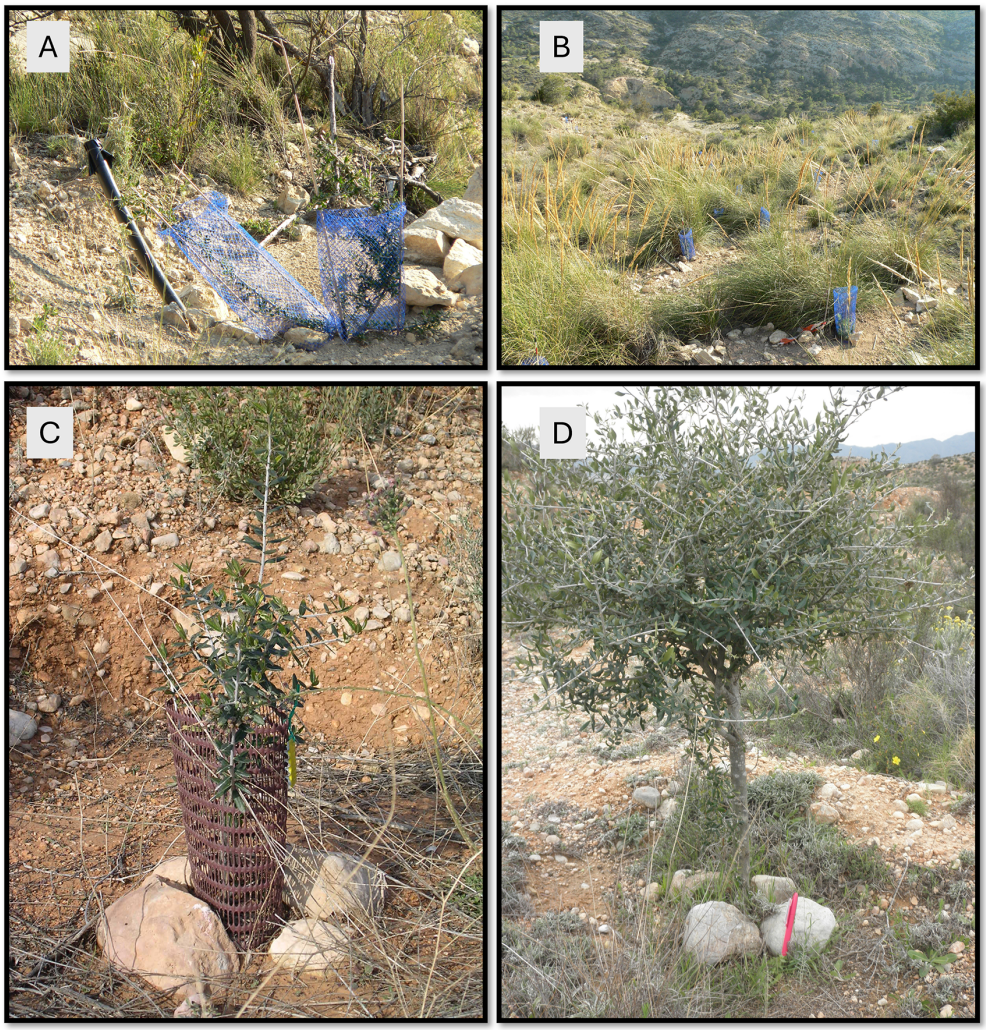


Figure 11.17 Images illustrating several *Olea europaea* field plantations and the implementation of several treatments, such as deep irrigation (A), protective meshes (B and C) and plant performance 10 years after reforestation. *(Luna Morcillo and Alberto Vilagrosa.)*

The authors found that water addition clearly improved *O. europaea* survival rates. Some low-cost techniques, such as microcatchments or dry wells proben being a good alternative to provide pulses of water in critical periods what resulted essential for the survival of seedlings and young individuals of *O. europaea* (Valdecantos et al., 2014). On the contrary, other solutions such as hydrogels have been poorly tested for this species, since the soils on which this species is usually reforested tend to be loamy or silty, which is not appropriate for the use of these polymers (del Campo et al., 2011).

The use of mycorrhizal plants has been shown to be effective in improving the establishment of the species in harsh areas, with positive effects on photosynthetic rate, transpiration, and stomatal conductance of *O. europaea* (Caravaca et al., 2002).

The installation of protective devices on the seedlings, such as tree shelters, is frequent for this species, particularly in areas with a certain risk of frost (del Campo et al., 2013). In warm areas, nevertheless, the use of tree shelter could be avoided, since *O. europaea* is a very tolerant species to full sunlight conditions. In fact, the use of nurse plants to provide shade (among other possible facilitative effects) to *O. europaea* seedlings in semiarid conditions, resulted in prevalent net competition for resources, since *O. europaea* did not required to benefit from shade to survive during the establishment period in the field (Morcillo and Bautista, 2022). However, if there is predation risk the installation of protective meshes is recommended (Fig. 11.17B and C). Combined field treatments as microcatchments, the use of tree shelter or the addition of compost to the plantation hole increase the growth of this species over other typical Mediterranean such as *Quercus coccifera* or *Pistacia lentiscus* (Fig. 11.17D, Kribeche et al., 2012).

Chapter References

Acebedo, M.M., Lavee, S., Liñán, J., Troncoso, A., 1997. *In vitro* germination of embryos for speeding up seedling development in olive breeding programs. Sci. Hort. 69 (3–4), 207–215.

Alameda, D., Villar, R., 2009. Estrategias de respuesta a la sequía en plántulas de 17 especies leñosas y el efecto de la lluvia estival. En: Actas del 5 Congreso Forestal Español. [cd-rom]. In: Sociedad Española de Ciencias Forestales. Junta de Castilla y León, Ávila.

Cantos, M., Liñán, J., Troncoso, J., Aparicio, A., Troncoso, A., 2001. El cultivo in vitro, un método para mejorar la germinación de plantas con interés forestal en Andalucía. In: de Andalucía, J. (Ed.), En: Actas del III Congreso Forestal Español. Mesa 3. Granada, pp. 683–689. Available in: http://congresoforestal.es.

Caravaca, F., Barea, J.M., Figueroa, D., Roldán, A., 2002. Assessing the effectiveness of mycorrhizal inoculation and soil compost addition for enhancing reforestation with *Olea europaea* subsp. *sylvestris* through changes in soil biological and physical parameters. Appl. Soil Ecol. 20 (2), 107–118.

Dag, A., Yermiyahu, U., Ben-Gal, A., Zipori, I., Kapulnik, Y., 2009. Nursery and post-transplant field response of olive trees to arbuscular mycorrhizal fungi in an arid region. Crop Pasture Sci. 60, 427–433.

del Campo, A.D., 2002. Régimen de cultivo, desarrollo en vivero, calidad de planta y respuesta al establecimiento en cuatro especies de frondosas mediterráneas. PhD. Universidad de Córdoba, Córdoba.

del Campo, A.D., Navarro-Cerrillo, R.M., 2004. Calidad de lotes comerciales de acebuche (*Olea europaea* var. *sylvestris* Brot.). Evaluación de su respuesta en campo. Cuad. Soc. Esp. Cienc For. 17, 43–49.

del Campo, A., Segura, G., 2009. Definición de protocolos para el control de calidad de planta y puesta en obra de la misma. Informe 2009. Universidad Politécnica de Valencia y Consellería de Medio Ambiente, Agua, Urbanismo y Vivienda, GVA.

del Campo, A., Navarro-Cerrillo, R.M., 2013. Olea europaea L. var. sylvestris Brot. in Puerta-Piñero, C. (2013). "Producción y manejo de semillas y plantas forestales" (tomos I y II) (2012–2013). Coordinado por Jesús Pemán y colaboradores. Ecosistemas 22 (1), 92-92.

del Campo, A.D., Hermoso, J., Flors, J., Lidón, A., Navarro-Cerrillo, R.M., 2011. Nursery location and potassium enrichment in Aleppo pine stock 2. Performance under real and hydrogel-mediated drought conditions. Forestry 84 (3), 235–245.

Di Vaio, C., Testa, A., Cirillo, A., Conti, S., 2021. Slow-release fertilization and *Trichoderma harzianum*-based biostimulant for the nursery production of young olive trees (*Olea europaea* L.). Agron. Res. 19, 1396–1405.

Dirr, M.A., Heuser, A.C., 1987. The Reference Manual of Woody Plant Propagation: From Seed to Tissue Culture. Varsity Press Inc., Georgia.

Domínguez, M.T., Marañón, T., Murillo, J.M., Hidalgo, J.A., Madejón, P., 2004. Crecimiento y morfología foliar de especies leñosas en las reforestaciones del corredor verde del Guadiamar (Sevilla). Cuad. Soc. Esp. Cienc. For. 20, 173–179.

Estaún, V., Camprubi, A., Calvet, C., Pinochet, J., 2003. Nursery and field response of olive trees inoculated with two arbuscular mycorrhizal fungi, *Glomus intraradices* and *Glomus mosseae*. J. Am. Soc. Hort. Sci. 128, 767–775.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal de la Comunidad Valenciana. In: Banc de Llavors Forestals, Conselleria de Medi Ambient, Generalitat Valenciana, Valencia, p. 27.

Herrera, C.M., 1998. Long-term dynamics of Mediterranean frugivorous birds and fleshy fruits: a 12-year study. Ecol. Monogr. 68, 511–538.

Herrero-Sierra, N., Nicolás-Peragón, J.L., 2013. Anexo III Datos característicos de lotes de semillas. CNRGF El Serranillo. In: Pemán- García, J., Navarro-Cerrillo, R.M., Nicolás Peragón, J.L., Prada-Saez, M.A., Serrada-Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Ministerio de agricultura, alimentación y medio ambiente., Madrid, Spain, pp. 734–739.

Kassout, J., Terral, J.F., El Ouahrani, A., Houssni, M., Ivorra, S., Kadaoui, K., El Mahroussi, M., Paradis, L., Ater, M., 2022. Species distribution based-modelling under climate change: The case of two native wild *Olea europaea* subspecies in Morocco, *O. e.* subsp. *europaea* var. *sylvestris* and *O. e.* subsp. *maroccana*. In: Leal Filho, W., Manolas, E. (Eds.), Climate Change in the Mediterranean and Middle Eastern Region. Climate Change Management. Springer, Cham.

Kribeche, H., Aguilar, S.B., Miranda, E.C., Carmona, A.V., Calzada, V.R.V., 2012. Effects of landscape spatial heterogeneity on dryland restoration success: the combined role of site conditions and reforestation techniques in southeastern Spain. Ecol. Mediterr.: Int. J. Mediterran. Ecol. 38 (1), 4–17.

Lal, S., Ahmed, N., Srivastava, K.K., Singh, D.B., 2015. Olive (*Olea europaea* L.) seed germination as affected by different scarification treatments. Afr. J. Agric. Res. 10 (35), 3570–3574.

Lambardi, M., Ozudogru, E.A., Roncasaglia, R., 2013. In vitro propagation of olive (*Olea europaea* L.) by nodal segmentation of elongated shoots. In: Protocols for Micropropagation of Selected Economically-Important Horticultural Plants, pp. 33–44.

Lambardi, M., Fabbri, A., Micheli, M., Vitale, A., 2023. Olive propagation and nursery. In: Fabbri, A., Baldoni, L., Caruso, T., Famiani, F. (Eds.), The Olive: Botany and Production. CABI.

Landis, T.D., 1989. Mineral nutrients and fertilization. In: Landis, T.D., Tinus, R.W., McDonald, S.E., Barnett, J.P. (Eds.), The Container Tree Nursery Manual. U.S. Department of Agriculture, Forest Service, Washington, USA, pp. 1–70.

Louro, V., Pinto, G., 2011. Sementes, uma ponte entre o passado e o futuro da floresta. Ministério da Agricultura, Mar, Ambiente e Ordenamento do Territorio. CENASEF, pp. 31–38.

Martin, G.C., 2008. *Olea europea* L. En. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual, United States Department of Agriculture, Forest Service, vol 727. Agriculture Handbook, Washington, pp. 753–756.

Marchi, G., Mori, B., Pollacci, P., Mencuccini, M., Surico, G., 2009. Systemic spread of *Pseudomonas savastanoi* pv. *savastanoi* in olive explants. Plant Pathol. 58, 152–158. https://doi.org/10.1111/j.1365-3059.2008.01935.x.

Morcillo, L., Bautista, S., 2022. Interacting water, nutrients, and shrub age control steppe grass-on-shrub competition: implications for restoration. Ecosphere 13 (5), e4093.

Navarro-Cerrillo, R.M., Del Campo, A., Alejano, R., Alvarez, L., 1998. Caracterización de calidad final de planta de encina (*Quercus ilex* L.), alcornoque (*Quercus suber* L.), algarrobo (*Ceratonia siliqua* L.) y acebuche (*Olea europaea* L. var. sylvestris) en cinco viveros en Andalucía. Informaciones Técnicas 49/98. Junta de Andalucía, Consejería de Agricultura y Pesca, Sevilla.

Navarro-Cerrillo, R.M., Gálvez, C., 2001. Manual para la identificación y reproducción de semillas de especies vegetales autóctonas de Andalucía. Tomo II. Consejería de Medio Ambiente. Junta de Andalucía, Córdoba, pp. 304–309.

Negash, L., 2003. Vegetative propagation of the threatened African wild olive [Olea europaea L. subsp.cuspidata (Wall. ex DC.) Ciffieri]. New For 26, 137–146.

Orinos, T.H., Mitrakos, K., 1991. Rhizogenesis and somatic embryogenesis in calli from wild olive (*Olea europaea* var. sylvestris (Miller) Lehr) mature zygotic embryos. Plant Cell Tiss. Organ Cult. 27 (2), 183–187.

Padilla-Ruiz, F.M., Pugnaire, F.I., Marín, R., Hervás, M., Ortega, R., 2004. El uso de especies arbustivas para la restauración de la cubierta vegetal en ambientes semiáridos. Cuad. Soc. Esp. Cien. For. 17, 103–107.

Picher-Morelló, M., Ventimilla-Antón, P., Campos-Fuster, E., 2013. Anexo II Datos característicos de lotes de semillas. Banc de llavors forestals, Dirección General del Medio Natural, Generalitat Valenciana. In: Pemán-García, J., Navarro-Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Ministerio de agricultura, alimentación y medio ambiente, Madrid, Spain, pp. 731–733.

Piotto, B., Di Noi, A. (Eds.), 2001. Propagazione per seme di alberi e arbusti della flora mediterranea. ANPA, Roma, 108 pp.

Rey, P.J., Alcántara, J.M., Valera, A.M., Sánchez-La Fuente, A.M., Garrido, J.L., Ramírez, J.M., Manzaneda, A.J., 2004. Seedling establishment in *Olea europaea*: seed size and microhabitat affect growth and survival. EcoScience 11 (3), 310–320.

Rinaldi, L., 2004. Relationships between seed biomass, germination time, and seedling growth in *Olea europaea*. Hortscience 39 (4), 788.

Rostami, A.A., Shasavar, A., 2009. Effects of seed scarification on seed germination and early growth of olive seedlings. J. Biol. Sci. 9 (8), 825–828.

Ruiz de la Torre, J., 2006. Flora Mayor. Organismo Autónomo Parques Nacionales. Dirección General para la Biodiversidad, Madrid, pp. 1027–1032.

Rugini, E., Mencuccini, M., Biasi, R., Altamura, M.M., 2005. In: Jain, S.M., Gupta, K. (Eds.), Olive (*Olea europaea* L.) En: Protocolfor Somatic Embryogenesis in Woody Plants. Springer, The Netherlands, pp. 345–360.

Sánchez, Arroyo, 2003. Transplante de material vegetal de porte arbóreo por el método de cepellón escayolado. Consejería de Medio Ambiente. Junta de Andalucía, Sevilla. ISBN: 8495785730.

Sánchez, J., Ortega, R., Hervás, M., Padilla, F.M., Pugnaire, F.I., 2004. El microrriego, una técnica de restauración de la cubierta vegetal para ambientes semiáridos. Cuad. Soc. Esp. Cienc. For. 17, 109–112.

Sardá, P., Aguilar, A., Valle, G., Villar-Salvador, P., Peñuelas, J.L., 2005. Crecimiento radical de plantones de *Retama sphaerocarpa, Pistacia terebinthus y Olea europaea* durante el período húmedo del año. En: Actas del IV Congreso Forestal Español. [cd-rom]. In: Sociedad Española de Ciencias Forestales. Zaragoza. Available in: http://congresoforestal.es.

Suárez, M.P., López-Rivarés, E.P., Lavee, T.,A., 1999. Rooting capability of olive cuttings, cv. Gordal: influence of the presence of leaves and buds. Acta Hort. 474, 39–42.

Valdecantos, A., Fuentes, D., Smanis, A., Llovet, J., Morcillo, L., Bautista, S., 2014. Effectiveness of low-cost planting techniques for improving water availability to Olea europaea seedlings in degraded drylands. Restor. Ecol. 22, 327–335. https://doi.org/10.1111/rec.12076.

Vilagrosa, A., Chirino, E., Bautista, S., Urgeghe, A.M., Alloza, J.A., Vallejo, V.R., 2008. Proyecto de demostración de lucha contra la desertificación: Regeneración y plan de manejo de zona semiáridas degradadas en el T.M. de Albatera (Alicante). Cuad. Soc. Esp. Cien. For. 28, 317–322.

Picea abies (L.) Karst

Vladan Ivetić[1] **and Valeriu-Norocel Nicolescu**[2]
[1]University of Belgrade, Faculty of Forestry, Belgrade, Serbia; [2]Faculty of Silviculture and Forest Engineering, Transilvania University of Braşov, Braşov, Romania

Propagation

Seed propagation

Cones can be harvested from standing trees from the end of October to March (Stilinović, 1985), but some authors recommend November (Hoffmann et al., 2007). Cones are harvested manually or using scissors or hooks. They can also be collected on the ground by previously placing tarpaulin or plastic foils (Iancu, 1999). At harvest the cones contain about 40% water, while seeds have a water content of 10%–20% less (Vlase, 1982).

Cones should be predried to a moisture content of 25%–30% before seed extraction, especially if they are harvested in late autumn or early winter, as they then have a higher moisture content compared with those harvested later. All large impurities, for example, branches and stones, should be removed before drying for seed extraction. To extract seed, cones should be heated and dried, which can be done by exposing the cones to the sun or in different facilities where it is possible to control the temperature and humidity in drying chambers. Chambers accelerate reaching the targeted temperature and humidity values for maintaining seed viability and germination capacity.

Cones of *P. abies* will open at temperatures between 45 and 55°C and at moisture content of 10%–15%. The selected temperature and exposure time will depend on the moisture content of the cone. Cones with a higher moisture need to be dried slowly and at lower temperatures (Ivetić, 2023). As a reference, seed drying lasts for 65 h if the temperature is 35°C and approximately 20–23 h if the drying temperature is 45–55°C (Damian, 1978). Extracted seeds need to be dewinged, which can be done using dry or wet procedures. As the moisture content of the extracted seeds usually ranges between 10% and 14%, additional drying is needed following wet dewinging. The seed should be graded at least based on size. To improve the quality of a seedlot additional separation is recommended based on both gravity and specific mass, to remove remaining impurities in seeds of the same size but of different weight. This is especially important for container stock production, where the aim is to produce seedlots with a 100% purity. The yield of cone extraction is 1.0–1.9 kg of winged seeds or 0.8–1.6 kg of wing-free seeds hL^{-1} of cones (Iancu, 1999).

P. abies seed is orthodox and can be dried to low moisture content allowing storage in a very cold environment. For operational use, seeds with a moisture content of 5%–6% can be stored for many years at temperatures between −3 and −10°C in tightly sealed containers that should never be opened before the end of storage (Suszka et al., 2005). *P.*

Table 11.29 Characteristics of seedlots of *Picea abies* (L.) Karst.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
2.5–5 (3)	90–94 (90)	111,000–192,000 (130,000)	80–90 (85)

Mean values in brackets.
Jovković (1952); Regent (1980); Stilinović (1985); Milev et al. (1999); Hoffmann et al. (2007); Ivetić (2023).

abies seed does not require any pregermination treatment and seeds germinate rapidly after sowing. Seeds stored at temperatures below 0°C need to be gradually warmed up to at least +3°C before sowing (Szabla and Pabian, 2009). To achieve more uniform germination, some authors recommend immersing the seeds in water at 17–20°C for 24 h (Stilinović, 1987; Hoffmann et al., 2007; Himanen, 2018; Houšková et al., 2021). The quantitative characteristics of Picea abies seeds are presented in Table 11.29.

Vegetative propagation

P. abies can be vegetatively propagated through grafting, rooting, and somatic embryogenesis. Recently, combining rooting and somatic embryogenesis by using emblings as a donor for cuttings has shown promising potential for accelerating production of clonal material for field testing, and potentially for planting stock (Tikkinen et al., 2017).

Grafting has been used for decades to establish clonal seed orchards and clonal archives (OECD, 2006). Grafting is achieved by uniting compatible scions, taken from the crown of mature ortet trees and rootstock made of young, healthy *P. abies* seedlings. Scions should be vigorous, disease-free, with at least two to three buds, taken from the targeted genotype in late winter or early spring. The scion's wedge-shaped base is inserted into a cut made on the rootstock, ensuring close contact between the cambium layers, with no gaps. The grafting union should be secured with grafting tape or rubber bands and protected from desiccation and infection with grafting wax or a sealant. Grafts must be kept in a greenhouse or a sheltered area with moist conditions, away from direct sunlight and strong winds, until the graft is established, and new growth appears. Once the graft has taken, the grafting tape or rubber bands should be removed, and the grafted plant should be gradually acclimated to outdoor conditions.

Rooting of juvenile *P. abies* cuttings is relatively easy and has great potential for producing selected material for large planting projects. Cuttings should be harvested before bud flushing in the early spring or after the end of shoot growth in late summer, taken from young *P. abies* plants (2- to 4-year-old, but not older than 10 years) (Girouard, 1974; Stilinović, 1987). To keep the propagation material in a juvenile state several methods can be used (Girouard, 1974): periodically taking the cuttings from older rooted cuttings, pruning trees as hedges, or performing a series of grafting and cutting

operations where scions from old trees are grafted onto rootstocks, followed by cutting collection from developing shoots. Cuttings should be collected from the lower and lateral shoots as they root more easily than cuttings from upper and terminal shoots. Both lignified cuttings (taken in winter of the last season's growth and before bud flushing) and semilignified cuttings (taken in summer when shoot elongation is nearly complete and buds are forming) can be used but must be rooted before late summer. Intermittent mist must be applied. Cuttings as short as 5–8 cm can be used, but higher rooting efficiency is achieved using cuttings of 0.3–0.4 cm in diameter and 9–12 cm in length (OuYang et al., 2015). Hormone treatment can increase rooting by dipping the cutting ends in 150 mg/kg IBA for 0.5 h or in 200 mg/kg IBA for 2 h (OuYang et al., 2015).

Different materials and mixtures (usually 1:1, v:v) can be used as the rooting medium, for instance, medium-to-coarse sand alone, a mixture of peat and sand, peat and rice hulls, pear and perlite, or peat and vermiculite. During rooting, the air temperature should be maintained between 10 and 22°C while the temperature of the rooting medium should be above 13°C. The application of weak nutrient solutions to the rooting medium and the use of fungicides can enhance root and shoot formation, as well as improve survival.

The process of somatic embryogenesis of *P. abies* can be divided into the following steps (von Arnold et al., 1996): (1) initiation of somatic embryos from the primary explant, (2) proliferation of somatic embryos, (3) maturation of somatic embryos, and (4) plant regeneration. As protocols for somatic embryogenesis are too complex to be presented here, readers can find more details in von Arnold et al. (1995); Denchev and Grossnickle (2019); Farinha et al. (2020); Hazubska-Przybył et al. (2022).

Seedling cultivation

Container plants

The two most common container stock types of *P. abies* are 1-year-old seedlings (1 + 0) and 1.5-year-old seedlings (0.5 + 1) (Fig. 11.18). To produce 1 + 0 seedlings, sowing starts in a greenhouse in early March or in April (the sooner, the better) depending on the region. For producing 0.5 + 1 plants, sowing can begin in May or June. They are cultivated in a greenhouse until the end of the growing season (Szabla and Pabian, 2009; Himanen, 2018) to develop frost hardiness (Rikala, 2000). After winter, 0.5 + 1 seedlings are grown outdoors for a second growing season. Small seedlings (0.5 + 0) are another stock type. They are sown from summer until early autumn in small germination containers (3.5–13 mL) at densities of 1800–3500 seedlings/m^2. These seedlings are cold-stored (around +2°C) or frozen-stored (around −3°C) from

late autumn until spring, after which they planted directly in the field (Wallin et al., 2019).

The most used growing medium is peat, either alone or mixed with perlite or vermiculite. Commercial substrates are usually preconditioned regarding the pH and nutrient content, a factor that should be considered when designing the nursery fertilization regime. While container filling and sowing can be done manually on a small scale, mechanical processes yield better results. Mechanical compactors compress the substrate to the desired density and mechanical dibblers create cavities for precise seed placement during mechanical sowing (Ivetić, 2023). The sowing rate is one to three seeds per container cell, depending on their quality. After sowing, seeds are covered with perlite, vermiculite or other materials such as sand or sawdust. Containers are placed on metal frames to keep them together for more efficient handling and to keep them above the ground to prevent roots growing into the ground.

The optimal ambient temperature during germination is 20–22°C (Bergsten, 1987) with a reduced germination rate at temperatures above 23°C (Leinonen et al., 1992). After germination, the greenhouse air temperature should be kept at 20°C during the day and 18°C during the night (Szabla and Pabian, 2009). Seeds usually germinate within 15–20 days, and containers with seedlings should be moved outdoors after 4–8 weeks.

The containers can be hard plastic, soft plastic, or Styrofoam, with cell size depending on the targeted stock type, from 3.5 to 300 mL. The container cell volumes for production of *P. abies* seedlings 1 + 0 range between 100 and 150 mL. For 0.5 + 1 seedlings, the seeds are sown in 50 cm^3 cells where the seedlings are grown until autumn, when they are transplanted into larger volume containers of 250–300 mL (Szabla and Pabian, 2009).

The first irrigation after sowing should saturate the growing media. During germination, irrigation should keep the growth medium near field capacity with frequent, light watering events. During the rapid growth phase, irrigation should keep water content of peat at 40%–50% (v:v) (Helenius et al., 2005). The frequency of irrigation should be slightly reduced in the hardening phase.

There are many formulated soluble fertilizers available on the market for fertigation programs, but they can be also prepared in the nursery. The amount and time of fertilization should be based on electrical conductivity of growing media leakage (Ivetić, 2023). During the rapid growth phase, Ingestad (1979) suggests that nutrient optimal uptake is fulfilled at 100 mg/L N provided that the nutrient proportions within the seedlings match the proportion in the nutrient solution (N:P:K:Ca:Mg:S = 100:50:16:5:5:9). Nutrient loading by autumn fertilization is recommended to increase the nutrient content of seedlings without significant changes in seedling size (Rikala et al., 2004; Nilsson et al., 2010).

The need for controlling light differs along the production cycle. Artificial lighting should be used in the greenhouse during germination if the sowing is early, such as in March, when the days are still short (Szabla and Pabian, 2009). To induce growth cessation, a short-day (or long-night) treatment is routinely applied from early July for seedlings intended for autumn planting or winter storage. This is achieved using the blackout technique, where seedlings are covered with opaque curtains for 2–3 weeks (Nilsson et al., 2010).

Only healthy and viable seedlings should be accepted and damaged, deformed, or pest infested seedlings must be rejected. Requirements regarding the morphological attributes of seedling quality differ among countries and regions. For instance, in Polish nurseries the targeted values for 1 + 0 seedlings are 85%–90% of containers with seedlings with a minimum height of 15 cm, and a root collar diameter (RCD) of 4 mm. For 0.5 + 1 plants, the targeted values are 90%–95% of containers with seedlings reaching a minimum height of 30 cm and an RCD of 12–14 mm (Szabla and Pabian, 2009).

If the seedlings are not outplanted in the autumn, they can be stored outdoors during the winter in regions with mild and short winters, or, as practiced in Fennoscandia, placed in cardboard boxes and moved to dark freezers (Wallin et al., 2019). When storing seedlings outdoors, artificial snow should be applied if natural snow is insufficient, or the seedlings can be covered with insulating agro textile to protect them from severe frost. After winter storage, rapid thawing of seedlings results in a greater frost tolerance, higher carbohydrate contents, and later budbreak (Fløistad and Kohmann, 2001).

The risk of damping-off disease is reduced by growing *P. abies* seedlings in a low pH growing medium (Himanen, 2018). Scleroderris canker, birch rust, snow blights and *Lophodermium* needle cast need chemical control every summer (Lilja et al., 1997). Weeds should be prevented by removing the weed seed sources in and around the nursery as a good Integrated Pest Management practice. Competitive mosses are partially prevented by extending the mulching material over the seeds following sowing. In the case of weeds emerging, manual removal is the most common method of control. The current herbicide restrictions for nursery purposes in many European countries should be considered in nursery planning.

Bareroot plants

The most common bareroot stock types of *P. abies* are 2 + 0, 2 + 1, 2 + 2 in southern regions of its range (Ivetić, 2023) and 2 + 2 and 1 + 2 (grown in a greenhouse in the first year) in the northern regions (Lilja et al., 1997). Bareroot *P. abies* seedling production generally involves the following steps: (1) soil and seedbed preparation, (2) sowing, (3) culturing, (4) lifting for outplanting or transplanting, and if transplants are produced, (5) transplanting in a growing area and (6) lifting for outplanting. Each operation varies

depending on the nursery location (which is strongly affected by environmental conditions, e.g., soil, water quality), and available equipment and tools.

Soil preparation usually consists of subsoiling and plowing. Subsoiling is recommended if soil is compacted below the plow layer or there are water drainage issues. Plowing should be performed in autumn after seedlings are lifted or when preparing new fields. Granular mineral fertilizer should be applied during plowing, and organic material, mainly manure, can be incorporated depending on soil characteristics. Phosphorus (P) and potassium (K) are the primary targets of fertilization, with K in the sulfate form to avoid chlorine's negative effects on conifers (Baule and Fricker, 1978). Aldhous (1972) recommends applying 55–110 kg/ha of P and 100–130 kg/ha of K for seedbeds. Manure, applied at a rate of 10–40 Mg/ha depending on soil properties and origin, is the most used organic fertilizer to improve soil physical and chemical properties (Ivetić, 2023). If soil pH is too high, it can be lowered using ammonium-sulfate, sulfur, peat, humus, or crop rotation. A raised seedbed should be formed following harrowing and rototilling. Seedbeds are usually 1–1.3 m wide; the width depends on the machinery used for seedling cultivation.

Although seeds can be broadcast over the seedbed (Fig. 11.18), band sowing and drill sowing are preferable for mechanical cultivation and root culturing. *P. abies* seeds should be sown in early spring, when soil has warmed enough and there is no risk of late frost, usually in April. For 1 + 0 and 2 + 0 seedlings, seeds are sown in 3–6 cm wide bands with 10 cm spacing for 1 + 0 and 15 cm for 2 + 0 seedlings. Approximately 3–4 g/m of seed are sown to get 80–100 seedlings/m, or 800–1000 seedlings per m^2 (Petrović, 1952). Seed should be covered with 1.5–2.5 cm of loose soil, preferably humus-rich soil, and the seedbed rolled to press the soil around the seed. The first irrigation should follow immediately after sowing, with slow watering during germination to prevent a crust layer forming.

Cultivation usually includes shading, pest protection, irrigation, supplemental fertilization, weed control, root culturing, and transplanting. In the southern range of *P. abies*, seedbeds with recently emerged seedlings should be shaded on sunny days and protected from birds until seedlings shed their seed coats. Pest protection is similar to that used in container production. Sprinklers are used for irrigation in most nurseries, but recently drip systems are being used in some nurseries. If pH of water is above 6.8 or it contains more than 150 mg/L $CaCO_3$, water should be conditioned (Ivetić, 2023).

Supplemental fertilization mainly aims to supply Aldhous (1972) recommends applying 50–100 kg/ha of N and 0–200 kg/ha of K after sowing or transplanting divided into several smaller applications. Liquid fertilizers, such as urea, can be used for rapid N supplementation, with good results reported for conifers (Flicker, 1978). Weed control should be mechanical, or by using herbicides. Root culturing include undercutting, wrenching, and pruning (vertical or lateral, box or bench) (Grossnickle and Ivetić, 2022). It must be considered that height growth of *P. abies* is significantly greater in

Table 11.30 Observed morphological and physiological attributes and recommended values of high-quality seedlings of *Picea abies* (2 + 0 container, 2 + 0 bareroot).

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	7.8–31	>8	13.9–64	15–43
Root collar diameter (cm)	0.14–0.32	>0.2	0.3–1.4	>0.4
Shoot dry weight (g)	0.4–5.4	>3	9.5	>7
Root dry weight (g)	0.3–1.8	>1	4.1	>3
Plant dry weight (g)	0.7–7.2	>4	13.6	>9
Shoot/root dry weight ratio	1.6–4.9	2.5–3	2–3.7	1.5–2.5
Shoot [N] (mg/g)	11–21	13–23	16.1	15–24
Shoot [P] (mg/g)	1.8–3.2	1.4–3	2.1	1–3
Shoot [K] (mg/g)	7.3–12.3	5.2–12	6	5–11
Root [N] (mg/g)[a]	10.4–11.5	10–11	10.4–11.5	10–11
Root [P] (mg/g)[a]	0.63–0.74	0.65–0.7	0.63–0.74	0.65–0.7
Root [K] (mg/g)[a]	2.65–3.66	2.8–3.5	2.65–3.66	2.8–3.5

[a]Data for root nutrient concentration are for 1 + 0 seedlings.
Baule and Fricker (1978); Aldhous (1994); Zhang and George (2009); Rikala (2000); DzU (2004); Rikala et al. (2004), Jurásek et al. (2009); Heiskanen et al. (2009); Mellert and Göttlein (2012); Repáč et al. (2014); Ivetić et al. (2017); Labelle and Kammermeier (2019); Talkner et al. (2019).

the second year of seedbed cultivation than in the first (Sutton, 1967). Transplanting is needed for producing 2 + 1 and 2 + 2 stock types. Only well-developed seedlings should be transplanted, with spacing 20–25 cm between rows and 5–6 cm or 10 cm within rows for 2 + 1 plants and for 2 + 2 plants, respectively (Table 11.30).

Field establishment

P. abies is a late successional, shade-tolerant species with slow initial growth that can be further reduced by transplant shock during the establishment phase (Grossnickle, 2000). The most planted stock types are 1+ container seedlings, and due to their slow growth in both the nursery and at the planting site in the years after outplanting, 2 + 1, 1 + 2, and 2 + 2 bareroot seedlings.

P. abies can be established by direct seeding on the forestation site with recorded germination rates of 27%–72% and establishment rates of 7%–17% (reviewed by Grossnickle and Ivetić, 2017), which strongly depends on site preparation. Spring direct seeding of *Picea abies* resulted in greater seedling establishment and larger seedlings after 3 years compared with summer-sown seeds (de Chantal et al., 2004).



Figure 11.18 Left: Container *Picea abies* seedling. Right: Bareroot *P. abies* seedlings in raised beds. *(Vladan Ivetić.)*

P. abies seedlings are sensitive to competition, which makes good site preparation and vegetation control necessary. Mechanical site preparation (MSP) is recommended. In the mineral soils of Fennoscandia, MSP can increase survival by 15%–20% over a period of 10 years (Sikström et al., 2020). Recommended MSP techniques include patch scarification, disc trenching, mounding, soil inversion, and plowing. Site and soil disturbance should be considered when selecting the MSP method, as recent studies have shown that site preparation intensity had no effect on *P. abies* seedling growth and survival, while the MSP methods differed in soil disturbance (Nordin et al., 2022).

Seedlings are usually planted in spring and early autumn, though container seedlings can be successfully planted in summer without apparently negative effects on field performance (Luoranen et al., 2006) provided they are properly handled from nursery to the planting site and well-watered at planting (Helenius et al., 2005). In Finland, freeze-stored seedlings can be used until mid-June (Nilsson et al., 2010).

As in the nursery, outplanted seedlings are susceptible to frost damage, which can significantly reduce seedling growth, especially if damage persists over several years, though survival is not significantly affected (Langvall et al., 2001). Wildlife, such as roe deer (*Capreolus capreolus*) and red deer (*Cervus elaphus*) can cause severe damage to planted seedlings through browsing and bark stripping (Nilsson, 2020). Plants can be protected either by repellents or by fencing. *P. abies* is also susceptible to damage from the pine weevil (*Hylobius abietis* L.), a major pest of newly planted conifer seedlings

in Europe. Sand-coating stems and spraying insecticides such as lambda-cyhalothrin and acetamiprid (commonly used in Finland) in the nursery prior to delivery significantly reduces weevil damage (Luoranen et al., 2022).

Chapter References

Aldhous, J.R., 1972. Nursery Practice. Forestry Commission Bulletin No. 43, pp. 11–40. London.

Aldhous, J.R., 1994. Nursery policy and planning. In: Aldhous, J.R., Mason, W.L. (Eds.), *Forest Nursery Practice*. Bulletin 111 of the Forestry Commission. HMSO., pp. 104–111

Baule, H., Fricker, C., 1978. Đubrenje Šumskog Drveća, Jugoslovenski Poljoprivredno-Šumarski Centar – Služba Šumske Proizvodnje, Beograd, Str, pp. 130–140.

Bergsten, U., 1987. Incubation of *Pinus Sylvestris* L. And *Picea abies* (L.) Karst. Seeds at Controlled Moisture Content as an Invigoration Step in the IDS Method. Dissertation. Swedish University of Agricultural Sciences, Department of Silviculture, 98 p.

Damian, I., 1978. Impaduriri (Afforestations). Editura Didactica si Pedagogica. Bucarest, Romania, 374 p.

de Chantal, M., Leinonen, K., Ilvesniemi, H., Westman, C.J., 2004. Effects of site preparation on soil properties and on morphology of *Pinus sylvestris* and *Picea abies* seedlings sown at different dates. N. For. 27, 159–173. https://doi.org/10.1023/A:1025042632491.

Denchev, P., Grossnickle, C.S., 2019. Somatic embryogenesis for conifer seedling production. Reforesta 7, 109–137. https://doi.org/10.21750/REFOR.7.08.70.

DzU, 2004. Rozporządzenie Ministra Środowiska Z Dnia 18 Lutego 2004 R. W Sprawie Szczegółowych Wymagań, Jakie Powinien Spełniać Leśny Materiał Rozmnożeniowy. https://isap.sejm.gov.pl/isap.nsf/DocDetails.xsp?id=WDU20040310272.

Farinha, A., Zufferey, R., Zheng, P., Armanini, S.F., Kovac, M., 2020. Unmanned aerial sensor placement for cluttered environments. IEEE Rob. Autom. Lett. 5 (4), 6623–6630. https://doi.org/10.1109/LRA.2020.3015459.

Fløistad, I.S., Kohmann, K., 2001. Effects of thawing procedure on frost hardiness, carbohydrate content and timing of bud break in *Picea abies*. Scand. J. For. Res. 16 (1), 30–36. https://doi.org/10.1080/028275801300004389.

Girouard, R.M., 1974. Propagation of spruce by stem cuttings. New Zealand J. Fore. Sci. 4 (2), 140–149.

Grossnickle, S.C., 2000. Ecophysiology of northern spruce species: The performance of planted seedlings. NRC Research Press, Ontario, Canada.

Grossnickle, S.C., Ivetić, V., 2022. Root system development and field establishment: effect of seedling quality. N. For. 53, 1021–1067. https://doi.org/10.1007/s11056-022-09916-y.

Hazubska-Przybył, T., Wawrzyniak, M.K., Kijowska-Oberc, J., Staszak, A.M., Ratajczak, E., 2022. Somatic embryogenesis of Norway spruce and Scots pine: possibility of application in modern forestry. Forests 13, 155. https://doi.org/10.3390/f13020155.

Heiskanen, J., Lahti, M., Luoranen, J., Rikala, R., 2009. Nutrient loading has a transitory effect on the nitrogen status and growth of outplanted Norway spruce seedlings. Silva Fenn. 43, 210. https://doi.org/10.14214/sf.210.

Helenius, P., Luoranen, J., Rikala, R., 2005. Physiological and morphological responses of dormant and growing Norway spruce container seedlings to drought after planting. Ann. For. Sci. 62 (3), 201–207. https://doi.org/10.1051/forest:2005011.

Himanen, K., 2018. Seed quality attributes in seedling production of Norway spruce (*Picea abies* (L.) Karst.). Dissertat. Forest. 261. https://doi.org/10.14214/df.261, 74 p.

Hoffmann, J., Chvalova, K., Palatova, E., 2007. Lesne Semenarstvo Na Slovensku. Itgamma, s.r.o Sliač, 195 pp.

Houšková, K., Klepárník, J., Mauer, O., 2021. How to accelerate the germination of Scots pine and Norway spruce seeds? J. For. Sci. 67, 134–142.

Iancu, I., 1999. Manualul padurarului. Teoria si practica meseriei (Manual of Forest Ranger. Theory and Practice). Tipografia Pegasus, Bucuresti, Romania, 622 p.

Ingestad, T., 1979. Mineral nutrient requirements of *Pinus silvestris* and *Picea abies* seedlings. Physiol. Plant. 45, 373–380. https://doi.org/10.1111/j.1399-3054.1979.tb02599.x.

Ivetić, V., 2023. Forest Reproductive Material – Biology and Technology of Production of Forest Trees Seed and Seedlings. University of Belgrade – Faculty of Forestry, Belgrade, 304 pp.

Jovković, B.J., 1952. Šumsko Semenarstvo I Rasadnici. Svjetlost, Sarajevo, 80 pp.

Jurásek, A., Leugner, J., Martincová, J., 2009. Effect of initial height of seedlings on the growth of planting material of Norway spruce (*Picea abies* [L.] Karst.) in mountain conditions. J. For. Sci. 55 (3), 112–118. https://doi.org/10.17221/97/2008-JFS.

Labelle, E.R., Kammermeier, M., 2019. Above- and belowground growth response of *Picea abies* seedlings exposed to varying levels of soil relative bulk density. Eur. J. For. Res. 138, 705–722. https://doi.org/10.1007/s10342-019-01201-6.

Langvall, O., Nilsson, U., Örlander, G., 2001. Frost damage to planted Norway spruce seedlings — influence of site preparation and seedling type. For. Ecol. Manag. 141 (3), 223–235. https://doi.org/10.1016/S0378-1127(00)00331-5.

Leinonen, K., Nygren, M., Rita, H., 1992. Temperature control of germination in the seeds of *Picea abies*. Scandinav. J. Forest. Res. 8, 107–117.

Lilja, A., Lilja, S., Kurkela, T., Rikala, R., 1997. Nursery practices and management of fungal diseases in forest nurseries in Finland. A review. Silva Fenn. 31 (1), 79–100.

Luoranen, J., Laine, T., Saksa, T., 2022. Field performance of sand-coated (Conniflex®) Norway spruce seedlings planted in mounds made by continuously advancing mounder and in undisturbed soil. For. Ecol. Manag. 517, 120259.

Luoranen, J., Rikala, R., Konttinen, K., Smolander, H., 2006. Summer planting of *Picea abies* container-grown seedlings: effects of planting date on survival, height growth and root egress. For. Ecol. Manag. 237 (1), 534–544. https://doi.org/10.1016/j.foreco.2006.09.073.

Mellert, K.H., Göttlein, A., 2012. Comparison of new foliar nutrient thresholds derived from van den Burg's literature compilation with established central European references. Eur. J. For. Res. 131, 1461–1472. https://doi.org/10.1007/s10342-012-0615-8.

Milev, M., Petkova, K., Aleksandrov, P., Iliev, N., 1999. Posevni Materiali Ot Iglolistni Vidove. LTU, Sofia, Bulgaria.

Nilsson, O., 2020. Establishment and Growth of Scots Pine and Norway Spruce A Comparison between Species. PhD THESIS. Swedish University of Agricultural Sciences Alnarp Acta Universitatis Agriculturae Sueciae 71. https://pub.epsilon.slu.se/18698/1/Oscar%20Nilsson.pdf.

Nilsson, U., Luoranen, J., Kolström, T., Örlander, G., Puttonen, P., 2010. Reforestation with planting in northern Europe. Scand. J. For. Res. 25 (4), 283–294. https://doi.org/10.1080/02827581.2010.498384.

Nordin, P., Olofsson, E., Hjelm, K., 2022. Successful spruce regenerations – impact of site preparation and the use of variables from digital elevation models in decision-making? Scand. J. For. Res. 37 (1), 33–44. https://doi.org/10.1080/02827581.2022.2028895.

OECD, 2006. Section 3 - Norway spruce (*Picea abies* (L.) KARST). In: Safety Assessment of Transgenic Organisms, Volume 2: OECD Consensus Documents, OECD Publishing, Paris. https://doi.org/10.1787/9789264095403-4-en.

OuYang, F., Wang, J., Li, Y., 2015. Effects of cutting size and exogenous hormone treatment on rooting of shoot cuttings in Norway spruce [*Picea abies* (L.) Karst.]. N. For. 46, 91–105. https://doi.org/10.1007/s11056-014-9449-1.

Petrović, D., 1952. Rad U Šumskim Rasadnicima. Izdanje Glavne uprave za šumarstvo NRS.

Regent, B., 1980. Šumsko semenarstvo. u: Dokumentacija za tehniku i tehnologiju u šumarstvu, Beograd: Jugoslovenski poljoprivredno-šumarski centar, br. 7.

Repáč, I., Balanda, M., Vencurik, J., Kmet, J., Krajmerová, D., Paule, L., 2014. Effects of substrate and ectomycorrhizal inoculation on the development of two-years-old container-grown Norway spruce (*Picea abies* Karst.) seedlings. iForest 8, 487–496. https://doi.org/10.3832/ifor1291-007.

Rikala, R., 2000. Planting stock production. In: Mälkönen, E., Babich, N.A., Krutov, V.I., Markova, I.A. (Eds.), Forest Regeneration in the Northern Parts of Europe. Proceedings of the Finnish-Russian Forest

Regeneration Seminar in Vuokatti, Finland Sept. 28th – Oct. 2nd, 1998. Metsäntutkimuslaitoksen Tiedonantoja 790, pp. 127–146. http://urn.fi/URN:ISBN:951-40-1757-9.

Rikala, R., Heiskanen, J., Lahti, M., 2004. Autumn fertilization in the nursery affects growth of *Picea abies* container seedlings after transplanting. Scand. J. For. Res. 19 (5), 409–414. https://doi.org/10.1080/02827580410030190.

Sikström, U., Hjelm, K., Holt Hanssen, K., Saksa, T., Wallertz, K., 2020. Influence of mechanical site preparation on regeneration success of planted conifers in clearcuts in Fennoscandia – a review. Silva Fenn. 54 (2), 10172. https://doi.org/10.14214/sf.10172.

Stilinović, S., 1985. Semenarstvo šumskog i ukrasnog drveća i žbunja. In: Univerzitet U Beogradu. Šumarski Fakultet, Beograd.

Stilinović, S., 1987. Proizvodnja sadnog materijala šumskog i ukrasnog drveća i žbunja. In: Univerzitet U Beogradu. Šumarski Fakultet, Beograd.

Suszka, B., Chmielarz, P., Walkenhorst, R., 2005. How long can seeds of Norway spruce (*Picea abies* (L.) Karst.) be stored? Ann. For. Sci. 62 (1), 73–78. https://doi.org/10.1051/forest:2004082.

Sutton, R.F., 1967. Influence of root pruning on height increment and root development of outplanted spruce. Can. J. Bot. 45 (9), 1671–1682. https://doi.org/10.1139/b67-174.

Szabla, K., Pabian, R., 2009. Szkółkarstwo kontenerowe - Nowe technologie i techniki w szkółkarstwie leśnym. In: Centrum Informacyjne Lasów Pañstwowych.

Talkner, U., Riek, W., Dammann, I., Kohler, M., Gottlein, A., Mellert, K.H., Meiwes, K.L., 2019. Nutritional status of major forest tree species in Germany. In: Wellbrock, N., Bolte, A. (Eds.), Status and Dynamics of Forests in Germany, Ecological Studies, vol 237. Springer, Cham. https://doi.org/10.1007/978-3-030-15734-0_9.

Tikkinen, M., Varis, S., Peltola, H., Aronen, T., 2017. Norway spruce emblings as cutting donors for tree breeding and production. Scand. J. For. Res. 33 (3), 207–214. https://doi.org/10.1080/02827581.2017.1349925.

Vlase, I., 1982. Conservarea Semintelor Forestiere (Conservation of Forest Seeds). Editura Ceres, Bucuresti, Romania, p. 278.

von Arnold, S., Clapham, D., Egertsdotter, U., Mo, L.H.,, 1996. Somatic embryogenesis in conifers—a case study of induction and development of somatic embryos in *Picea abies*. Plant Growth Regul. 20, 3–9. https://doi.org/10.1007/BF00024050.

von Arnold, S., Egertsdotter, U., Ekberg, I., Gupta, P., Mo, H., Nørgaard, J., 1995. Somatic embryogenesis in Norway spruce (*Picea abies*). In: Jain, S., Gupta, P., Newton, R. (Eds.), Somatic Embryogenesis in Woody Plants, vols 44–46. Springer, Dordrecht. https://doi.org/10.1007/978-94-011-0960-4_2.

Wallin, E., Gräns, D., Stattin, E., Verhoef, N., Mikusiński, G., Lindström, A., 2019. Evaluating methods for storability assessment and determination of vitality status of container grown Norway spruce transplants after frozen storage. Scand. J. For. Res. 34 (6), 417–426. https://doi.org/10.1080/02827581.2019.1622036.

Zhang, J.l., George, E., 2009. Rhizosphere effects on ion concentrations near different root zones of Norway spruce (Picea abies (L.) Karst.) and root types of Douglas-fir (Pseudotsuga menziesii L.) seedlings. Plant Soil 322, 209–218. https://doi.org/10.1007/s11104-009-9909-0.

Picea omorika (Pančić) Purkyně

Milan Mataruga and Branislav Cvjetković
Faculty of Forestry, University of Banja Luka, Republic of Srpska, Bosnia and Herzegovina

Propagation

Seed propagation

To date, 35 species of the *Picea* genus have been described, mainly in the northern hemisphere. However, only two species are found in Europe: *Picea abies* (L.) Karst. (Norway spruce), which is widespread across almost the entire European continent and is economically very important in forestry, and *Picea omorika* Pančić/Purkyně (Serbian spruce), a tertiary relict and endemic species (Fig. 11.19). The Serbian spruce finds refuge in the Drina River canyon, marking the border between the Republic of Serbia and Republic Srpska/Bosnia and Herzegovina.

Picea omorika shares many seed and seedling production characteristics with *P. abies*. Cones ripen in October of the first year. Depending on weather conditions, they can open during autumn or spring. Cones can be harvested from October to February or March of the following year. A maximum of 3 kg of cones can be collected per tree (Jevtić, 1960),



Figure 11.19 Crown structure of the *Picea omorika* tree in its natural habitat (Left) and collected *P. omorika* cones (Right). *(Milan Mataruga)*

Table 11.31 Characteristics of seed lots of *Picea omorika*.

Cone to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
100 kg cones = 1.5 kg seeds (1%)	99	420,000	90

Stilinović (1985); Pintarić (1957); Jevtić (1960).

but in our experience the actual amount is even less; 1.9 kg was most cones we collected from one tree in a study with 111 trees (Mataruga and Cvjetković, 2023).

Collecting cones is very difficult due to the pronounced pyramidal form of the tree (Fig. 11.19), making collecting almost impossible by climbing the trees. Therefore, cone collection is usually organized by climbing neighboring trees (usually *Pinus nigra*) and picking cones from the tops of *P. omorika* trees. The characteristics of the seeds are presented in Table 11.31. One hectoliter (100 L) of cones weighs about 67 kg, and about 1.5 kg of seeds can be harvested per 100 kg of cones. The mass of 1000 seeds is 2.4 g (Stilinović, 1985).

Cones are processed in the same way as for *P. abies* except that drying at 48°C should be longer. The mean germination rate of fresh seeds is high (Table 11.31), and the germination rate of seeds stored in glass jars at room temperature remains 90%–95% (Pintarić, 1957). Germination is tested at a temperature of 20–30°C for 7–21 days. If seeds are dried to low humidity (i.e., 5–10%) and stored at −10°C, they maintain high germination rates for more than 10 years. No presowing treatment is required.

Vegetative propagation

There is little information on the vegetative propagation of *P. omorika*. Vegetative propagation by rooting of the lowest branches (Fig. 11.20), however, has been observed in nature and described by several authors (Fukarek, 1951; Čolić, 1953, 1959).

Traditional methods of vegetative propagation for this species have been partially successful. Grafting is also possible but infrequently used. In our research (unpublished results), cuttings were taken at three different times: before shoot elongation in early April, at the onset of the vegetation movement phase, and during the secondary growth phase. In all cases, cuttings were taken from the upper and lower parts of the crown in 23 trees, with lengths ranging from 5–10 cm (Fig. 11.19). The cut ends were powdered with 2% indole-butyric acid, a phytohormone that stimulates root growth. Previously, cuttings were fumigated with the fungicide "Pinozeba" and then placed on rooting substrate (sand without additional temperature regulation). Rooting success was less than 1%; all cuttings were obtained from the same parent tree. In contrast, Međedović (1986, 1987) used cuttings from young trees harvested in spring and treated them with 1% indole-3-acetic acid, achieving a rooting rate greater than 70%.

Given the low success with rooting cuttings and grafting, many authors have investigated propagation by tissue culture. Budimir and Vujičić (1992) and Budimir et al. (1998) describe in vitro methods for the preservation and propagation of this endemic species. The possibilities of *P. omorika* propagation by tissue culture are well documented (Vujičić and Budimir, 1995; Kolevska-Pletikapić and Buturović-Derić, 1995; Tramišak-Milaković et al., 1999; Mihaljević et al., 2001; Mihaljević and Jelaska, 2005; Leljak-Levanić et al., 2009; Hazubska-Przybył et al., 2010).

Seedling cultivation

Container plants

There is no published information on the container production of *P. omorika*. Due to the small number of seedlings being produced, and the similarity with *P. abies*, we recommend the cultivation method for *P. abies* described elsewhere in this volume. The first production of *P. omorika* seedlings in containers at the level of half-sib lines (in Bosnia and Herzegovina) is being carried out in 2023–24 (Fig. 11.20).

Bareroot plants

P. omorika seedlings grow more slowly and are more sensitive in the juvenile phase to stress factors than *P. abies* seedlings. Sowing is done in the spring after the danger of late frosts has passed and when the soil is warm enough. The seeds are sown without prior treatment. To achieve better germination, seeds are soaked in water for 24 h immediately before sowing. Nurseries in Bosnia and Herzegovina treat the seeds with pesticides before sowing to protect them from birds and rodents.

Since the seeds are very small (Table 11.32), sowing must be shallow (up to 1 cm deep), and then covered with sand or mulch (usually sawdust). Due to the slow growth of this species, only two-year-old or older seedlings are produced. Watering, shading, feeding, and control of pathogens, pests, and weeds are mandatory. Irrigation is carried out by automated systems connected to a system of piezometers that register the level of soil saturation and activate the irrigation system if soil moisture is suboptimal for growth and development of the seedlings. Shading is recommended against high insolation. Insecticide can be applied depending on national pesticide regulations. In addition, beds can be sterilized using hot water steam, which is recommended as a nature-friendly approach.

Fertilization depends on the level of technological equipment of the nursery. Fertilization is done three or more times during the first growing season. Complex mineral fertilizers are most often used annually as a starter fertilizer, such as NPK 10:20:30 at a rate of 700 kg/ha. Also, NPK 15:15:15 at 500 kg/ha can be used. The final fertilization employs a fertilizer rich in potassium and phosphorus, with low nitrogen content. Fertilization is usually stopped at the beginning of July to facilitate the lignification of the



Figure 11.20 *Above:* Cuttings of *Picea omorika* (left) being rooted in a sand bed (right). *Below: P. omorika* seedlings during the first growing season with marked half-sib lines (left bed) and *P. abies* seedlings (right bed). *(Milan Mataruga)*

Table 11.32 Measured morphological attributes of different stock types of bareroot *Picea omorika* seedlings.

Attributes	**2 + 0**	**3 + 0**	**2 + 1**	**2 + 2**	**2 + 3**
Shoot height (cm)	5.4 to 14.0	9.1	~18.0	~28	49.9
Root collar diameter (cm)	0.1	0.16			1.08
Root length (cm)	10.9	14.0			36.2

Jevtić (1962); Isajev (1987).

stem, allowing it to harden off. In the second season, fertilization and weeding are carried out similar to the first year.

Picea omorika seedlings emerge after 20–30 days. Because the seedlings are susceptible to attack by *Fusarium* species, plants are treated every 7 days with fungicides based on copper and sulfur until the first needles are formed. Copper sulfate (0.5%–1%) and copper oxychloride (2%) in a dose of 5–6 L/m^2 are mainly used. In addition, weeds are regularly controlled with herbicides approved by local authorities.

After 2 years, the seedlings are transplanted into the growing beds that have been prepared in the same way as described for the sowing procedure. Soil pH should not be higher than 5.2 as seedling growth is reduced if the soil is too basic (Marković and Marković, 1987). Seedlings are transplanted when dormant, usually between October and November. Due to slow growth, they usually remain for another two or 3 years (2 + 2 or 2 + 3). Fertilizer application and weed control have a similar pattern to that of seedlings before transplanting. The fertilizer rate is 50 kg/ha in the first two applications, with the rate increasing in the third application to 400–500 kg/ha of NPK 7:20:30. It is not necessary to prune the root system, and storage and delivery are carried out as for *P. abies* (Stilinović, 1987).

Field establishment

There is limited information about this species in plantations. Tests of artificial regeneration through sowing and different site preparations have shown none or poor germination and establishment in open-field tests (Dinić, 1989a, 1994a, 1994b), whereas better results were achieved in forested habitats (Dinić, 1988, 1989a, 1989b, 1991a, 1991b). A significant issue lies in the amount of light required for seedling development in early stages, which may explain the regeneration challenges (Tucić et al., 2005, 2006). Nevertheless, two-year-old bare root seedlings fare well in forests at lower altitudes, such as the zone of beech forests (Dinić et al., 1994c). Information on planting techniques, seedling survival, soil fertilization, protection methods against browsing and climate conditions, competition control, and irrigation is currently lacking.

The main environmental limitations for the establishment of *P. omorika* seedlings have not been thoroughly investigated. There is a known problem of natural regeneration in native forests. It has been speculated that its regeneration is linked to disturbances such as fires. *P. omorika* is a poor competitor compared with other coexisting trees such as *P. abies, Pinus* spp., and *Fagus sylvatica* (Wardle, 1956).

Currently, *P. omorika* is primarily used as a horticultural species and it has long been recognized as one of the most beautiful conifers (Wyman, 1936; Haigh, 1967). Almost every park, arboretum, or botanical garden in Europe boasts several specimens of this species (Kasesalu, 2002; Král, 2002; Isajev et al., 2016; Ballian et al., 2016). It is highly tolerant of air pollution, thrives in a wide range of habitats, requires no specific soil

conditions, and can be planted almost anywhere. While it grows well in various habitats (Meyer, 1962; Bambič et al., 2020), it performs best in shaded locations.

Chapter References

Ballian, D., Ravazzi, C., Caudullo, G., 2016. Picea Omorika in Europe: Distribution, Habitat, Usage and Threats. European Atlas of Forest Tree Species. Publications Office of the European Union, Luxembourg, p. 157.

Bambič, S., Jarni, K., Božič, G., Brus, R., 2020. Growing of spruce (*Picea omorika* (Pančić) Purk.) in the generative seed plantation Počivalnik near Postojna. Acta Silvae et Ligni 123, 1–11.

Budimir, S., Vujičić, R., 1992. Benyzladenine induction of buds and somatic embryogenesis in *Picea omorika* (Panč.) Purk. Plant Cell Tiss. Org. Cult. 31, 89–94.

Budimir, S., Stojičić, D., Ćulafić, L., Vujičić, R., 1998. Regeneration of *Picea omorika* and *Pinus heldreichii* by tissue culture methods. In: Tsekos, I., Moustakas, M. (Eds.), Progress in Botanical Research. Springer, Dordrecht, pp. 469–472. https://doi.org/10.1007/978-94-011-5274-7_106.

Čolić, D., 1953. Staništa Pančićeve Omorike Na Desnoj Strani Drine (In Serbian/Serbian Spruce Habitats on the Right Side of the Drina River). Naučna knjiga, Beograd.

Čolić, D., 1959. Prilog poznavanja ekologije vegetativnog razmnožavanja Pančićeve omorike. (in Serbian/A contribution to knowledge of the ecology of vegetative propagation of Serbian spruce). Arhiv Bioloških Nauka II (1–4), 41–66.

Dinić, A., 1988. Uticaj korenovog sistema drveća i crnjuše (Erica carnea L.) na klijavost semena i prve faze rasta Pančićeve omorike u reliktnoj zajednici smrče, omorike i crnog bora na planini Tari. Zbornik radova sa simpozijuma "Proučavanje biljnog i životinjskog sveta sa aspekta problema zaštite i unapređenja životne sredine 45–53 (Kragujevac).

Dinić, A., 1989a. Eksperimentalna ispitivanja klijavosti semena Pančićeve omorike na različitim staništima u rezervatu Crveni potok na planini Tari. Zaštita Prirode 41–42, 87–95.

Dinić, A., 1989b. The influence of tree root system and the whortleberry (*Vaccinium myrtillus* L.) on the seed germinability of the Serbian spruce (*Picea omorika* (Pančić) Purkyne) in the relict forest of Norway and Serbian spruce, common silver fir and beech on the Tara Mt. Arhiv. Biol. Nauka 4 (1–2), 1P–2P.

Dinić, A., 1991a. Eksperimentalno ispitivanje klijavosti semena omorike (*Picea omorika* Pančić) u reliktnim šumama na planini Tari. Simpozijum Nedeljko Košanin i botaničke nauke 169–174 (Ivanjica).

Dinić, A., 1991b. Ogledi sa setvom semena omorike u rezervatu Crvene stene i rasadniku na planini Tari. Zaštita prirode 43–44, 91–103. Beograd.

Dinić, A., 1994a. Eksperimentalna Fitocenologija Šumskih Ekosistema Srbije. Matica srpska, Novi Sad.

Dinić, A., 1994b. Klijanje semena i preživljavanje ponika omorike u šumskim rezervatima Nacionalnog parka Tara; Monografska studija Omorika - Picea omorika (Pančić) Purkyne na području Nacionalnog parka Tara. Bajina Bašta.

Dinić, A., Sijak, M., Kalinic, M., 1994c. Experiments with seed sowing and planting the 2-year plants of the Serbian spruce (Picea omorika Pancic/Purkyne) in different vegetation zones on the Kopaonik Mt. [Serbia, Yugoslavia]. Sumarstvo (Yugoslavia).

Fukarek, P., 1951. Današnje rasprostranjenje Pančićeve omorike (Picea omorika Pančić) i neki podaci o njenim sastojinama. (in Serb./Nowaday distribution of Serbian spruce (Picea omorika Pančić) and some information about its stands). Godišnjak Biol. Instituta u Sarajevu br. 1–2, 141–198.

Haigh, R., 1967. Picea omorika - one of the most ornamental conifers. Arboricultural Assoc. J. 1 (5), 125–127.

Hazubska-Przybył, T., Chmielarz, P., Michalak, M., Bojarczuk, K., 2010. Cryopreservation of embryogenic tissues of *Picea omorika* (*Serbian spruce*). Plant Cell Tiss. Organ Cult. 102, 35–44. https://doi.org/10.1007/s11240-010-9701-0.

Isajev, V., 1987. Oplemenjivanje Omorike (Picea Omorika/Panč./Purkyne) Na Genetičko-Selekcionim Osnovama. Beograd: Šumarski Fakultet, Doktorska Disertacija/Serbian Spruce (Picea Omorika/Panč./Purkyne) Breeding on Genetic-Selection Basis. Faculty of Forestry, doctoral dissertation, Belgrade.

Isajev, V., Lavadinović, V., Popović, V., Lučić, A., 2016. Selection and breeding of Serbian spruce (Picea omorika) for urban areas in Serbia. In: Proceedings of the 17th International Symposium on Landscape Ecology, pp. 306–311.

Jevtić, J., 1960. Neka zapažanja o urodu semena Pančićeve omorike na Tari. (in Serb. Some observations about Serbina spruce seed yield on Tara mountin). Šumarstvo Br 1–2, 79–85.

Jevtić, J., 1962. Uporedna ispitivanja prirasta i produkcije biomase omorike, smrče i sitke u rasadničkim uslovima. (in Serb. Comparative studies of growth and biomass production of Norway, Serbian and Sitka spruce in nursery conditions). Šumarstvo 3–4, 147–158.

Kasesalu, H., 2002. Serbian spruce (Picea omorika (Panchic) purkyne) in Estonia. Dendrolog. Res. Estonia III (Estonia).

Kolevska-Pletikapić, B., Buturović-Derić, Z., 1995. Regeneration of Picea omorika plants via organogenesis. Plant Cell Tiss. Organ Cult. 41, 189–192. https://doi.org/10.1007/BF00051590.

Král, D., 2002. Assessing the growth of Picea omorika [Panč.] Purkyně in the Masaryk forest training forest enterprise at Křtiny. J. For. Sci. 48, 388–398.

Leljak-Levanić, D., Mihaljević, S., Jelaska, S., 2009. Variations in DNA methylation in *Picea omorika* (Panč) Purk. Embryogenic tissue and ability for embryo maturation. Propag. Ornam. Plants 9 (1), 3–9.

Marković, L., Marković, D., 1987. Suitability of Substrate with the Organic Litter Component from Our Forests for Growing Serbian Spruce (*Picea omorika* Panc.) Seedlings in Dunemann-Beds [Yugoslavia]. Zbornik radova-Institut za sumarstvo i drvnu industriju (Yugoslavia).

Mataruga, M., Cvjetković, B., 2023. Laboratorijska klijavost 101 linije polusrodnika Pančićeve omorike. X Simpozijum društva selekcionere i semenara Republike Srbije i VII simpozijum sekcije za oplemenjivanje organizama društva genetičara Srbije. Zbornik apstrakata 201–0202.

Međedović, S., 1986. Ožiljavanje reznica omorike, munike i molike u cilju razvijanja tehnologije masovne proizvodnje sadnica. (in Serb. Root cuttings of Serbina spruce, Bosnian and Macedonian pine in order to develop the technology of mass production of seedlings). Zbornik rezimea sa VII Kongresa biologa Jugoslavije. Budva 37.

Međedović, S., 1987. Physiological induction of the rhizogenesis in the auto vegetative propagation of forest tree cuttings. In: Zbornik Rezimea Sa III Simpozijuma Jugoslovenskog Društva Za Fiziologiju Biljaka. Tuheljske Toplice, p. 57.

Meyer, H., 1962. Results of trial plantings of Picea omorika. Mitt. Dtsch. Dendrol. Ges. 61, 68–70.

Mihaljević, S., Jelaska, S., 2005. Omorika spruce (Picea omorika). In: Jain, S.M., Gupta, P.K. (Eds.), Protocol for Somatic Embryogenesis in Woody Plants, Forestry Sciences, vol 77. Springer, Dordrecht. https://doi.org/10.1007/1-4020-2985-3_4.

Mihaljević, S., Perić, M., Jelaska, S., 2001. The sensitivity of embryogenic tissue of *Picea omorika* (Panč.) Purk. to antibiotics. Plant Cell Tiss. Organ Cult. 67, 287–293. https://doi.org/10.1023/A:1012760403379.

Pintarić, K., 1957. Uticaj Starosti Sjemena I Djelovanje Svjetla Na Proces Klijanja Kod Sjemena Pančićeve Omorike Picea Omorika Panč. (In Serb. The Influence of Seed Age and the Effect of Light on the Germination Process of Serbian Spruce), vol 2. Radovi Poljoprivredno-šumarskog fakulteta Univerziteta u Sarajevu. God II, Br., pp. 89–104

Stilinović, S., 1985. Semenarstvo Šumskog I Ukrasnog Drveća I Žbunja. Univerzitet U Beogradu, Šumarski Fakulte, Udžbenik, pp. 130–131. /Seed production of forest and ornamental trees and shrubs. University of Belgrade, Faculty of Forestry, Textbook).

Stilinović, S., 1987. Proizvodnja Sadnog Materijala Šumskog I Ukrasnog Drveća I Žbunja. Univerzitet U Beogradu, Šumarski Fakulte, Udžbenik, 130-131./Seedling production of forest and ornamental trees and shrubs. University of Belgrade, Faculty of Forestry, Textbook.

Tramišak-Milaković, T., Mihaljević, S., Jelaska, S., 1999. Effects of abscisic acid and carbohydrates on the maturation of Picea omorika (Panč.) Purk. somatic embryos. Acta Bot. Croat. 58, 87–97.

Tucić, B., Dučić, J., Pemac, D., 2006. Phenotypic responses to early signals of neighbour proximity in *Picea omorika*, a pioneer conifer tree. Basic Appl. Ecol. 7 (5), 443–454.

Tucić, B., Pemac, D., Dučić, J., 2005. Life history responses to irradiance at the early seedling stage of *Picea omorika* (Pančić) Purkyňe: adaptiveness and evolutionary limits. Acta Oecol. 27 (3), 185–195.

Vujičić, R., Budimir, S., 1995. Somatic embryogenesis and plant regeneration in *Picea omorika*. In: Jain, S., Gupta, P., Newton, R. (Eds.), Somatic Embryogenesis in Woody Plants, vol 3. Kluwer Academic Publishers, Printed in Netherlands, pp. 81–97.
Wardle, P., 1956. Picea omorika in its natural habitat. Forestry 29 (2), 91–117.
Wyman, D., 1936. A few evergreens. Bullet. Pop. Informat. (Arnold Arboretum, Harvard University) 4 (15), 91–94.

Pinus brutia Ten.

Marianthi Tsakaldimi[1] and Nebi Bilir[2]
[1]Laboratory of Silviculture, Department of Forestry and Natural Environment, Aristotle University of Thessaloniki, Thessaloniki, Greece; [2]Faculty of Forestry, Isparta University of Applied Sciences, Isparta, Türkiye

Propagation

Seed propagation

Pinus brutia Ten. (Brutia or red pine) is known for producing numerous serotinous cones, small seeds, and its early reproduction (Ganatsas et al., 2021). The timing of harvesting is important as it affects the quality and quantity of *P. brutia* seeds. A regular production of fully developed cones starts from 10 years in plantations to 15–20 years in natural stands (Gezer, 1986). Cones need 3 years to ripen (Ganatsas et al., 2021). The optimal time of harvesting is when the cones turn brown, usually by the end of spring (Thanos and Daskalakou, 1993; Boydak, 2004; Ganatsas et al., 2011). Harvesting cones before full ripening results in lower seed quality. Cone opening can be achieved by thermal treatment at 45–72 °C in an oven, for 3 days (Ganatsas et al., 2011). Alternatively, cones can be warmed to 40–45°C, moistened for a few hours, and then reheated to 40–45°C for seed extraction (Aslan, 2001a). The seed-to-cone fresh weight proportion is 4%–5% (Aslan, 2001a) and the proportion of filled seeds ranges from 88% to 92% (Boydak et al., 2006).

Pinus brutia seeds are orthodox so they should be stored under dry conditions, preferably at a temperature of 1–5°C (Skordilis, 1992). The optimal seed moisture content for storage is below 10%, being usually between 6.8% and 10%. Seeds with high moisture content are prone to fungal and bacterial infections, which can reduce seed viability and quality. Seed characteristics of seed lots are summarized in Table 11.33, but the main characteristics can show significant variation among provenances (Sefik, 1964; Yilmaz et al., 2013). *P. brutia* seeds are typically small and dark brown. The weight of one seed ranges between 30 and 31.1 mg, with a mean weight of 30.2 mg. A sample of 1000 seeds weighs 51.9 g, ranging between 41.0 and 66.2 g (Aslan, 2001a). Seed size

Table 11.33 Characteristics of seed lots of *Pinus brutia*. The values of germination capacity are under controlled environment.

N° fully developed seeds per cone	Purity (%)	No. seeds/kg	Germination capacity (%)
39–98 (69)	98.5–99.4 (99)	16,340–33,104 (24,720)	65–91 (85)

Mean values in brackets.
Sefik (1964), Panetsos (1981), Thanos and Marcou (1991), Skordilis (1992), Thanos and Daskalakou (1993), Spanos et al. (2000), Paitaridou et al. (2005), Chambel et al. (2013), Yilmaz et al. (2013), Ganatsas et al. (2021).

and seedling quality are positively correlated. For nursery cultivation, seeds should be longer than 7 mm (Aslan, 2001a).

No pretreatment is required for seeds germination but cold stratification for 1–3 months under chilling temperatures significantly increases germination (Skordilis and Thanos, 1995; Tilki and Dirik, 2007). The optimal seed germination temperature is 20°C (Sefik, 1964; Skordilis and Thanos, 1995; Yilmaz et al., 2013) and average germination percentage is about 85% after 4–6 weeks of chilling treatment with important germination differences among provenances (Skordilis and Thanos, 1995; Paitaridou et al., 2005). Under favorable conditions, seeds germinate in the nursery 2–3 weeks after sowing (Anonymous, 1986; Aslan, 2001a; Paitaridou et al., 2005).

Vegetative propagation

Somatic embryogenesis of *P. brutia* has been successfully achieved over the last decades (Panetsos et al., 1994). However, this experimental technique has not been scaled up for production.

Seedling cultivation

Container plants

There is little information on nursery cultivation of quality *P. brutia* container seedlings (Fig. 11.21). Given the ecological and functional similarity between *P. brutia* and *Pinus halepensis*, we recommend using knowledge from container cultivation of *P. halepensis* as a reference for *P. brutia*.

P. brutia has been cultivated in polybags in Türkiye. Using a 1:1 mixture of compost and silty clay loam soil in these containers resulted in optimal growth and seedlings with high quality index values (Karim et al., 2017). Fertilizing greenhouse-grown seedlings



Figure 11.21 Cultivation of 1 + 0 container (left side) and bareroot (right side) seedlings of *Pinus brutia*. *(Nebi Bilir.)*

with 200 kg/ha N, 100/ha P, and 150/ha K led to optimal growth in one-year-old *P. brutia* seedlings (Al-Naqib, 1982). *P. brutia* seedlings rapidly develop deep taproots, reaching depths of 65 cm in 5–6 months after germination (Boydak, 2004), making container depth an important consideration. For instance, 280 mL containers with a depth of 23 cm produced optimal-size seedlings (Boydak et al., 2006).

Nursery research in Türkiye often report *P. brutia* seedling sizes that exceed the standards set by the Turkish Standard Institute (Anonymous, 1988). Seedling size shows large differences depending on seed source, seed size, and nursery conditions (Dilaver et al., 2015; Karim et al., 2017). Yilmazer and Bilir (2016) reported higher growth in seedlings raised in a forest nursery at a lower elevation (elevation 120 m) than values observed at higher elevation. The observed and recommended morphological characteristics of *P. brutia* container and bareroot stock are shown in Table 11.34.

Bareroot plants

For bareroot *P. brutia* production (Fig. 11.21) the soil must be cleared of any weeds or debris and loosened to a depth of at least 25–30 cm. The ideal soil for *P. brutia* seedling growth is a well-drained substratum with a pH value between 5.8 and 7.2 (Giulimondi, 1972). Sowing in the seedbeds takes place in early spring. Since *P. brutia* germination is epigeal, seeds should not be buried to a depth more than 2 or 3 times the seed's diameter, about 1–2 cm depth, and should be covered with a mixture of soil, peat, and forest

Table 11.34 Morphological attributes of 1 + 0-year container and bareroot seedlings of *P. brutia*. The values of attributes are under controlled nursery conditions and seed provenance.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	14.2–14.8 (14.5)	≥14.0	6.3–21.9 (18.1)	≥14.0
Root collar diameter (cm)	0.31–0.46 (0.36)	≥0.35	0.18–0.49 (0.39)	≥0.35
Shoot dry weight (g)	0.04–3.88 (0.76)	–	2.1–2.8 (2.4)	–
Root dry weight (g)	0.01–1.84 (0.44)	–	0.9–1.1 (1.0)	–
Plant dry weight (g)	0.05–5.72 (1.21)	–	1.7–6.9 (2.9)	–
Shoot/root dry weight ratio	–	≥3.0	2.2–2.8 (2.4)	≥3.0

Mean values in brackets.
Anonymous (1988); Aslan (2001b); Dilaver et al. (2015); Karim et al. (2017), Yilmazer and Bilir (2016), Özel et al. (2018), Bilir (2019), Çetinkaya and Bilir (2020, 2021, 2022), Çerçioğlu and Çetinkaya (2021).

humus. Seeds should be watered immediately after sowing in March–April. Initial watering should be done regularly (avoiding flooding) and reduced to once or twice a week once the seedlings emerge (Aslan, 2001b).

Because *P. brutia* grows relatively slowly in the first year, fertilizers can accelerate growth and increase the plant's nutrient content. Two foliar applications of a nano NPK (20-20-20) fertilizer in combination with 5 g/L mineral NPK (12-12-36 applied twice in the fall and twice in spring significantly increased growth of one-year-old *P. brutia* bareroot seedlings (Shareef et al., 2021).

Dilaver et al. (2015) compared stock and found that container seedlings exceeded bareroot in size. Yilmazer and Bilir (2016), however, reported that bareroot seedlings had higher growth than container seedlings. They also found that growth depended on seed sources, nursery practices, and nursery location.

After extraction from seedbeds, 1 + 0 bareroot seedlings can be stored at 4°C by inserting the roots into wet sand at 95%–98% humidity for 40–50 days (Aslan, 2001b). Morphological values for high-quality bareroot seedlings of *P. brutia* are shown in Table 11.34.

Field establishment

There is limited quantitative information linking *P. brutia* field performance with nursery cultivation regimes, seedling quality, soil preparation methods, and other factors, which limits providing solid recommendations for best field establishment techniques.

P. brutia occurs naturally from sea level to 1500 m in Mediterranean countries, with its largest range in Türkiye (5.3 million ha, representing 23% of Türkiye's forest area) (OGM, 2022). It also grows in Greece, Cyprus, Georgia, Iran, Azerbaijan, Syria, Lebanon, Russia, and Ukraine. In addition, it is used for afforestation outside its natural range in western and central Mediterranean regions, as well as in Australia, Pakistan, and Tajikistan, due to its tolerance of dry, unirrigated areas. *P. brutia* provenances show remarkable differences in their resistance to aridity (Dirik, 2000). Although *P. brutia* is adapted to dry areas, it achieves optimum growth in regions where annual precipitation is higher than 900–1000 mm (Boydak, 2004). As a light-demanding, fast-growing, and drought resistant species, *P. brutia* is considered a key tree species for afforestation under climate change scenarios, with high resilience to climate impacts (Lebanon's Ministry of Environment/UNDP, 2011).

Reforestation success by direct seeding is generally low (less than 20% after 1–2 years) due to abiotic and biotic stressors and limited availability of high-quality seeds (Grossnickle and Ivetić, 2017). Planted trees can outperform naturally regenerated ones in height and diameter (Ertuğrul, 2024). Therefore, planting one-year-old seedlings is recommended, at a maximum density of 2200 seedlings/ha (Anonymous, 1986). For ornamental and landscape purposes, larger, older plants may be used.

In most of its natural range, the roots of *P. brutia* can grow during winter. Thus, autumn planting, during the rainiest season, is recommended to allow seedlings to have time establish before the dry summer.

P. brutia grows on many soil types, particularly on rendzina soils, soft limestones, and marls but does not tolerate poorly drained soils (Quézel, 2000; Boydak, 2004). In Türkiye, soil preparation typically involves contour tillage using heavy disc harrows, followed by row planting (Tonguç and Arslantaş, 2019).

Outplanting survival is quite variable (Sternberg et al., 2001). Weed removal significantly increases survival rate (>60%) and it can be done manually or using herbicides within a 0.5 m radius around seedlings. Survival of 1 + 0 bareroot seedlings is higher when larger seedlings are used (Dirik, 1993). After the first growing season, one-year-old seedlings show height growth between 16.7 and 17.5 cm (Sternberg et al., 2001). By the sixth year, seedlings reach 2.9 m in height with a root collar diameter of 4.45 cm (Tonguç and Arslantaş, 2019).

Thirty-two-year-old container seedlings of *P. brutia* from seed stands and seed orchards had heights of 14.07 and 14.2 m, and base diameters of 17.02 and 17.7 cm, respectively (Özbey and Bilir, 2022). In a field test with 2-0 bare-root seedlings spaced at 3 × 2 m, Ertuğrul (2024) reported that after 5 years average seedling height and base diameter were 53.7 cm (22.9–121.9 cm) and 13.8 mm (5.0–32.9 mm), respectively, while survival ranged from 51% to 100%.

Chapter References

Al-Naqib, A.I., 1982. Growth and Nutrition of *Pinus brutia* Ten. Seedlings for Silvicultural Benefits. State University of New York College of Environmental Science and Forestry.

Anonymous, 1986. Nursery practices. In: General Directorate of Turkey. Ankara.

Anonymous, 1988. Seedling Quality Classification of Coniferous. Turkish Standard Institute Press, Ankara.

Aslan, S., 2001a. Seed Characteristics of Brutian Pine. Ed. Erol Öktem, Handbook of Brutian Pine. Forest Research Institute publications, Ankara, 52/2.

Aslan, S., 2001b. Nursery technique of Brutian pine. In: Öktem, E. (Ed.), Handbook of Brutian Pine. Forest Research Institute publications, Ankara, 52/2.

Bilir, N., 2019. Seedling quality in brutian pine (*Pinus brutia* Ten.). J. Graduate School Nat. Appl. Sci. Mehmet Akif Ersoy University 10 (1), 95–101 (in Turkish).

Boydak, M., 2004. Silvicultural characteristics and natural regeneration of *Pinus brutia* Ten.-A review. Plant Ecol. 171, 153–163. https://doi.org/10.1023/B:VEGE.0000029373.54545.d2.

Boydak, M., Dirik, H., Çalıkoğlu, M., 2006. Biology and Silviculture of Brutian Pine (*Pinus brutia* Ten.). OGEM-Vak Press, Ankara.

Çerçioğlu, M., Çetinkaya, D., 2021. Comparison of seedling morphological characteristics in Brutian pine (*Pinus brutia* Ten.) provenances. Artvin Coruh Univers. J. Forest. Facul. 22 (1), 60–66 (in Turkish).

Çetinkaya, D., Bilir, N., 2020. Effect of using harvesting residues as a nursery media on seedling weight in Brutian pine (*Pinus brutia* Ten.). Reforesta 9, 37–43.

Çetinkaya, D., Bilir, N., 2021. Effect of harvesting residues on seedling morphology and quality in Brutian pine (*Pinus brutia* Ten.). Fresenius Environ. Bull. 30, 4585–4590.

Çetinkaya, D., Bilir, N., 2022. Morphological characteristics and quality in Turkish red pine (*Pinus brutia* Ten.) seedlings. In: 2nd Global Conference on Engineering Studies, 7-10 September P. 89-92 (in Turkish).

Chambel, M.R., Climent, J., Pichot, C., Ducci, F., 2013. Mediterranean pines (*Pinus halepensis* mill. And *brutia* ten.). In: Pâques, L.E. (Ed.), Forest Tree Breeding in Europe: Current State-Of-The-Art and Perspectives. Springer, Dordrecht, pp. 229–265.

Dilaver, M., Seyedi, N., Bilir, N., 2015. Seedling quality and morphology in seed sources and seedling type of Brutian pine (*Pinus brutia* Ten.). World J. Agricult. Res. 3, 83–85. https://doi.org/10.12691/wjar-3-2-9.

Dirik, H., 1993. The relationships between important seedling characteristics and planting success in Turkish red pine (*Pinus brutia* Ten.). J. Forest. Facul. Istanbul Univers. A (43), 51–75 (in Turkish).

Dirik, H., 2000. Analysis of pressure-volume (P-V) curves within dry season of Calabrian pine (*Pinus brutia* Ten.) provenances from different bioclimatic zones. J. Forest. Facul. Istanbul Univers. A (50), 100–103 (in Turkish).

Ertuğrul, G.S., 2024. Evaluation of industrial plantation applications results in terms of natural and artificial regeneration methods. Theoret. Appl. Fores. 4, 6–10. https://doi.org/10.53463/tafor.2024vol4iss1pp6-10 (in Turkish).

Ganatsas, P., Tsakaldimi, M., Zachariadis, G., 2011. Effect of air traffic pollution on seed quality characteristics of *Pinus brutia*. Environ. Exp. Bot. 74, 157–161. https://doi.org/10.1016/j.envexpbot.2011.05.014v.

Ganatsas, P., Giannakaki, M., Gouvas, A., Tsakaldimi, M., 2021. Is the reproduction capacity of *Pinus brutia* stands 20 years after wildfire efficient to secure forest restoration in the case of a fire re-occurrence? Forests 12 (8), 991. https://doi.org/10.3390/f12080991.

Gezer, A., 1986. The Sylviculture of *Pinus Brut*ia in Turkey. CIHEAM, Paris, pp. 55–66.

Giulimondi, G., 1972. Influence of soil reaction on the nursery growth of *Pinus brutia* and *P. radiata*. Agricol. Forest. Forest. Abst. 34, 414–415.

Grossnickle, S.C., Ivetić, V., 2017. Direct seeding in reforestation – a field performance. Reforesta 4, 94–142. https://doi.org/10.21750/REFOR.4.07.46.

Karim, S.A., Ameen, T.M., Ali, N.S., 2017. Interaction effect of seed size, growing media and period of sampling on growth characters of *Pinus brutia* Ten. seedling at Grdarasha/Erbil. J. Garmian Univers. 4, 419–432. ICBS Conference.

Lebanon's Ministry of Environment/UNDP, 2011. Climate Change Vulnerability and Adaptation-Forestry. https://climatechange.moe.gov.lb/viewfile.aspx?id=69.

OGM, 2022. Forest inventory of Turkey-2021. In: General Directorate of Forestry of Türkiye, Ankara, Türkiye.

Özbey, A.A., Bilir, N., 2022. Variation in growth among provenances of *Pinus brutia* Ten. in a 32-year-old experiment from Southern Turkey. J. Agric. For. 68, 83–91. https://doi.org/10.17707/AgricultForest.68.2.06.

Özel, H.B., Yücedağ, C., Bilir, N., Ölmez, Z., Aydinhan, V., 2018. The effect of seedling types on morphological characteristics in Turkish red pine (*Pinus brutia* Ten.) seedlings. Bartın University Int. J. Nat. Appl. Sci. 1 (1), 43–47 (in Turkish).

Paitaridou, D., Ganatsas, P., Sotiriou, K., Varvarigos, G., 2005. Studying seed quality of four Greek native pines. In: Proc. of 12th National Congress of Forestry Society, Drama City, 2–5 October (In Greek).

Panetsos, C.P., 1981. Monograph of *Pinus halepensis* Mill. And *P. brutia* (Ten.). Annales Forestales 9 (2), 39–77.

Panetsos, K., Scaltsoyiannes, A., Alizoti, P., 1994. Effect of genotype and cutting type on the vegetative propagation of the pine hybrid (*Pinus brutia* (Ten) × *Pinus halepensis* (Mill)). Ann. Des. Sci. For. 51, 447–454. https://doi.org/10.1051/forest:19940502.

Shareef, S.S., Qasim, H.A.J., Omar, O.M., 2021. Effect of (NPK) nano and mineral fertilizer on some growth characteristics of *Pinus brutia* Ten. seedlings by foliar application. IOP Conf. Series: Earth and Env. Sci. 910 (1), 012012. https://doi.org/10.1088/1755-1315/910/1/012012.

Skordilis, A., 1992. Laboratory Ecophysiological Study of Seed Germination and Stem Development in *Pinus halepensis* and *P. brutia* Species (Doctoral Dissertation, Dept. of Biology, Athens, Greece) (in Greek).

Skordilis, A., Thanos, C.A., 1995. Seed stratification and germination strategy in the Mediterranean pines *Pinus brutia* and *P. halepensis*. Seed Sci. Res. 5 (3), 151–160. https://doi.org/10.1017/S0960258500002774.

Spanos, I.A., Daskalakou, E.N., Thanos, C.A., 2000. Postfire, natural regeneration of *Pinus brutia* forests in Thasos island, Greece. Acta Oecol. 21 (1), 13–20. https://doi.org/10.1016/S1146-609X(00)00107-7.

Sternberg, M., Danin, A., Noy-Meir, I., 2001. Effects of clearing and herbicide treatments on coniferous seedling establishment and growth in newly planted Mediterranean forests. For. Ecol. Manag. 148 (1–3), 179–184. https://doi.org/10.1016/S0378-1127(00)00528-4.

Sefik, Y., 1964. Investigation on cone and seeds of Brutian pine (*Pinus brutia* Ten.). J. Istanbul Forest. Facul. 14 (2), 35–70.

Thanos, C.A., Marcou, S., 1991. Post-fire regeneration in *Pinus brutia* forest ecosystems of Samos island (Greece): 6 years after. Acta Oecol. 12, 633–642.

Thanos, C.A., Daskalakou, E.N., 1993. Seed characteristics of *Pinus brutia* from various locations of Samos Island. In: International Brutian Pine Symposium, P. 295–302, 18–23 October, Marmaris, Turkey.

Tilki, F., Dirik, H., 2007. Seed germination of three provenances of *Pinus brutia* (Ten.) as influenced by stratification, temperature and water stress. J. Environ. Biol. 28 (1), 133–136.

Tonguç, F., Arslantaş, M., 2019. Investigation of 6-year plantation results of Turkish red pine (*Pinus brutia* Ten.), Black pine (*Pinus nigra* Arnold subsp. *pallasiana*) and Taurus cedar (*Cedrus libani* A. Rich) species in Atatürk Forest Farm. Turk. J. Forest Sci. 3 (2), 159–169 (in Turkish).

Quézel, P., 2000. Taxonomy and biogeography of Mediterranean pines (*Pinus halepensis* and *P. brutia*). In: Ne'eman, G., Trabaud, L. (Eds.), Ecology, Biogeography and Management of *Pinus halepensis* and *P. Brutia* Forest Ecosystems in the Mediterranean Basin. Backhuys Publishers, Leiden, pp. 1–12.

Yilmaz, M., Kaplan, A., Vermez, Y., 2013. Some seed characteristics of three extreme populations of Turkish redpine (*Pinus brutia* Ten.). KSU J. Nat. Sci. 16 (1), 55–61.

Yilmazer, C., Bilir, N., 2016. Effect of seedling type in morphology and quality of Brutian pine (*Pinus brutia* Ten.) seedlings. IJRSRST 25, 237–240.

Pinus cembra L.

Peter Petrík[1,2], Daniel Kurjak[3] and Kateřina Houšková[4]
[1]Institute of Forest Botany and Forest Zoology, Technical University of Dresden (TUD), Tharandt, Germany; [2]Karlsruhe Institute of Technology (KIT), Institute of Meteorology and Climate Research—Atmospheric Environmental Research (IMK-IFU), Garmisch-Partenkirchen, Germany; [3]Faculty of Forestry, Technical University in Zvolen, Zvolen, Slovakia; [4]Faculty of Forestry and Wood Technology, Mendel University in Brno, Brno, Czech Republic

Seed propagation

Pinus cembra (Swiss stone pine or Arolla pine), is a conifer tree native to the Alps and other mountain ranges in central and eastern Europe (Gugerli et al., 2023). First cones usually appear after 40–60 years (Caudullo and de Rigo, 2016) but at lower altitudes, cone production may begin at 30–40 years (Hoffman et al., 2005). Sexual maturity is reached at 60–70 years in trees within stands, but 30–40 years in isolated trees (Mauer, 2009). *P. cembra* is a masting species and high cone production occurs every 5–10 years (Šnajperk, 1954; Zong et al., 2010; Bonner and Karrfalt, 2008; Bisi et al., 2016). Cones finish growth 2 years after pollination, producing cinnamon-brown-colored cones, and are usually harvested in October–November, or in September at lower altitudes (Vincent, 1965). Cones can be collected from standing or felled trees in natural forest, although seed orchards have also been established (Heinze and Holzer, 2013). Subsequently, cones are spread on the ground and mixed in well-ventilated areas where they dry, disintegrate, and release their seeds (Vincent, 1965; Palátová, 2008). The wingless seeds need to be cleaned to remove debris, typically using vibration sieves and gravitational separators. Large seed lots are processed with specially adapted threshers that separate seeds from cone scales and spindles and then further cleaned on separators (Vincent, 1965; Palátová, 2008). The ideal temperature for short-term storage (1–2 years) of *P. cembra* seeds or whole cones is around 2–5°C (Rohmeder and Loebel, 1940). For long-term storage (>1 year), Palátová (2008) recommends storing at −4 to −15°C with seeds maintained at 9%–12% moisture content. The average characteristics of seed lots are summarized in Table 11.35.

Seeds can be pretreated to increase germination through stratification at 3°C for 180–270 days, which corresponds to the period between seed collection and nursery sowing. Alternatively, seeds can be stratified at 20°C for 12 weeks, followed by

Table 11.35 Seed characteristics of *Pinus cembra*.

Cone to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
20–30 (35)	90	3,500–5,100 (4,400)	10–62 (44)

Mean values in brackets.
SCST (1993); Frehner and Schönenberger (1994); Hättenschwile and Körner (1995); Hoffman et al. (2005); Bonner and Karrfalt (2008); Salzer and Gugerli (2012); CABI Compendium (2019).

16–20 weeks at 3°C (Hoffman et al., 2005). Palátová (2008) and Vincent (1965) also recommended a combination of warm and cold stratification, as dormancy in *P. cembra* seeds is due to the impermeable coat and the underdeveloped embryo. Warm stratification promotes embryo growth. Hagner and Simak (1958) noted that seeds from higher altitudes, which have shorter and less developed embryos, need longer stratification time to overcome dormancy than seeds from lower altitudes. The recommended stratification growing media can be moist vermiculite, sand, or a 1:1 mixture of sand and peat. Seeds should be mixed with the medium at a 3:1 ratio (Hoffman et al., 2005).

Seedling cultivation

The growth of *P. cembra* is slow during the juvenile stages, with seedlings typically requiring 3–6 years to become suitable for forestation, either as bareroot or container stock (Šmelková, 2009).

Container plants

The average yield from 1 kg of seeds in the Czech Republic and Slovakia is approximately 1000 container plants (Šmelková, 2009; Cafourek and Kotrla, 2022). Given the low germination capacity of *P. cembra* seeds (Table 11.35), it is more cost-effective to cultivate bareroot seedlings on beds in mineral soil or in substrate in a greenhouse and then to transplant them into containers. Seedlings can be transplanted into containers after they reach a shoot height of 8–10 cm (Gugerli et al., 2023). Plastic containers are currently used for cultivating planting stock. For *P. cembra* seedlings (10–20 cm tall), containers of approximately 0.3 L are suitable. However, Heumader (1994) described pressed peat pots, which decompose within a few years and allow root penetration. Seedlings in these pots are ready for afforestation 2–3 months after transplanting, once roots pierce the walls. Transplanting from beds to pots should ideally occur while seedlings are dormant in spring, although summer transplanting is also feasible (Heumader, 1994).

P. cembra seedlings should not be cultivated in too wet substrates, as they perform better in well-drained soils. They tolerate some degree of drought and exhibit higher drought tolerance than other tree species such as *Picea abies* (Glerum and Pierpoint, 1968) and *Pinus sylvestris* (Hättenschwile and Körner, 1995). Thus, *P. cembra* has low water demand and does not require specific watering regimes. Peat is usually used, for instance mixing 70% blonde peat, 20% Lingofibre, and 10% black peat with pH = 6. It is possible to use an unfertilized substrate for transplanting and to fertilize the substrate only during the growing season (Mauer et al., 2013), but usually a substrate fertilized with a complete slow-release (4–5 month release time) complete fertilizer (NPK + micronutrients) can be used. Subsequently, it is also possible to fertilize during the growing season once a week with a foliar fertilizer (e.g., N:P:K = 14:3:21 + 2% Mg and trace

elements at 2.5 g/L/m^2). Moreover, according to Mauer et al. (2013) nitrogen fertilization should be completed in mid-July, and it is necessary to continue with phosphate-potassium fertilizers to the end of growing season (usually until October).

Bareroot plants

The average yield from 1 kg of seeds in the Czech Republic and Slovakia is approximately 700 bareroot plants (Šmelková, 2009; Cafourek and Kotrla, 2022). The optimal sowing time is immediately after harvesting in autumn. However, autumn sowing increases the risk of seed damage by rodents. Alternatively, spring sowing is carried out after germination pretreatment (Vacek et al., 2021). In the first half of May, seeds are sown in beds in rows 2–3 cm deep, in loose, humus-rich soil, using 7–8 g seeds/m of row (Šmelková, 2009; Mauer et al., 2013) with optimal density of 1200–1800 seedlings m^2 (Frehner and Schönenberger, 1994) (Fig. 11.22a).

Seedlings are usually transplanted to new beds after adjusting the root system by pruning in spring as 2-year-old plants when cultivated in mineral soil beds. Plants suitable for forest plantations are usually 4–5 years old (2 + 2 or 2 + 3), that is, 2-year-old seedlings are usually transplanted and cultivated for an additional 2–3 years in new

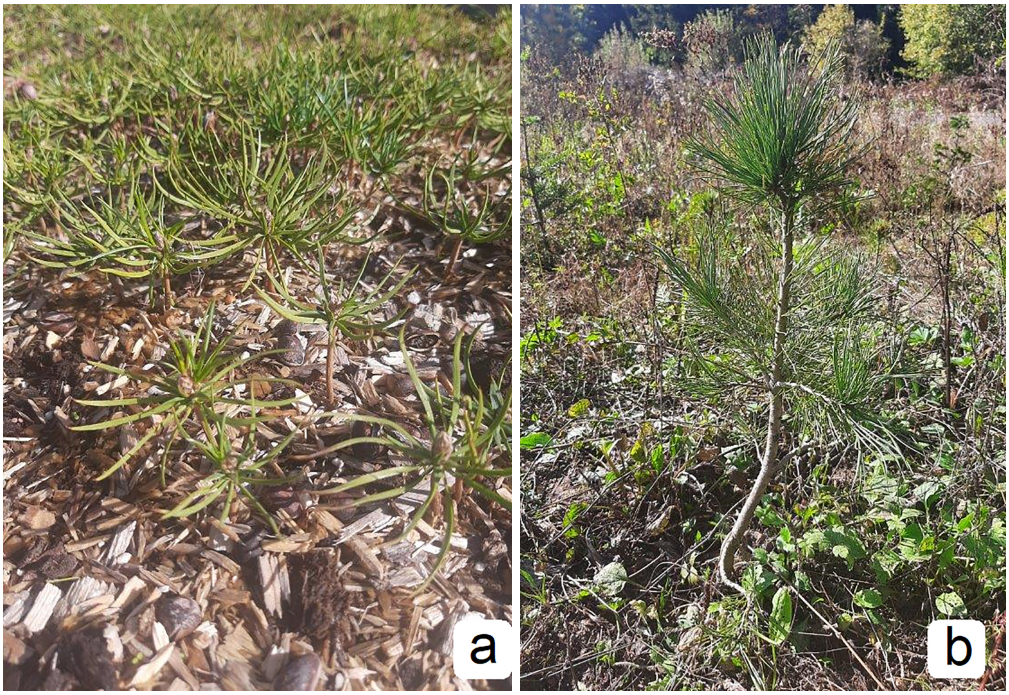


Figure 11.22 A 1-year-old *Pinus cembra* seedling in a bareroot forest nursery (a) and a 5-year-old planted sapling on a Carpathian mountain site (b). *(Peter Petrik.)*

mineral soil beds (CABI Compendium, 2019). Cultivation in peat substrates with a slightly acidic pH (ca. 6) inside greenhouses initially for a few weeks, can accelerate seedling emergence, making even 1-year-old seedlings suitable for transplanting (Mauer et al., 2013). However, to acclimate plants to outdoor conditions before winter, it is necessary to remove the foil from the greenhouse at the beginning of August at the latest. The resulting plant for forest planting would then be 3–4 years old. Spring and summer are the best seasons for transplanting *P. cembra* seedlings, although autumn transplanting is possible. However, autumn transplanting can lead to frost heaving issues, particularly in poorly rooted transplants (Heumader, 1994). To produce vigorous plants for high-elevation afforestation, Heumader (1994) recommends a special treatment of soil in the nursery and a biological cultivation method to promote mycorrhization with the following characteristics:

- **Mycorrhizal inoculation**: Seed and transplant beds should be inoculated with *Suillus placidus* and *Suillus plorans*, which yield the best results.
- **Soil enrichment**: Compost and peat should be added to improve soil conditions for mycorrhizae growth, maintaining a pH of 6.0–6.5.
- **Green manuring**: Instead of chemical fertilizers, plant beds are green-manured every fourth year with a mix of annual plants, emphasizing legumes for soil regeneration (Czell and Redlich, 1966).
- **Weed management**: Herbicides are avoided, and weeding is performed manually.

The optimal proportion of organic matter in the soil for planting stock cultivation is 5%–7% (Mauer et al., 2013), and it is advisable to supplement the soil before planting and fertilize as well. Dušek and Nárovec (1989) state the following target level of nutrients in soil: P of 35–48 mg/kg, K of 130–200 mg/kg, and Mg of 50–100 mg/kg; the specific values of the fertilizer depend on the physical properties of the soil. The heavier the soil, the higher the target nutrient levels and typically the greater the fertilizer dose. Dušek (1977) recommends applying 100–130 kg N/ha annually. Fermented organic fertilizers are most effective for promoting growth, but urea should be avoided as a nitrogen source due to sensitivity of the pine roots (Neuwinger and Schinner, 1980).

During the growing season, *P. cembra* should be fertilized frequently at low rates. Fertilization can be done weekly or biweekly until mid-July using complete fertilizers, followed by phosphate-potassium fertilizers (Mauer et al., 2013). Neuwinger and Schinner (1980) suggest a maximum standard fertilizer with an N:P:K ratio of 12:12:18 (6 g N, 6 g P_2O_5, 9 g K/m^2), plus 2% Mg and trace elements. Fertilizers containing cupric sulfate (e.g., 25 g $CuSO_4 \cdot 5H_2O/m^2$) enhance *P. cembra* seedling growth (Neuwinger and Schinner, 1980).

Field establishment

The elevational range of *P. cembra* forests is from 1500 to 2400 m a.s.l. (with isolated individuals found at 1200–2850 m) in the Alps and from 1300 to 1700 m a.s.l. (900–1985

m) in the Carpathians. It forms the treeline in these mountains, appearing as low-stature trees within stands, which transition into fragmented groups, islands, or isolated shrubs with increasing elevation (Šmelková, 2009). *P. cembra* grows on all exposures and in its mountainous habitats, it plays a vital ecological role in preventing avalanches and protecting soil, while also tolerating atmospheric pollutants. Its lower elevational boundary is determined by competition with other tree species, primarily *Picea abies*, while its upper boundary is constrained by physiological and climatic limits.

Due to its slow growth and irregular form, *P. cembra* has little economic value as a timber species, with other trees, such as larch, being preferred in alpine habitats (Farjon, 2010, Caudullo and Rigo, 2016). It has a more ecological and protection function for slopes and valleys against avalanches and soil erosion (Bussotti, 2002). Large-scale *P. cembra* afforestation between 1885 and 1952 in the Swiss Prealps were a great failure, with only 650 trees surviving out of the 450,000 planted seedlings (Fragnière et al., 2022). *P. cembra* can be established through direct sowing or by planting nursery-grown plants, usually derived from seeds. Grafted plants are used exceptionally (CABI Compendium, 2019). Container *P. cembra* generally outperform bareroot seedlings. Planting is best carried out in spring, as it results in lower mortality rates compared with autumn planting (Heumader, 1994).

P. cembra juveniles (Fig. 11.22b) are vulnerable to browsing and lethal fungal infestations caused by prolonged snow cover. Consequently, the species is absent from microhabitats or regions with extended snow cover. In addition, *P. cembra* is more susceptible to high ozone exposures than sympatric pine species but shows higher resistance to other air pollutants (Wieser et al., 2006).

Deep snow layers persisting into late winter or spring causes browning needle diseases by snow mold fungi, such as *Phacidium infestans*, *Gremmeniella abietina*, and *Herpotrichia juniperi*, resulting in seedling mortality (Senn, 1999; Li and Yang, 2004; Schneider et al., 2009). Conversely, early snowmelt in warm spring months or early snowfall in autumn reduces soil freezing, enhancing seedling survival (Vittoz et al., 2008). Increased soil warming favors cell activity, differentiation, and root growth (Gruber et al., 2009). At mountain sites, seedlings benefit from the microclimate created by dwarf shrubs, although as they grow taller, they may encounter microclimatological bottlenecks during their transition to mature trees (Grace, 2002).

Planting should be concentrated on carefully chosen microsites with 3-5 *P. cembra* seedlings per planting spot. Ideal microsites are areas around old tree stumps or on raised terrain, while cold, wet terrain depressions, with late snow disappearance are unsuitable. To mitigate the negative impacts of snow, wind, and rockfalls, artificial barriers such as permeable banks, snow fences, or other protective structures should be constructed (Mauer et al., 2013). This author also suggests the use of preparatory species such as *Betula pendula* ssp. *pendula*, *Betula pubescens* ssp. *carpatica*, *Salix aurita*, *Salix lapponum*, *Salix*

silesiaca, *Sorbus aucuparia* ssp. *glabrata*, *Alnus viridis*, and *Pinus mugo* to establish stands. These preparatory stands can later be restored with target species like *P. cembra*.

In steep, rocky terrain, the labor-intensive planting process is best conducted using the classic pickaxe-hole planting method. Mauer et al. (2013) suggests using microsites slightly above ground level, preferably near stumps and on flat sites. Mounds can be created to help plants grow quickly beyond the frost zone and reduce the negative effects of weeds. Seedlings must be protected from browsing by game (red deer) and livestock, using either mechanical barriers (tree shelters, fencing) or chemical repellants (denatonium benzoate, capsaicin).

Provenance significantly influences the growth and establishment success of *P. cembra* seedlings (Blada, 1997). For plantations near the tree line, reproductive material should come from the upper elevational range of the planting site (Budeanu et al., 2024). Seedlings grown at lower elevations should be transplanted to nurseries above 1500 m a.s.l. for the final stages of cultivation to ensure proper hardening and adaptation to alpine conditions. Because of the irregular seed production and slow growth of *P. cembra*, planning a plantation should start well in advance. Establishing seed orchards at lower elevations with high-elevation provenances can help to ensure a more consistent seed supply.

Chapter References

Bisi, F., von Hardenberg, J., Bertolino, S., Wauters, L.A., Imperio, S., Preatoni, D.G., Provenzale, A., Mazzamuto, M.V., Martinoli, A., 2016. Current and future conifer seed production in the Alps: testing weather factors as cues behind masting. Eur. J. For. Res. 135, 743–754. https://doi.org/10.1007/s10342-016-0969-4.

Blada, I., 1997. Stone pine (*Pinus cembra* L.) provenance experiment in Romania. Silvae Genet. 46, 197.

Bonner, F.T., Karrfalt, R.P. (Eds.), 2008. The woody plant seed manual, 727. Agriculture Handbook, US, pp. 637–647.

Department of Agriculture Forest Service, Washington, DC, USA

Budeanu, M., Popescu, F., Besliu, E., Apostol, E.N., 2024. Adaptability of Swiss stone pine (*Pinus cembra*) in two different environmental conditions of Romanian Carpathians. Appl. Sci. 14, 7428. https://doi.org/10.3390/app14167428.

Bussotti, F., 2002. Pinus cembra L. In: Pines of Silvicultural Importance. CAB International, pp. 50–53.

CABI, 2019. *Pinus cembra* (Arolla pine). CABI Compendium. https://doi.org/10.1079/cabicompendium.41574.

Cafourek, J., Kotrla, P., 2022. Average Yield of Planting Material of Forest Tree Species in the Czech Republic of Seedlings and Saplings (In Czech). Ministry of Agriculture of the Czech Republic, 6 pp.

Caudullo, G., de Rigo, D., 2016. *Pinus cembra* in Europe: Distribution, Habitat, Usage and Threats. European Atlas of Forest Tree Species. Publ. Off. EU, Luxembourg e01bd9b+.

Czell, A., Redlich, G.C., 1966. Die Beeinflussung des Gebrauchswertes von Junglarchen durch kombinierte. Centralblatt fur das gesamte Forstwessen Forstwesen 83, 65–84, 2 Osterr. Agrarverlag, Wien.

Dušek, V., 1977. Instructions for Forest Nurseries of State Forestry Organizations (in Czech). Ministry of Forestry and Water Management, 28 pp.

Dušek, V., Nárovec, V., 1989. Instructions for using the results of analyzes of soil samples from forest nurseries carried out by agricultural laboratories (in Czech). Forest. Game Manage. Res. Instit. 33.

Farjon, A., 2010. A Handbook of the World's Conifers. Brill, Leiden.

Fragnière, Y., Sonnenwyl, V., Clément, B., Kozlowski, G., 2022. Large-scale historical afforestation failure with *Pinus cembra* in the Swiss Prealps. N. For. 53, 533–553. https://doi.org/10.1007/s11056-021-09871-0.

Frehner, E., Schönenberger, W., 1994. Experiences with Reproduction of *Cembra pine*.

Glerum, C., Pierpoint, G., 1968. The influence of soil moisture deficits on seedling growth of three coniferous species. For. Chron. 44, 26–29. https://doi.org/10.5558/tfc44026-5.

Grace, J., 2002. Impacts of climate change on the tree line. Ann. Bot. 90, 537–544. https://doi.org/10.1093/aob/mcf222.

Gruber, A., Zimmermann, J., Wieser, G., Oberhuber, W., 2009. Effects of climate variables on intra-annual stem radial increment in *Pinus cembra* (L.) along the alpine treeline ecotone. Ann. For. Sci. 66. https://doi.org/10.1051/forest/2009038, 503–503.

Gugerli, F., Brodbeck, S., Bebi, P., Bollmann, K., Dauphin, B., Gossner, M., Krumm, F., Peter, M., Queloz, V., Reiss, G., Rellstab, C., Stofer, S., von Arx, G., Wasem, U., Zweifel, R., 2023. Swiss stone pine - portrait of a mountain forest tree, WSL: fact sheet. Swiss Federal Institute for Forest. Snow Landscape Res. WSL. 72 (32467). https://doi.org/10.55419/wsl.

Gugerli, F., Brodbeck, S., Lendvay, B., Dauphin, B., Bagnoli, F., Van Der Knaap, W.O., Tinner, W., Höhn, M., Vendramin, G.G., Morales-Molino, C., Schwörer, C., 2023. A range-wide postglacial history of Swiss stone pine based on molecular markers and palaeoecological evidence. J. Biogeogr. 50, 1049–1062. https://doi.org/10.1111/jbi.14586.

Hagner, S., Simak, M., 1958. Ein Stratifizierungsversuch mit Samen von *Pinus cembra*. [A Stratification experiment with seeds of *Pinus cembra*. Statens Skogsforskningsinstitut 227–275 (in German)].

Hättenschwiler, S., Körner, C., 1995. Responses to recent climate warming of *Pinus sylvestris* and *Pinus cembra* within their montane transition zone in the Swiss Alps. J. Veg. Sci. 6, 357–368. https://doi.org/10.2307/3236235.

Heinze, B., Holzer, K., 2013. A review of research on *Pinus cembra* in Austria, with special reference to the conservation of genetic resources. In: Greimler, C., Moser, W., Englisch, E., Zukrigl, M. (Eds.), Contributions to the 5th International Conference on the Protection of the Natural Resources in the Alpine Region. Austrian Academy of Sciences Press, Vienna, pp. 279–283.

Heumader, J., 1994. Cultivation of cembra pine plants for high-elevation regeneration. In: Proceeding: International Workshop on Subalpine Stone Pines and Their Environment: The Status of Our Knowledge. United States Department Agriculture, Forest Service, pp. 298–301.

Hoffman, J., Chválová, K., Palátová, E., 2005. Forest Seed Management in Slovakia (In Slovak). PEREX K+K, Bratislava, 195 p.

Lasota, J., Małek, S., Jasik, M., Błońska, E., 2021. Effect of planting method on C:N:P stoichiometry in soils, young silver fir (*Abies alba* Mill.) and stone pine (*Pinus cembra* L.) in the upper mountain zone of Karpaty Mountains. Ecol. Indic. 129, 107905. https://doi.org/10.1016/j.ecolind.2021.107905.

Li, M.-H., Yang, J., 2004. Effects of microsite on growth of *Pinus cembra* in the subalpine zone of the Austrian Alps. Ann. For. Sci. 61, 319–325. https://doi.org/10.1051/forest:2004025.

Mauer, O., 2009. Forests Establishment I (In Czech). Mendel University, Brno, 174 p.

Mauer, O., et al., 2013. Planting Stock Production (In Czech). Mendel University of Agriculture and Forestry, Brno, p. 206.

Neuwinger, I., Schinner, F., 1980. The influence of compound fertilizer and cupric sulfate on the growth and the bioelement content of cembra pine seedlings (*Pinus cembra*). Plant Soil 57, 257–270. https://doi.org/10.1007/BF02211686.

Palátová, E., 2008. Forest Establishment I, Forest Seed Management (In Czech). Mendel University of Agriculture and Forestry, Brno, 121 p.

Rohmeder, E., Loebel, M., 1940. Germination tests with *Pinus cembra*. Forstwissensch. Zentralb. 62, 25–36.

Salzer, K., Gugerli, F., 2012. Reduced fitness at early life stages in peripheral versus core populations of Swiss stone pine (*Pinus cembra*) is not reflected by levels of inbreeding in seed families. Alp Botany 122, 75–85. https://doi.org/10.1007/s00035-012-0106-z.

Schneider, M., Grünig, C.R., Holdenrieder, O., Sieber, T.N., 2009. Cryptic speciation and community structure of *Herpotrichia juniperi*, the causal agent of brown felt blight of conifers. Mycol. Res. 113, 887–896. https://doi.org/10.1016/j.mycres.2009.04.008.

SCST-Society of Commercial Seed Technologists, 1993. Rules for seed testing. J. Seed Technol. 16 (3), 1113. http://www.jstor.org/stable/23432704.

Senn, J., 1999. Tree mortality caused by Gremmeniella abietina in a subalpine afforestation in the central Alps and its relationship with duration of snow cover. For. Pathol. 29, 65–74. https://doi.org/10.1046/j.1439-0329.1999.00131.x.

Šmelková, Ľ., 2009. Forest Establishment (In Czech). Technical University in Zvolen, Zvolen, 338 p.

Šnajperk, R., 1954. Forest Seed Science (In Czech). Státní zemědělské nakladatelství, Praha, 332 p.

Vacek, Z., Vacek, S., Bílek, L., Baláš, M., 2021. Basics of Silviculture (In Czech). Czech University of Life Sciences, Prague, 98 pp.

Vincent, G., 1965. Forest Seed Management (In Czech). Státní zemědělské nakladatelství, Prague, 329 p.

Vittoz, P., Rulence, B., Largey, T., Freléchoux, F., 2008. Effects of climate and land-use change on the establishment and growth of Cembran Pine (*Pinus cembra* L.) over the altitudinal treeline ecotone in the central Swiss Alps. Arctic Antarct. Alpine Res. 40, 225–232. https://doi.org/10.1657/1523-0430(06-010. VITTOZ]2.0.CO;2.

Wieser, G., Manning, W.J., Tausz, M., Bytnerowicz, A., 2006. Evidence for potential impacts of ozone on *Pinus cembra* L. at mountain sites in Europe: an overview. Environ. Pollut. 139, 53–58. https://doi.org/10.1016/j.envpol.2005.04.037.

Zong, C., Wauters, L.A., Van Dongen, S., Mari, V., Romeo, C., Martinoli, A., Preatoni, D., Tosi, G., 2010. Annual variation in predation and dispersal of Arolla pine (*Pinus cembra* L.) seeds by Eurasian red squirrels and other seed-eaters. For. Ecol. Manag. 260, 587–594. https://doi.org/10.1016/j.foreco.2010.05.014.

Pinus halepensis Mill.

Jaime Puértolas[1], Juan A. Oliet[2], Marianthi Tsakaldimi[3] and Alberto Vilagrosa[4, 5]

[1]Department of Botany, Ecology, and Plant Physiology, University of La Laguna, Facultad de Farmacia, San Cristóbal de La Laguna, Canary Islands, Spain; [2]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [3]Laboratory of Silviculture, Department of Forestry and Natural Environment, Aristotle University of Thessaloniki, Thessaloniki, Greece; [4]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain; [5]Department of Ecology, University of Alicante, Alicante, Spain

Propagation

Seed propagation

Seeds of *Pinus halepensis* (Aleppo pine) can be harvested by collecting fallen cones manually or, more frequently, climbing into tree canopies. The species is serotinous, that is, cones remain on branches for several years, even after naturally opening. Therefore, an economically efficient alternative is to collect cones from felled trees during timber operations. However, harvest should be conducted in spring, between March and May to avoid humid weather or excessive heat. Since humidity can close opened cones, harvesting in very humid weather makes it impossible to distinguish between cones with and without seeds. In regions where *P. halepensis* coexists with *P. brutia* (i.e., Greece), populations of *P. halepensis* can be identified from average values of seed coat weight fraction, average seed weight, and cone length/width ratio (see Daskalou and Thanos, 2010) for discrimination values).

After harvesting, cones must be dried and opened by heating using kilns or spreading the cones over a dry rain-sheltered surface. Kiln-dried cones must be predried for a few days before introducing them into the kiln. Seeds are extracted by tumbling and shaking cones either manually or using rotary drums. Seeds are then passed through dewingers, and impurities are eliminated by air and screen separation. Clean seed weight ranges between 2% and 4% of cone weight (Alía-Miranda et al., 2009; Navarro-Cerrillo and Gálvez-Ramírez, 2001). The range of values of characteristics of seed lots are summarized in Table 11.36.

Table 11.36 Characteristics of seed lots of *Pinus halepensis*.

Number of seeds per cone	Purity (%)	No. seeds/kg	Seed mass (mg)	Seed moisture content (%)	Germination capacity %
24–70 (47)	95–100 (96)	44,000–100,000 (60,800)	12–30 (20)	7.1–9.4 (8.25)	60–100 (81)

Mean values in brackets.
Alía-Miranda et al. (2009); Catalán (1991); Daskalakou and Thanos (1996); García-Fayos (2001); Matziris (1997); Nathan et al. (1999); Navarro-Cerrillo and Gálvez-Ramírez (2001); Paitaridou et al. (2005); Thanos (2000).

Seeds must be dried below 7% moisture content for storage in airtight containers at 4°C, which guarantees seed viability for many years. Germination trials generally have found an optimal temperature between 15 and 20°C (Paitaridou et al., 2005; Skordilis and Thanos, 1995). Seed stratification before sowing is not advisable. It accelerates germination and homogenizes differences in germination rate between seed lots, but at the expense of reducing the proportion of germinated seeds. Seeds should be rehydrated in water at room temperature for 24–48 h before sowing. Germination velocity is generally slower than in other pine species from colder areas such as *P. nigra* or *P. sylvestris*. Diurnal light generally accelerates germination velocity and rate (Thanos, 2000). Seedlings have 6–10 cotyledons and short juvenile needles directly inserted on the plant main axis.

Seed germination rules provided by the International Seed Testing Association to determine germinative potential include instructions for this species prescribing germination at 20°C on top of moist paper under white light and counting germination between the seventh and the 28th day after sowing (Catalán, 1991).

No pretreatment is required for germination, but it is necessary before sowing to remove empty seeds by floating seeds in water. The tested healthy seeds can be sown directly into the container (2-3 seeds per cavity) and covered with a 3–4 mm layer of fine-grained, clean sand or perlite. Seeds germinate in the nursery from the second and third week (Tsakaldimi, 2001).

Vegetative propagation

Aleppo pine is rarely propagated vegetatively. However, good rooting rates were obtained using cuttings and in vitro propagation techniques from 1- and 2-year-old material (Zazo-Muncharaz et al., 1993). Storage of cuttings at 4°C for 4 weeks and immersion in a solution of 400 mg/L of indole 3-butyric acid, 5 mg/L of the auxin conjugate 2-(2,4-dichlorophenoxy)propanoic acid-glycine methyl ester, and 0.01% of azoxystrobin (uncoupler of respiration) improves rooting of mature cuttings (Riov et al., 2020). In addition, micropropagation techniques using whole and parts of embryos (Lambardi et al., 1993) and somatic embryogenesis (Montalbán et al., 2013) are available for this species.

Seedling cultivation

Container

Even though successful *P. halepensis* plantations using bareroot seedlings were achieved at the beginning of the 20th century, in the last 60 years seedlings have been exclusively produced in containers to enhance planting success probability on the dry, degraded sites where this species is usually planted (Zagas et al., 2004). Therefore, information on cultivation of this species will focus exclusively on container stock.

Since *P. halepensis* seedling growth is mainly determined by temperature, the cultivation schedule must be adjusted to the local temperature regime. Thus, in locations with a short vegetative period is advisable to sow inside greenhouses in winter. In warmer locations, sowing should be conducted in March–April to avoid frosts and excessive heat. Under typical Mediterranean climate, plants generally need around 30–32 weeks to reach the target size. However, in continental Mediterranean areas, growth is slower (del Campo et al., 2011a) and 2-year-old cultivation may be necessary.

Initially, *P. halepensis* seedlings were produced in polyethylene bags, but after the 1980s cultivation shifted to containers due to deformations and poor quality of the root system in bags (Peñuelas and Ocaña, 1996). Rigid or semirigid multicell containers with internal vertical ribs are used (Fig. 11.23), open at the bottom to avoid root spiraling and facilitate air pruning (Tsakaldimi, 2001). Root pruning has produced positive results from coating the interior walls of the plastic containers with various copper compounds. Seedlings raised in copper-coated plastic containers were significantly larger (greater height and diameter) than controls (Tsakaldimi and Ganatsas, 2006).

Containers of 300 mL capacity are generally large enough even in semiarid areas (Table 11.37, Fig. 11.24). High fertilization rate is a more economical way to enhance seedling size and hence quality (see below), since larger containers decrease culture density and increase production costs (Puértolas et al., 2012). Seedling density with 300 mL containers should range between 250 and 400 seedlings/m^2.

Growing media are usually coir or peat moss mixed with perlite, vermiculite, or pine bark, although mixtures with other material such as activated sewage sludge or rice hulls also render good results and reduce the use of peat (Mañas et al., 2010; Tsakaldimi,



Figure 11.23 Seedling size of *Pinus halepensis* grown in 150, 300, 600 and 1000 cm^3 containers (from left to right, size of the ruler in the middle is 50 cm). Postplanting survival and growth in semiarid and dry Mediterranean sites was optimized by growing in the 300 cm^3 container (Puértolas et al., 2012). *(Jaime Puértolas.)*

Table 11.37 Recommended ranges of morphological and physiological characteristics for 1-year-old *P. halepensis* container seedlings.

Attributes	Minimum	Maximum
Height (cm)	15	40
Root collar diameter (cm)	0.25	a
Slenderness (height/diameter; cm/mm)	5	10
Shoot biomass (g)	1.5	4
Root biomass (g)	0.8	a
Total biomass (g)	3	a
Shoot:Root ratio	1.5	2.5
Dickson quality index	0.25	a
Number of branches	4	a
N concentration (% dry weight)[b]	1.5	2.0
Root growth potential – RGP[c]	15	a

[a]No maximum limit.
[b]Maximum for seedlings to be planted in early-frost prone areas.
[c]RGP: number of new roots (>1 cm) 28 days after transplanting in optimal conditions; Fig. 11.24.
del Campo et al. (2007b); Oliet et al. (2009b); Tsakaldimi (2001); Tsakaldimi et al. (2013).



Figure 11.24 Root growth capacity (RGC) test in high fertilization (left), low fertilization but fall N loading (middle), and low fertilization (right) seedlings. High fertilization clearly enhances RGC, which is not related to the internal N concentration (very high in the middle seedling) but to seedling size (highest in the seedlings on the left). *(Pedro Villar-Salvador.)*

2006). Low quality substrates commonly used in the past based on topsoil (i.e., raw substrates), however, produce low survival and poor growth after transplanting (Vallejo et al., 2009). Electrical conductivity (0.5–4.3 dS/m) and pH (between 4 and 8) ranges indicate the wide tolerance of this species to growing medium quality parameters. However, precautions must be taken when irrigating with strongly alkaline water, which is frequent in the distribution area of *P. halepensis*. If highly alkaline water is used, acid growing media (pH between 5 and 6) is advisable. This species is also very tolerant to high substrate porosity (ranging 78%–93% in total porosity).

P. halepensis responds strongly to fertilization (Fig. 11.24), which can be applied by fertigation or slow-release fertilizers. Nutrient concentration is not usually correlated with field performance (del Campo et al., 2011b), but seedling nitrogen content (i.e., biomass x concentration) is positively correlated with postplanting performance (Puértolas et al., 2003). Optimal values of 150 mg/plant of N can be achieved by a weekly fertigation application with N-concentration between 100 and 150 mg/L (Cuesta et al., 2008) or slow-release concentration in the substrate of 7 g/L (Oliet et al., 2004). Not only is high N fertilization recommended, but also a high nitrate to ammonium ratio (Uscola et al., 2014a). The use of organic sources of N such as amino acids is as effective in growing *P. halepensis* seedlings as inorganic (ammonium nitrate) sources, but also stimulates mechanisms of cold acclimation (Sigala et al., 2021). Foliar fertilization with urea can be an effective alternative to increase plant N-content (Uscola et al., 2014b). Nitrogen deprivation during autumn hardening is not effective or even can be detrimental (Villar-Salvador et al., 2005) and does not notably increase cold hardening (Puértolas et al., 2005). On the contrary, fall N loading does not enhance root growth potential (Fig. 11.24), it significantly increases frost tolerance of both shoots and roots (Toca et al., 2018).

More studies are needed to ascertain the optimal NPK proportions for *P. halepensis*. Nitrogen and phosphorus tissue concentrations are directly related to their applied amount, but potassium is usually diluted as N and P increases (Oliet et al., 2004), so increasing K proportion over 60% of N proportion is advisable under high N fertilization. Phosphorus is also determinant in rooting capacity after transplanting (Oliet et al., 1999). Therefore, P fertilization must be maximized when substrate pH is very high or low, as P is less available in those situations.

Strong shading is not recommended during cultivation as it reduces root development even after transplanting. Even moderate shading (60%) reduces seedling root development and increases shoot: root ratio (Puértolas et al., 2009). Drought hardening enhances water avoidance acclimation mechanisms such as stomatal closure, higher root growth capacity, and accumulation of non-structural carbohydrates (Puértolas, 2002; Villar-Salvador et al., 1999) but not drought tolerance (e.g., osmotic adjustment) (Royo et al., 2001; Villar-Salvador et al., 1999). However, some of these acclimation

changes are transient (Puértolas, 2002) and there is no evidence that they improve field performance (Puértolas et al., 2024).

P. halepensis is extremely sensitive to "damping-off" caused by *Fusarium* and *Pythium* species, in particular *F. redolens, F. solani* and *P. ultimum* (Lazreg et al., 2014, 2016). Especially when sowing in spring, seeds and seedlings must be treated with a fungicide until the hypocotyl stem is lignified, alternating the use of products like hymexazol or captan. Deformities are not frequent and are usually caused by the fungus *Sirococcus congenitus*, which is transmitted via seeds, or due to micronutrient deficiencies. Therefore, is important to treat seeds carefully (Muñoz-López, 1999) and regulate growing media pH to avoid limitations to micronutrient absorption (Pisa et al., 2001). Weeds are a frequent problem in *P. halepensis* nurseries, which can be reduced by hygienic measures during seed manipulation, cleaning tools between trays, and using sterilized growing media.

Nursery application of mycorrhizal fungi species like *Pisolithus tinctorius* or *Suillus collinitus* enhance transplanting performance (Rincón et al., 2007) but studies under a semiarid climate did not observe significant effects on survival or growth (Valdecantos et al., 1996). However, high N fertilization is usually incompatible with mycorrhization success (Honrubia et al., 1997). Since both options are mutually exclusive but beneficial, nursery managers should decide which option is the most suitable after considering environmental (mycorrhization is superior) and economical (N fertilization is usually less expensive) factors. Coinoculating mycorrhizal species and rhizobacteria like *Pseudomonas fluorescens* further enhances seedling growth in the nursery (Domínguez-Núñez et al., 2013). Nevertheless, *P. halepensis* can form mycorrhizae with edible fungi, in particular *Lactarius* species, which might be commercially interesting (Díaz et al., 2009). Mycorrhization with these species can be achieved by applying 10 mL/plant of mycelial suspension and low fertilization (35 mg/plant of N). Even though *P. halepensis* does not produce ascocarps of black truffle (*Tuber melanosporum*), mycorrhization with this species is possible, also enhanced by inoculation with *Pseudomonas fluorescens*, which improves nutrient uptake and seedling quality. Thus, *P. halepensis* could function as an inocula carrier of this economically important fungus species in restored forest ecosystems (Domínguez et al., 2012).

Morphological parameters and N-concentration can determine outplanting performance (Cuesta et al., 2010; Puértolas et al., 2003; Tsakaldimi et al., 2013; Tsakaldimi and Ganatsas, 2006). Initial seedling root collar diameter classes, Dickson's quality index (DQI), and total biomass can be used as reliable predictors for field survival (Tsakaldimi et al., 2013). Root parameters, such as root collar diameter, white tip number, or root length to white tip number ratio can be positively correlated with root growth potential (Fig. 11.24) and hence seedling vigor (del Campo et al., 2007b). Seedling quality can be critical when planting on degraded sites under a warm climate (del Campo et al., 2007a). In those situations, seedlings with morphological and physiological features within the ranges shown in Table 11.37 should be planted.

The conservative water use of this species maintains good seedling quality with shoot:root ratio as high as 2.5 (Oliet et al., 2009b).

Field establishment

This species is important for reforestation in the Mediterranean region because it is an early successional species, drought tolerant, and it has a woody deep tap root with vigorous laterals. In deep soils, the diameter of the taproot reaches 15–20 cm at a depth of 1 m, while in shallow soils with low water availability, it forms an asymmetric root system (Ganatsas and Spanos, 2007).

In general, field survival of high-quality *P. halepensis* seedlings planted in manually excavated holes can be as high as 93%–100% (Ganatsas et al., 1998; Tsakaldimi, 2001). Nevertheless, due to the aridity and soil degradation that generally occurs in *P. halepensis* planting sites, some mechanical soil preparation improves soil water availability (Querejeta et al., 2001). On these planting sites, a minimum depth of 40 cm is considered to guarantee success (Serrada et al., 2021). Subsoiling is usually the best option, but the impact on established vegetation, soil removal, and alterations to surface terrain restrict its use, especially in steep slopes (García Matallana et al., 2022; Romero-Diaz et al., 2010). In marl soils with moderate stoniness and smooth slopes, digging deep (60 cm) holes by backhoe with a micro basin (Fig. 11.25) is a better option since the basin redirects surface water flow to the planting hole, minimizing soil erosion (Saquete Ferrándiz et al., 2006; Smanis et al., 2021). Plowing on abandoned agricultural sites is not recommended, as this preparation is too shallow (Bocio et al., 2004).



Figure 11.25 *Pinus halepensis* seedling planted in the middle of a microbasin in a semiarid site near Albatera (Alicante, south-Eastern Spain). The microbasin was prepared by digging a 60 cm deep hole and constructing a mound downhill with a backhoe. Seedlings are protected from herbivory by a mesh that allows most of the light to reach the seedling, as the species is sensitive to reductions in incoming light radiation. *(Jaime Puértolas.)*

Under dry and semiarid climates, seedlings should be planted in autumn as soon as the soil is wet enough (but not excessively) and never later than February. *P. halepensis* initial root growth, which is critical for early performance, is extremely fast and can occur even in winter (Corchero et al., 2002). Under wetter continental climate with cold winters, planting could be delayed somewhat but not later than April. Seedlings grown in nurseries in warm climates should not be planted on frost prone continental sites (Pardos et al., 2003). Seedling storage time at the planting site should be minimized to avoid excessive container water loss and hence lower early root growth capacity (Vallas et al., 1999). Deep planting or burying living parts of the shoot is not recommended as it reduces growth and non-structural carbohydrate concentration (Domínguez-Lerena et al., 2001).

Organic amendments and fertilization with waste products have rendered positive outplanting performance (Fuentes et al., 2007; Querejeta et al., 2001). Combining organic amendments and postplanting mycorrhization noticeably improves seedling performance and soil aggregate stability (Caravaca et al., 2002). The current high cost of these techniques can be justified by the transition to a circular economy, as it can achieve the disposal of waste products such as sewage residues (Fuentes et al., 2007) and improve carbon balance. In some cases, the effect is not short-term, but it is noticeable only a few years after application (Cortina et al., 2004). Nevertheless, these amendments should be used cautiously since *P. halepensis* is usually planted on dry sites where the risk of soil salinization is high. As a promising alternative, inoculation with rhizobacteria such as *Azospirillum brasilense* or *Pantoea dispersa* is less costly and more effectively enhances field performance (Mengual et al., 2014).

Planting density in monospecific plantations in dry regions has been around 1000 seedlings/ha (3 × 3 m spacing) (Pastor and Martín, 1989). Nowadays, however, *P. halepensis* is planted mixed with broadleaved species, and it is best planted in aggregates and not interspersed because the fast-growing *P. halepensis* rapidly dominates broadleaved seedlings (Bonet Lledós et al., 2008). When planting objectives are to create shelter to facilitate successional processes, site aridity is critical for determining plantation density. For example, in Sierra Nevada (Southern Spain), facilitation is maximized at 1000 trees/ha, while in more xeric sites the optimal density decreases to 500 trees/ha due to competition for soil water between the trees and the facilitated species (Gómez-Aparicio et al., 2009).

Common plastic tube shelters are not recommended for *P. halepensis* because they create a low light microenvironment that decreases root growth (Puértolas et al., 2010), which tends to increase seedling vulnerability to intense drought events even years after planting (Oliet et al., 2009a). This is not observed in mesh shelters that transmit most of the light radiation inside (Oliet et al., 2023; Fig. 11.25). Since strong branch growth through meshes can cause deformities and damage the plants when shelters are removed, clear plastic tube shelters could be a suitable option. Some results show that

biomass and root growth inside clear plastic tubes can be similar or even higher than non-sheltered seedlings (Flors, 2008; Puértolas et al., 2010), with a threshold of 40% light transmissivity (Vázquez de Castro et al., 2014). Nevertheless, apart from light transmissivity, tube shelter effects seem to be site specific, and factors like high dust deposition on seedlings, low winter temperatures, or high winds might explain why seedlings in clear tube shelters outperformed nonsheltered seedlings in a continental compared with a coastal site (Oliet et al., 2021). Moreover, tubes can condense water vapor when temperatures drop below the dew point, increasing soil water moisture (del Campo et al., 2006).

Despite that field irrigation has not been necessary in the past, under current climate change scenarios projecting more frequent heatwaves and prolonged drought periods, watering might be considered in the future. In general, hydrogel applications have not shown clear effects (Oliet et al., 2003), although the application of Stocksorb in a sandy soil increased *P. halepensis* survival (Hüttermann et al., 1999). Removal of competing vegetation is not generally needed (Oliet et al., 2023) except in abandoned arable lands, where plowing between planting rows twice a year is usually effective. Alternatively, mulching around the planting hole can prevent vegetation outcompeting seedlings.

Chapter References

Alía-Miranda, R., García-del Barrio, J.M., Iglesias-Sauce, S., Mancha-Núñez, J.A., De Miguel-y del Ángel, J., Nicolás-Peragón, J.L., Pérez-Martín, F., Sánchez-de Ron, D., 2009. Regiones de procedencia de especies forestales en España. Organismo Autónomo Parques Nacionales, Madrid.

Bocio, I., Navarro, F.B., Ripoll, M.A., Jiménez, M.N., De Simón, E., 2004. Holm oak (*Quercus rotundifolia* Lam.) and Aleppo pine (*Pinus halepensis* Mill.) response to different soil preparation techniques applied to forestation in abandoned farmland. Ann. For. Sci. 61, 171–178. https://doi.org/10.1051/forest:2004009.

Bonet Lledós, J.A., Arias Abril, J.J., Pemán García, J., 2008. Un ejemplo de utilización de *Pinus halepensis* y *Quercus ilex* subsp. *Ballota* con tubos invernadero en la restauración de zonas semiáridas. Cuadernos de la Sociedad Española de Ciencias Forest. 131–136.

Caravaca, F., Garcia, C., Hernández, M.T., Roldán, A., 2002. Aggregate stability changes after organic amendment and mycorrhizal inoculation in the afforestation of a semiarid site with *Pinus halepensis*. Appl. Soil Ecol. 19, 199–208. https://doi.org/10.1016/S0929-1393(01)00189-5.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: Organismo Autónomo Parques Nacionales, Madrid.

Coello, J., Ameztegui, A., Rovira, P., Fuentes, C., Piqué, M., 2018. Innovative soil conditioners and mulches for forest restoration in semiarid conditions in northeast Spain. Ecol. Eng. 118, 52–65. https://doi.org/10.1016/j.ecoleng.2018.04.015.

Corchero, S., Gozalo, M., Villar-Salvador, P., Peñuelas, J.L., 2002. Crecimiento radical en campo de *Pinus halepensis* y *Quercus ilex* plantados en diferentes momentos. Montes 68, 5–11.

Cortina, J., Bellot, J., Vilagrosa, A., Caturla, N., Maestre, F.T., Rubio, E., Ortíz de Urbina, J.M., Bonet, A., 2004. Restauración en el semiárido. In: Avances En El Estudio de La Gestión Del Monte Mediterráneo. Centro de Estudios Ambientales del Mediterráneo, pp. 345–406. Valencia.

Cuesta, B., Villar-Salvador, P., Puértolas, J., Jacobs, D.F., Benayas, J.M.R., 2010. Why do large, nitrogen rich seedlings better resist stressful transplanting conditions? A physiological analysis in two functionally contrasting Mediterranean forest species. For. Ecol. Manag. 260, 71–78. https://doi.org/10.1016/j.foreco.2010.04.002.

Cuesta, B., Villar-Salvador, P., Puértolas, J., Peñuelas, J.L., Rey-Benayas, J.M., 2008. Establecimiento de plantones de *Quercus ilex* y *Pinus halepensis* de morfología y estado nutricional contrastados en un escenario de fuerte competencia herbácea. Cuadernos de la Sociedad Española de Ciencias Forestales 55–60.

Daskalakou, E.N., Thanos, C.A., 1996. Aleppo Pine (*Pinus halepensis*) postfire regeneration: the role of canopy and soil seed banks. Int. J. Wildland Fire 6, 59–66. https://doi.org/10.1071/wf9960059.

Daskalakou, E.N., Thanos, C.A., 2010. Seed and cone morphometric indicators: A new tool for the discrimination between the common Mediterranean pines *Pinus halepensis* Mill. and *P. brutia* Ten. Plant Biosys. 144 (4), 819–825. https://doi.org/10.1080/11263504.2010.514127.

del Campo, A.D., Cerrillo, R.M.N., Hermoso, J., Ibañez, A.J., 2007a. Relationships between site and stock quality in *Pinus halepensis* Mill. reforestation on semiarid landscapes in eastern Spain. Ann. For. Sci. 64, 719–731. https://doi.org/10.1051/forest:2007052.

del Campo, A.D., Hermoso, J., Ceacero, C.J., Navarro-Cerrillo, R.M., 2011a. Nursery location and potassium enrichment in Aleppo pine stock 1. Effect on nursery culture, growth, allometry and seedling quality. Forestry 84, 221–234. https://doi.org/10.1093/forestry/cpr008.

del Campo, A.D., Hermoso, J., Flors, J., Lidón, A., Navarro-Cerrillo, R.M., 2011b. Nursery location and potassium enrichment in Aleppo pine stock 2. Performance under real and hydrogel-mediated drought conditions. Forestry 84, 235–245. https://doi.org/10.1093/forestry/cpr009.

del Campo, A.D., Navarro, R.M., Aguilella, A., González, E., 2006. Effect of tree shelter design on water condensation and run-off and its potential benefit for reforestation establishment in semiarid climates. For. Ecol. Manag. 235, 107–115. https://doi.org/10.1016/j.foreco.2006.08.003.

del Campo, A.D., Navarro-Cerrillo, R.M., Hermoso, J., Ibanez, A.J., 2007b. Relationship between root growth potential and field performance in Aleppo pine. Ann. For. Sci. 64, 541–548. https://doi.org/10.1051/forest:2007031.

Díaz, G., Carrillo, C., Honrubia, M., 2009. Production of *Pinus halepensis* seedlings inoculated with the edible fungus *Lactarius deliciosus* under nursery conditions. N. For. 38, 215–227. https://doi.org/10.1007/s11056-009-9142-y.

Domínguez, J.A., Martín, A., Anriquez, A., Albanesi, A., 2012. The combined effects of *Pseudomonas fluorescens* and *Tuber melanosporum* on the quality of *Pinus halepensis* seedlings. Mycorrhiza 22, 429–436. https://doi.org/10.1007/s00572-011-0420-0.

Domínguez-Lerena, S., Villar-Salvador, P., Fuentes, L., Peñuelas, J.L., 2001. Puede la profundidad de plantación afectar la calidad fisiológica y el desarrollo en campo de los brinzales de *Pinus halepensis*?. In: Actas Del III Congreso Forestal Español. Mesa 3. Presented at the III Congreso Forestal Español, SECF-Junta de Andalucía, Granada (Spain), pp. 49–54.

Domínguez-Núñez, J.A., Muñóz, D., De la Cruz, A., Saiz de Omeñaca, J.A., 2013. Effects of *Pseudomonas fluorescens* on the water parameters of mycorrhizal and non-mycorrhizal seedlings of *Pinus halepensis*. Agronomy 3, 571–582. https://doi.org/10.3390/agronomy3030571.

Flors, J., 2008. Estudio del efecto de distintos tratamientos culturales (tubos protectores e hidrogeles) y de fertilización potásica en el establecimiento de brinzales forestales (Undergraduate Dissertation). Universidad Politécnica de Valencia, Valencia.

Fuentes, D., Valdecantos, A., Cortina, J., Vallejo, V.R., 2007. Seedling performance in sewage sludge-amended degraded Mediterranean woodlands. Ecol. Eng. 31, 281–291. https://doi.org/10.1016/j.ecoleng.2007.09.001.

Ganatsas, P., Spanos, I., 2007. Root system asymmetry of Mediterranean pines. In: Eco-and Ground Bio-Engineering: The Use of Vegetation to Improve Slope Stability: Proceedings of the First International Conference on Eco-Engineering. Presented at the I International Conference on Eco-Engineering. Springer, Thessaloniki, pp. 127–134.

Ganatsas, P., Tsitsoni, T., Zagas, T., Hatzistathis, A., 1998. Post-fire regeneration and reclamation of mixed broadleaves-*Pinus halepensis* ecosystems, in Chalkidiki, North Greece. In: Proceedings of the VII International Congress of Ecology. Presented at the VII International Congress of Ecology, Florence.

García Matallana, R., Lucas-Borja, M.E., Gómez-Sánchez, M.E., Uddin, S.M.M., Zema, D.A., 2022. Post-fire restoration effectiveness using two soil preparation techniques and different shrubs species in pine forests of South-Eastern Spain. Ecol. Eng. 178, 106579. https://doi.org/10.1016/j.ecoleng.2022.106579.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal en la Comunidad Valenciana. Banc de Llavors Forestals. Generalitat Valenciana, Valencia.

Gómez-Aparicio, L., Zavala, M.A., Bonet, F.J., Zamora, R., 2009. Are pine plantations valid tools for restoring Mediterranean forests? An assessment along abiotic and biotic gradients. Ecol. Appl. 19 (1), 2124–2141. https://doi.org/10.1890/08-1656.

Honrubia, M., Carrillo, C., Peñuelas, J.L., Domínguez-Lerena, S., Villar-Salvador, P., Ocaña, L., 1997. Influencia de la fertirrigación en la micorrización controlada de *Pinus halepensis* en vivero. In: Actas Del I Congreso Forestal Español. Presented at the I Congreso Forestal Español, SECF-Xunta de Galicia, pp. 307–312. Pontevedra.

Hüttermann, A., Zommorodi, M., Reise, K., 1999. Addition of hydrogels to soil for prolonging the survival of *Pinus halepensis* seedlings subjected to drought. Soil Tillage Res. 50, 295–304. https://doi.org/10.1016/S0167-1987(99)00023-9.

Lambardi, M., Sharma, K.K., Thorpe, T.A., 1993. Optimization of in vitro bud induction and plantlet formation from mature embryos of Aleppo pine (*Pinus halepensis* Mill.). Vitro Cell Developm. Biol. Plant 29, 189–199. https://doi.org/10.1007/BF02632034.

Lazreg, F., Belabid, L., Sánchez, J., Gallego, E., 2016. Root rot and damping-off of Aleppo pine seedlings caused by Pythium spp. in Algerian forest nurseries. J. For. Sci. 62, 322–328. https://doi.org/10.17221/66/2014-JFS.

Lazreg, F., Belabid, L., Sanchez, J., Gallego, E., Bayaa, B., 2014. Pathogenicity of *Fusarium* spp. associated with diseases of Aleppo-pine seedlings in Algerian forest nurseries. J. For. Sci. 60, 115–120. https://doi.org/10.17221/65/2013-JFS.

Mañas, P., Castro, E., Vila, P., de las Heras, J., 2010. Use of waste materials as nursery growing media for *Pinus halepensis* production. Eur. J. For. Res. 129, 521–530. https://doi.org/10.1007/s10342-009-0349-4.

Matziris, D., 1997. Variation in growth, flowering and cone production in a clonal seed orchard of Aleppo pine grown in Greece. Silvae Genet. 46 (4), 224–228.

Mengual, C., Roldán, A., Caravaca, F., Schoebitz, M., 2014. Advantages of inoculation with immobilized rhizobacteria versus amendment with olive-mill waste in the afforestation of a semiarid area with *Pinus halepensis* Mill. Ecol. Eng. 73, 1–8. https://doi.org/10.1016/j.ecoleng.2014.09.007.

Montalbán, I.A., Setién-Olarra, A., Hargreaves, C.L., Moncaleán, P., 2013. Somatic embryogenesis in *Pinus halepensis* Mill.: an important ecological species from the Mediterranean forest. Trees (Berl.) 27, 1339–1351. https://doi.org/10.1007/s00468-013-0882-0.

Muñoz-López, C., 1999. Tipificación de los daños producidos por *Sirococcus conigenus* Canon & Minter en los brotes de *Pinus halepensis* Miller. Localización del hongo y características de sus aislamientos. Bol. Sanid. Vegetal Plagas 25, 557–571.

Nathan, R., Safriel, U.N., Noy-Meir, I., Schiller, G., 1999. Seed release without fire in *Pinus halepensis*, a Mediterranean serotinous wind-dispersed tree. J. Ecol. 87, 659–669. https://doi.org/10.1046/j.1365-2745.1999.00382.x.

Navarro-Cerrillo, R.M., Gálvez-Ramírez, C., 2001. Manual de identificación y reproducción de semillas de especies vegetales autóctonas de Andalucía. Tomo I. Consejería de Medio Ambiente. Junta de Andalucía, Sevilla.

Oliet, J., Planelles, R., Segura, M.L., Artero, F., Jacobs, D.F., 2004. Mineral nutrition and growth of containerized *Pinus halepensis* seedlings under controlled-release fertilizer. Sci. Hortic. 103, 113–129. https://doi.org/10.1016/j.scienta.2004.04.019.

Oliet, J., Segura, M.L., Dominguez, F.M., Blanco, E., Serrada, R., Arias, M.L., Artero, F., 1999. Los fertilizantes de liberación controlada lenta aplicados a la producción de planta forestal de vivero. Efecto de dosis y formulaciones sobre la calidad de *Pinus halepensis* Mill. Investigación Agraria: Sistemas y Rec. Forest. 8, 207–228.

Oliet, J.A., Artero, F., Planelles, R., López, P., Jacobs, D.F., 2009a. Efecto de la aplicación de sistemas de protección (mallas y tubos invernadero) y de mulchado individuales sobre la respuesta en plantación durante trece años de *Pinus halepensis* en el mediterráneo semiárido español. In: Presented at the XIII Congreso Forestal Mundial, FAO, Buenos Aires.

Oliet, J.A., Navarro-Cerrillo, R., Contreras, O., 2003. Evaluación de la aplicación de tubos y mejoradores en repoblaciones forestales. Junta de Andalucía, Córdoba.

Oliet, J.A., Planelles, R., Artero, F., Jacobs, D.F., 2023. Mesh-shelters provide more effective long-term protection than tube-shelters or mulching for restoration of *Pinus halepensis* in a Mediterranean arid ecosystem. Front. For. Global Change 5. https://doi.org/10.3389/ffgc.2022.1092703.

Oliet, J.A., Planelles, R., Artero, F., Valverde, R., Jacobs, D.F., Segura, M.L., 2009b. Field performance of *Pinus halepensis* planted in Mediterranean arid conditions: relative influence of seedling morphology and mineral nutrition. N. For. 37, 313–331. https://doi.org/10.1007/s11056-008-9126-3.

Oliet, J.A., Puértolas, J., Valenzuela, P., Vázquez de Castro, A., 2021. Light transmissivity of tree shelters interacts with site environment and species ecophysiology to determine outplanting performance in Mediterranean climates. Land 10, 753. https://doi.org/10.3390/land10070753.

Paitaridou, D., Ganatsas, P., Sotirou, K., Varvarigos, G., 2005. Research on seed quality of four native pine species. In: Proceedings of the 12th National Conference. Drama (Greece), pp. 2–5.

Pardos, M., Royo, A., Gil, L., Pardos, J.A., 2003. Effect of nursery location and outplanting date on field performance of *Pinus halepensis* and *Quercus ilex* seedlings. Forestry 76, 67–81. https://doi.org/10.1093/forestry/76.1.67.

Pastor, A., Martín, J., 1989. Tipificación de la calidad en repoblaciones de pino carrasco (*Pinus halepensis* Mill.) de Alicante. Implicaciones con la gestión forestal. Options Mediterranéenes. Série Séminaires 3, 313–317.

Peñuelas, J.L., Ocaña, L., 1996. In: Mundiprensa, E. (Ed.), Cultivo de plantas forestales en contenedor. Principios y fundamentos. Madrid.

Pisa, E., Siso, S., Martín-Bernal, E., Baraza, C., Gil-Pelegrín, E., 2001. Efectos de la deficiencia de micronutrientes en plántulas de *Pinus halepensis* Mill, in: Actas del III Congreso Forestal Español. Mesa 3. In: Presented at the III Congreso Forestal Español, SECF-Junta de Andalucía, Granada (Spain), pp. 107–110.

Puértolas, J., 2002. Efecto del riego y la fertilización nitrogenada sobre la calidad de planta de *Pinus halepensis* Mill. y su comportamiento en campo (Ph D Thesis). Universidad Politécnica de Madrid, Madrid.

Puértolas, J., Benito, L.F., Peñuelas, J.L., 2009. Effects of nursery shading on seedling quality and post-planting performance in two Mediterranean species with contrasting shade tolerance. N. For. 38, 295–308. https://doi.org/10.1007/s11056-009-9148-5.

Puértolas, J., Gil, L., Pardos, J.A., 2005. Effects of nitrogen fertilization and temperature on frost hardiness of Aleppo pine (*Pinus halepensis* Mill.) seedlings assessed by chlorophyll fluorescence. Forestry 78, 501–511. https://doi.org/10.1093/forestry/cpi055.

Puértolas, J., Gil, L., Pardos, J.A., 2003. Effects of nutritional status and seedling size on field performance of *Pinus halepensis* planted on former arable land in the Mediterranean basin. Forestry 76, 159–168. https://doi.org/10.1093/forestry/76.2.159.

Puértolas, J., Jacobs, D.F., Benito, L.F., Peñuelas, J.L., 2012. Cost–benefit analysis of different container capacities and fertilization regimes in *Pinus* stock-type production for forest restoration in dry Mediterranean areas. Ecol. Eng. 44, 210–215. https://doi.org/10.1016/j.ecoleng.2012.04.005.

Puértolas, J., Oliet, J.A., Jacobs, D.F., Benito, L.F., Peñuelas, J.L., 2010. Is light the key factor for success of tube shelters in forest restoration plantings under Mediterranean climates? For. Ecol. Manag. 260, 610–617. https://doi.org/10.1016/j.foreco.2010.05.017.

Puértolas, J., Villar-Salvador, P., Andivia, E., Ahuja, I., Cocozza, C., Cvjetković, B., Devetaković, J., Diez, J.J., Fløistad, I.S., Ganatsas, P., Mariotti, B., Tsakaldimi, M., Vilagrosa, A., Witzell, J., Ivetić, V., 2024. Die-hard seedlings. A global meta-analysis on the factors determining the effectiveness of drought hardening on growth and survival of forest plantations. For. Ecol. Manag. 572, 122300. https://doi.org/10.1016/j.foreco.2024.122300.

Querejeta, J.I., Roldán, A., Albaladejo, J., Castillo, V., 2001. Soil water availability improved by site preparation in a *Pinus halepensis* afforestation under semiarid climate. For. Ecol. Manag. 149, 115–128. https://doi.org/10.1016/S0378-1127(00)00549-1.

Rincón, A., de Felipe, M.R., Fernández-Pascual, M., 2007. Inoculation of *Pinus halepensis* Mill. with selected ectomycorrhizal fungi improves seedling establishment 2 years after planting in a degraded gypsum soil. Mycorrhiza 18, 23–32. https://doi.org/10.1007/s00572-007-0149-y.

Riov, J., Fox, H., Attias, R., Shklar, G., Farkash-Haim, L., Sitbon, R., Moshe, Y., Abu-Abied, M., Sadot, E., David-Schwartz, R., 2020. Improved method for vegetative propagation of mature *Pinus halepensis* and its hybrids by cuttings. Isr. J. Plant Sci. 67 (1–2), 5–15. https://doi.org/10.1163/22238980-20191118.

Romero-Diaz, A., Belmonte-Serrato, F., Ruiz-Sinoga, J.D., 2010. The geomorphic impact of afforestations on soil erosion in Southeast Spain. Land Degrad. Dev. 21, 188–195. https://doi.org/10.1002/ldr.946.

Royo, A., Gil, L., Pardos, J.A., 2001. Effect of water stress conditioning on morphology, physiology and field performance of *Pinus halepensis* Mill. seedlings. N. For. 21, 127–140. https://doi.org/10.1023/A:1011892732084.

Saquete Ferrándiz, A., Lledó Solbes, M.J., Escarré Esteve, A., Ripoll Morales, M.A., De-Simón Navarrete, E., 2006. Effects of site preparation with micro-basins on *Pinus halepensis* Mill. afforestations in a semiarid ombroclimate. Ann. For. Sci. 63, 15–22. https://doi.org/10.1051/forest:2005094.

Serrada, R., Navarro-Cerrillo, R., Gómez-Mampaso, V., Ocaña, L., Pemán, J., 2021. Procedimientos de preparación del suelo. In: Bases Técnicas y Ecológicas Del Proyecto de Repoblación Forestal. Ministerio para la Transición Ecológica y el Reto Demográfico Madrid, pp. 625–702.

Sigala, J.A., Oliet, J.A., Uscola, M., 2021. Assessing growth, frost tolerance, and acclimation of pine seedlings with contrasted dormancy strategies as influenced by organic nitrogen supply. Physiol. Plant. 173, 1105–1119. https://doi.org/10.1111/ppl.13506.

Skordilis, A., Thanos, C.A., 1995. Seed stratification and germination strategy in the Mediterranean pines *Pinus brutia* and *P. halepensis*. Seed Sci. Res. 5, 151–160. https://doi.org/10.1017/S0960258500002774.

Smanis, A., Fuentes, D., Fuente, P., Valdecantos, A., 2021. How far surface water fluxes determine restoration success in Mediterranean degraded areas? Implications for dryland precision restoration. J. Arid Environ. 187, 104445. https://doi.org/10.1016/j.jaridenv.2021.104445.

Thanos, C.A., 2000. Ecophysiology of seed germination in *Pinus halepensis* and *P. brutia*. In: Ne'eman, G., Trabaud, L. (Eds.), Ecology, Biogeography and Management of *Pinus halepensis* and *Pinus brutia* Forest Ecosystems in the Mediterranean Basin. Backhuys Publishers, Leiden, pp. 37–50.

Toca, A., Oliet, J.A., Villar-Salvador, P., Maroto, J., Jacobs, D.F., 2018. Species ecology determines the role of nitrogen nutrition in the frost tolerance of pine seedlings. Tree Physiol. 38, 96–108. https://doi.org/10.1093/treephys/tpx165.

Tsakaldimi, M., 2006. Kenaf (*Hibiscus cannabinus* L.) core and rice hulls as components of container media for growing *Pinus halepensis* M. seedlings. Bioresour. Technol. 97, 1631–1639. https://doi.org/10.1016/j.biortech.2005.07.027.

Tsakaldimi, M., Ganatsas, P., Jacobs, D.F., 2013. Prediction of planted seedling survival of five Mediterranean species based on initial seedling morphology. N. For. 44, 327–339. https://doi.org/10.1007/s11056-012-9339-3.

Tsakaldimi, M.N., 2001. Research on the Production and Quality Assessment of the Container-Planting Stock Used in the Afforestations (Ph D Thesis). Aristotle University, Greece.

Tsakaldimi, M.N., Ganatsas, P.P., 2006. Effect of chemical root pruning on stem growth, root morphology and field performance of the Mediterranean pine *Pinus halepensis* Mill. Sci. Hortic. 109, 183–189. https://doi.org/10.1016/j.scienta.2006.04.007.

Uscola, M., Oliet, J.A., Villar-Salvador, P., Díaz-Pinés, E., Jacobs, D.F., 2014a. Nitrogen form and concentration interact to affect the performance of two ecologically distinct Mediterranean forest trees. Eur. J. For. Res. 133, 235–246. https://doi.org/10.1007/s10342-013-0749-3.

Uscola, M., Villar-Salvador, P., Oliet, J., Warren, C.R., 2014b. Foliar absorption and root translocation of nitrogen from different chemical forms in seedlings of two Mediterranean trees. Environ. Exp. Bot. 104, 34–43. https://doi.org/10.1016/j.envexpbot.2014.03.004.

Valdecantos, A., Cortina, J., Seva, J.P., Bellot, J.F. de C., Vilagrosa, A.C. de E.A. del M., Vallejo, V.R.C., de, E.A. del M., 1996. Mycorrhization and application of urban compost in order to improve survival and growth of seeds of *Pinus halepensis* under semi-arid conditions. Cah. Options Mediterr. 20.

Vallas, J., Villar-Salvador, P., Peñuelas, J.L., Herrero, N., Domínguez-Lerena, S., Nicolás-Peragón, J.L., 1999. Efecto del aviveramiento prolongado sin riego en la pérdida de calidad funcional de los brinzales de *Pinus halepensis* Mill. y su comportamiento en campo. Montes 58, 51–58.

Vallejo, V.R., Serrasoles, I., Alloza, J.A., Baeza, M.J., Bladé, C., Chirino, E., Duguy, B., Fuentes, D., Pausas, J.G., Valdecantos, A., Vilagrosa, A., 2009. Long-term restoration strategies and techniques. In: Cerdá, A., Robichaud, P.R. (Eds.), Fire Effects on Soils and Restoration Strategies. Science Publishers Inc, Enfield, NH, pp. 373–398.

Vázquez de Castro, A., Oliet, J.A., Puértolas, J., Jacobs, D.F., 2014. Light transmissivity of tube shelters affects root growth and biomass allocation of *Quercus ilex* L. and *Pinus halepensis* Mill. Ann. For. Sci. 71, 91–99. https://doi.org/10.1007/s13595-013-0335-3.

Villar-Salvador, P., Ocaña, L., Penuelas, J., Carrasco, I., 1999. Effect of water stress conditioning on the water relations, root growth capacity, and the nitrogen and non-structural carbohydrate concentration of *Pinus halepensis* Mill. (Aleppo pine) seedlings. Ann. For. Sci. 56, 459–465. https://doi.org/10.1051/forest:19990602.

Villar-Salvador, P., Puértolas, J., Peñuelas, J.L., Planelles, R., 2005. Effect of nitrogen fertilization in the nursery on the drought and frost resistance of Mediterranean forest species. Forest Syst. 14, 408–418. https://doi.org/10.5424/srf/2005143-00935.

Zagas, T., Ganatsas, P., Tsitsoni, T., Tsakaldimi, M., 2004. Post-fire regeneration of *Pinus halepensis* Mill. stands in the Sithonia peninsula, northern Greece. Plant Ecol. 171, 91–99. https://doi.org/10.1023/B:vege.0000029386.12582.1e.

Zazo-Muncharaz, J., Rodríguez-Barreal, J.A., Gómez-Sanz, V., García-García, M., Saiz-de Omeñaca, J.A., 1993. Propagación vegetativa (macropropagación) de pinos españoles. In: Actas Del I Congreso Forestal Español. Presented at the I Congreso Forestal Español, SECG-Xunta de Galicia, Pontevedra.

Pinus nigra Arnold

Marianthi Tsakaldimi[1], Milan Mataruga[2] and Halil Barõş Özel[3]
[1]Laboratory of Silviculture, Department of Forestry and Natural Environment, Aristotle University of Thessaloniki, Thessaloniki, Greece; [2]Faculty of Forestry, University of Banja Luka, Republic of Srpska, Bosnia and Herzegovina; [3]Faculty of Forestry, Department of Silviculture, University of Bartõn, Bartõn, Türkiye

Propagation

Seed propagation

Pinus nigra Arnold (black pine) is a shade intolerant tree that starts cone production between 15 and 20 years when grown individually, and between 20 and 40 years in stands. The species has a masting behavior, producing large cone crops every 2–3 years (Kerr, 2000; Ganatsas, 2009). Cones ripen from October to December of the second year after flowering and can be collected before seed release in March–April of the following year (Ioannidis, 2018; Aliyar et al., 2020; Ayan et al., 2021). Approximately 1 kg of cones yields 50,000 seeds (Paitaridou et al., 2005; ÇEMGM, 2013). Tree climbers harvest cones using a ladder, small crane, or tree climbing equipment. The mean cone length is about 54 mm, and the width is about 27 mm. Heat accelerates cone dehiscence after drying at room temperature. Drying in a kiln or drying chamber starts at 30°C for 40 h or at 35–40°C for 4 h, gradually increasing temperature to 55–60°C, or alternatively, at 45 °C for 24–48 h.

As with most pine species, *P. nigra* seeds are orthodox and they can be safely dried to a 6%–7% moisture content before storage. Seed length varies between 5.9 and 6.1 mm, width between 3.3–3.4, and the mass of a single seed varies between 18 and 20 mg (Table 11.37). Seeds can be stored under cold conditions (3–4°C) in the dark (Mataruga et al., 2010; Temel et al., 2011; Alejano et al., 2019; Kaliniewicz and Tylek, 2019; Aliyar et al., 2020). Seeds germinate without any pretreatment but soaking them in water for 24 h speeds and homogenizes germination. Healthy seedlots show high germination rates when placed in a germination chamber at constant temperature of 25°C with a 12 h light/dark cycle (Turna and Bilgili, 2006) (Table 11.38). The average characteristics of seed lots are summarized in Table 11.38.

Table 11.38 Characteristics of seed lots of *Pinus nigra*.

No. of fully developed seeds per cone	Purity (%)	No. seeds/kg	Seed moisture content (%)	Germination capacity (%)
21	95–99	44,000–50,000	6.3–14.9	43–99

Kerr (2000), Paitaridou et al. (2005), Turna and Bilgili (2006); Oršanić et al. (2011); Mataruga et al. (2010); Temel et al. (2011); Alejano et al., 2019, Kaliniewicz and Tylek (2019); Šerý et al. (2020); Ayan et al. (2021).

Cone and seed size, as well as seed germination rate, are influenced by the mother tree (Mataruga, 2006), but not by tree age (Tíscar and Lucas, 2010; Alejano et al., 2019). The species does not produce serotinous cones (Isajev et al., 2009; Isajev et al., 2009), and its reproductive capacity persists into senescence, with older trees contributing as much as younger ones to forest regeneration (Tíscar, 2002).

Vegetative propagation

At present, grafting is the most common method for vegetatively propagating *P. nigra*. Although brachyblasts (needle fascicles) have been rooted, only brachyblasts from 1-year-old short shoots harvested from young (5-year-old) plants were able to form calluses or to root (Van Haverbeke, 2018). For rooting of *P. nigra* cuttings, the cuttings should be protected from direct sunlight and rooted in light shade conditions. Watering should be done when necessary. The complete rooting process takes between 1.5 and 4 months. In rooting studies using two different preparations, 34%–48% rooting was obtained (Honcharuk et al., 2022). The side graft is the usual method, but cleft and veneer grafts can also be used. Grafting is conducted on actively growing stock, and removal of the stock by pruning must be gradual after scion growth begins (Van Haverbeke, 2018). Stock-scion incompatibility in *P. nigra* is not a significant issue, especially when the stock and the scion belong to the same subspecies. *P. nigra* can be grafted onto *Pinus sylvestris*, *Pinus resinosa*, *Pinus khasya*, *Pinus montana*, *Pinus mugo*, and *Pinus contorta*; but semiincompatibility has been found with *P. ponderosa*, *P. radiata*, and *P. armandii* (Vidaković, 1974). A wide range of auxin concentrations can effectively promote the development of rooted plantlets from shoot tip explants (Kolevska-Pletikapic et al., 1983).

Seedling cultivation

Container plants

Nursery cultivation can start outdoors from March–April (Kolevska et al., 2020; Tsakaldimi et al., 2021) depending on the local climate and the elevation of the nursery, and in some cases, it may start as late as May (Ayan et al., 2021). Outplanted seedlings usually are two or 3-years old due to the species' relatively slow growth during the first year. Therefore, seedlings should be cultivated in containers with a volume of 200–370 (400) mL, and a depth of 15–18 cm to achieve suitable size and quality for planting (Biel et al., 2004; Öner and Eren, 2008; Ivetić and Škorić, 2013; Eken and Oner, 2017; Tsakaldimi et al., 2021). Plantable one-year-old stock can be produced by earlier sowing, such as in February inside a greenhouse, and with high rates of (Peñuelas Rubira et al., 2012).

A growth density of 240 seedlings/m^2 is recommended to achieve plantable size and high-quality seedlings (Tsakaldimi et al., 2021). Higher densities tend to reduce shoot

and root mass (Jinks and Mason, 1998). Seedlings can be cultivated in a mixture of peat with vermiculite or perlite in a 1:1 or 3:1 proportion (by volume) (Biel et al., 2004; Tsakaldimi et al., 2021). Some nurseries replace peat with coir.

The number of seeds per container cell ranges from 1-3, depending on the quality of the seeds and the environmental conditions during germination. After emergence, only one seedling should be left in each container cell (Lazarevic et al., 2012). During the first 3 weeks after emergence, seedlings should be treated with a fungicide to prevent damping-off (Ivetić et al., 2016).

Fertilization is of great importance in producing high-quality container seedlings with a strong potential for successful field performance (Villar-Salvador et al., 2012; Tsakaldimi et al., 2021). Two-year-old *P. nigra* container seedlings supplied during the rapid growth period with 10 g of a water-soluble fertilizer (30:10:10 NPK + micronutrients) per L of substrate (i.e., 972 mg of fertilizer per seedling weekly, distributed over 10 weeks) achieved high quality standards (Tsakaldimi et al., 2021). Slow-release fertilizers can also be mixed with unfertilized growing medium, with doses varying between 5 and 7 g/L of growing medium and per growth period (Seletković et al., 2011; Taïbi et al., 2016).

Inoculation with the ectomycorrhiza *Suillus granulatus* at a concentration of 106 spores/plant enhances nursery growth and can be reproduced at a large scale (Lazarevic et al., 2012). In contrast, inoculation with *Pisolithus tinctorius* did not show any outplanting growth benefit compared with control seedlings, either planted in sterile or in natural soil (Panagopoulos, 1995). Shading is recommended for *Pinus nigra* seedlings in the nursery, reducing irradiation by 40%–60% between May–August (Lazarevic et al., 2012; Ivetić et al., 2016; Tsakaldimi et al., 2021), especially in nurseries located in hot summer places.

Production methods play a basic role in the quality of two-year-old seedlings of *P. nigra* (Mataruga, 1998, 2003; Ivetić and Škorić, 2013; Ivetić, 2015; Eken and Oner, 2017). Observed and recommended morphological and physiological characteristics of *P. nigra* container stock are shown in Table 11.39 (Fig. 11.26).

Bareroot plants

Nursery cultivation can start between March–May, depending on the climate and elevation of the nursery (Mataruga et al., 2010; Ivetić et al., 2016; Ayan et al., 2021). For the production of 1 + 0 bareroot seedlings, some authors recommend replacing seedbed soil with a mixture of 50% peat and 50% humified bark and sawdust from beech and fir to a depth of 15 cm (Ivetić and Škorić, 2013). Others suggest a mix in the same proportion of peat, sand, and soil (Chiatante et al., 2002). As for other conifers, the recommended soil pH for *P. nigra* should range between 4.5 and 6.0.

Table 11.39 Observed morphological and physiological attributes and recommended values of high-quality 2 + 0 seedlings (container and bareroot) of *P. nigra*.

Attributes	Observed values (min-max)	
	Container seedlings	Bareroot seedlings
Shoot height (cm)	9.4–28.6	8.6–14.0
Root collar diameter (cm)	0.33–0.65	0.26–0.61
Shoot dry weight (g)	1.48–3.14	0.8–5.5
Root dry weight (g)	0.70–2.64	0.45–2.0
Plant dry weight (g)	5.0–6.0	1.73–3.9
Shoot/root dry weight	0.85–1.18	
Root area (cm^2)	45.8	
H/D (shoot height/root collar diameter)	2.0	
Root length (cm)	50.0–80.8	
Quality index	0.33–1.47	

Girard et al. (1997); Biel et al. (2004); Öner and Eren (2008); Deligöz et al. (2013); Ivetić and Škorić (2013); ÇEMGM (2013); Devetaković et al. (2017); Eken and Oner (2017); Devetaković et al. (2021); Tsakaldimi et al. (2021).



Figure 11.26 Well developed Pinus nigra container seedlings out of the plug (left) and planted in a rocky soil (left). *(Vladan Ivetić.)*

The sowing depth should be approximately 1.0–1.5 cm and seedling density in seedbeds should be kept at 300–500 seedlings/m^2 (Gezer and Yücedağ, 2013; Ivetić et al., 2016). Growing density in seedbeds greatly influences seedlings morphology. Lower density results in larger seedlings, while high growing densities can lead to a disproportionate increase in shoot to root mass ratio, which can reduce field survival (Devetaković et al., 2021). Ivetić et al. (2016) reported high survival (>95%) of one-year-old *Pinus*

nigra bareroot seedlings cultivated at a seedbed density of 500 seedlings/m^2, 3 years after field planting. Similar to container seedlings, bareroot seedlings should be treated with a fungicide during the first 3 weeks after emergence to prevent damping-off (Ivetić et al., 2016).

Root pruning is a common and essential practice in bareroot nurseries that significantly influences the root volume and physiological characteristics of saplings. This technique is particularly important in afforestation and restoration efforts in semiarid and arid environments, especially for *Pinus nigra* as it enhances establishment capacity by optimizing the use of soil moisture. According to Çetinkaya and Deligöz (2012), a twice lateral pruning regime combined with undercutting maximized root growth. However, the timing and intensity of root pruning are critical across all nursery species. The procedure has a pronounced effect on both root and total sapling biomass and must be carefully timed, as it cannot be performed throughout the year. Optimal root pruning of lateral roots should occur before bud burst, typically between the first and second growing seasons within the planting beds. During pruning, weakened, curled, damaged, or non-functional roots should be systematically removed, while care must be taken to avoid damaging or disturbing the main trunk, primary root system, and root axis (Çetinkaya and Deligöz, 2012). Pruning the roots of *P. nigra* at the beginning of the second growing season causes some stress on the seedlings, reduces height growth, shortens the central root, and promotes the development of lateral roots. This results in an improved shoot-to-root ratio and is therefore strongly recommended for the production of two-year-old *P. nigra* seedlings (Stilinović, 1987).

After bareroot seedlings are lifted from seedbeds, they should be covered with wet sheets and transported directly to the planting site. Observed and recommended morphological and physiological characteristics of *P. nigra* bareroot stock are shown in Table 11.39.

Field establishment

Despite the importance of the species in European forestry, relatively little is known about the factors driving seedling field survival, particularly in Mediterranean areas (Mataruga et al., 2012). The root growth capacity of *P. nigra* seedlings is sensitive to water stress (Girard et al., 1997). Mechanical damage to shoots and roots and the lack of hydraulic continuity between the soil and the roots after transplanting stresses seedlings, especially bareroot plants, by hindering water and nutrient uptake, stomatal conductance, and photosynthesis, which increase plant mortality (Girard et al., 1997; Grossnickle and El-Kassaby, 2016). Container plants are more suitable for planting in unfavorable environments, as their root system is protected in the plug, which also stores water and buffers against transplant shock (Jinks and Kerr, 1999; Espelta et al., 2003; Lucas -Borja et al., 2022). *P. nigra* is quite prone to transplanting shock that often results

in relatively high mortality and low growth in the first year after planting (Jinks and Kerr, 1999; Ivetić and Škorić, 2013).

Numerous studies have shown that two- or three-year-old *P. nigra* seedlings show satisfactory field survival and growth (Fig. 11.27). The minimum acceptable size for transplanting seedlings is at least 12 cm in shoot height, and 0.3 cm in root collar diameter (Jinks and Kerr, 1999; Ivetić et al., 2017; Yildiz et al., 2018; Tsakaldimi et al., 2021; Ivetić et al., 2021). A wide planting spacing is suggested, such as 3 × 1.5, 2 × 2, 3 × 3 m (Stiliniović, 1991; Öner et al., 2015; Ivetić et al., 2016; Taïbi et al., 2016; Yildiz et al., 2018).

Outplanting survival rates for *P. nigra* vary widely and are influenced by several factors, including nursery conditions, seedling quality, planting techniques, and site conditions (Öner et al., 2015; Ayan et al., 2021). For instance, the second- or third-year survival of 2 + 0 *P. nigra* seedlings in northern Serbia at elevations >700 m and using holes for soil preparation, was >90% (Ivetić et al., 2016). In Spain, survival of 1 + 0 *P. nigra* container seedlings ranged from 73%–83% on a north-oriented site but reduced to 30%–60% on a south-oriented plot (Taïbi et al., 2016).

Intensive site preparation, such as ripping to a depth of 80 cm and plowing the upper soil before planting seedlings, can enhance field establishment on dry sites. Even with strong soil preparation in arid and semiarid areas of Turkey, however, the field survival after 2 years of 1 + 0 container *P. nigra* seedlings ranged between 65% and 93% (Öner et al., 2015), while that of 2 + 0 container *P. nigra* seedlings was only 30% 5 years after outplanting (Yildiz et al., 2018).

We recommend controlling weeds mechanically or manually, starting in the first growing season and intensively for the first 5 years (Ertekin and Özel, 2010).



Figure 11.27 Planted *Pinus nigra* seedlings in mine restoration. *(Pedro Villar-Salvador.)*

Experiments simulating different water deficiency conditions showed that seedling survival is typically higher among provenances and half-sibling progeny from highly productive sites, compared with progeny from rocky locations (canyons, cliffs). Extreme values for many growth characteristics were often observed in half-sib progeny from populations growing on rocks, supporting the need for individual selection of drought-tolerant trees for seedling production (Mataruga, 2003, 2006).

Chapter References

Alejano, R., Domínguez-Delmás, M., García-González, I., Wazny, T., Vázquez-Piqué, J., Fernández-Martínez, M., 2019. The age of black pine (*Pinus nigra* Arn. ssp. *salzmannii* (Dunal) Franco) mother trees has no effect on seed germination and on offspring seedling performance. Ann. For. Sci. 76 (1), 1–10.

Aliyar, Z.B., Shafiei, A.B., Seyedi, N., Rezapour, S., Moghanjugi, S.M., 2020. Effect of traffic-induced air pollution on seed germination of Arizona Cypress (*Cupressus arizonica* green) and black pine *(Pinus nigra* Arnold). Urban For. Urban Green. 55, 126841.

Ayan, S., Yücedağ, C., Simovski, B., 2021. A major tool for afforestation of semi-arid and anthropogenic steppe areas in Turkey: *Pinus nigra* JF Arnold subsp. *pallasiana* (Lamb.) Holmboe. J. For. Sci. 67 (10), 449–463.

Biel, C., Savé, R., Habrouk, A., Espelta, J.M., Retana, J., 2004. Effects of restricted watering and CO_2 enrichment in the morphology and performance after transplanting of nursery-grown *Pinus nigra* seedlings. Hortic. Sci. 39 (3), 535–540.

ÇEMGM, 2013. Afforestation and rehabilitation guide in arid and semi-arid areas. Available at: https://www.tarimorman.gov.tr/CEM/Belgeler/erozyon%20belgeleri/Kurak%20ve%20Yar%C4%B1kurak%20Alanlarda%20A%C4%9Fa%C3%A7land%C4%B1rma%20ve%20Erozyon%20Rehberi%20son.pdf. (Accessed 10 November 2023).

Çetinkaya, D., Deligöz, A., 2012. The effects of root pruning on morphological characteristics of Anatolian black pine (*Pinus nigra* Arn. subsp. *pallasiana* (Lamb.) Holmboe) seedlings. Bartın Orman Fakültesi Dergisi 14 (Special issue), 49–58.

Chiatante, D., Di Iorio, A., Sarnataro, M., Scippa, G.S., 2002. Improving vigour assessment of pine (*Pinus nigra* Arnold) seedlings before their use in reforestation. Plant Biosyst. 136 (2), 209–216.

Deligöz, A., Bayar, E., Gür, M., Genc, M., 2013. An assessment of the important seedling properties for reforestation in *Pinus nigra* JF Arnold subsp. *nigra* var. caramanica (Loudon) Rehder from three provenances. In: International Caucasian Forestry Symposium, Vol. 24, 26).

Devetaković, J.R., Makimović, Z., Ivanović, B., Baković, Z., Ivetić, V., 2017. Stocktype effect on field performance of Austrian pine seedlings. Reforesta (4), 21–26.

Devetaković, J.R., Devetaković, J.R., Pavlović, S., Krinulović, L., Janković, I.K., 2021. Field performance of Austrian pine bareroot seedlings in comparision to seedlings pattern and density in the nursery. Reforesta 11, 27–35. https://doi.org/10.21750/REFOR.11.03.91.

Eken, O., Oner, N., 2017. An assessment of the important morphological properties of Anatolian black pine seedlings in semiarid forest nursery. Fresen. Environ. Bull. 26 (6), 4158–4162.

Ertekin, M., Özel, H.B., 2010. Black pine (*Pinus nigra* Arnold.) and Lebanon cedar (*Cedrus libani* A. Rich.) plantations for erosion control in Corum region. J. Bartin Facul. Forest. 12 (18), 77–85.

Espelta, J.M., Retana, J., Habrouk, A., 2003. An economic and ecological multi-criteria evaluation of reforestation methods to recover burned Pinus nigra forests in NE Spain. For. Ecol. Manage. 180 (1–3), 185–198. https://doi.org/10.1016/S0378-1127(02)00599-6.

Ganatsas, P., 2009. Forestry characteristics of Black Pine ecosystems and restoration of burnt stands. In: Proceedings of the International Conference on "New Approaches to the Restoration of Black Pine Forests", Sparta October 15–16, 2009, pp. 65–72 (in Greek). http://repository.biodiversity-info.gr/bitstream/11340/966/1/835.pdf.

Gezer, A., Yücedağ, C., 2013. Forest Tree Seeds and Seedling Growth Techniques, second ed. Süleyman Demirel University Publications, Isparta, p. 151 (in Turkish).

Girard, S., Clement, A., Cochard, H., Boulet-Gercourt, B., Ghuel, J.M., 1997. Effects of desiccation on post-planting stress in bare-root Corsican pine seedlings. Tree Physiol 17, 429–435.
Honcharuk, I., Matusyak, M., Pantsyreva, H., Kupchuk, I., Prokopchuk, V., Telekalo, N., 2022. Peculiarities of reproduction of "*Pinus nigra*" Arn. In Ukraine. In: Bulletin of the Transilvania University of Brasov. Series II: Forestry Wood Industry • Agricultural Food Engineering, vol 15, pp. 33–42.
Ioannidis, C., 2018. Genetic improvement of multiple traits in black pine (*Pinus nigra* Arn.). PhD thesis Dept. Forest. Nat. Environ. AUTH. https://doi.org/10.26262/heal.auth.ir.298751 (in Greek).
Isajev, V., Fady, B., Semerci, H., Andonovski, V., 2009. EUFORGEN, Guía técnica para la conservación genética y utilización del pino laricio (*Pinus nigra*).
Ivetić, V., Tsakaldimi, M., Ganatsas, P., Kerkez Janković, I., Devetaković, J., 2021. Freezing and heating tolerance of *Pinus nigra* seedlings from three south to north *Balkan provenances*. Sustainability 13 (16), 9290.
Ivetić, V., Škorić, M., 2013. The impact of seeds provenance and nursery provenance method on Austrian pine (*Pinus nigra* Arn.) seedlings quality. Ann. For. Res. 56, 297–306.
Ivetić, V., 2015. Reforestation in Serbia: success or failure? In: Ivetić, V., Stanković, D. (Eds.), Proceedings of the International Conference "Reforestation Challenges, pp. 1–12. Belgrade (Serbia) 03-06 Jun 2015. Reforesta, Belgrade, Serbia.
Ivetić, V., Grossnickle, S., Škorić, M., 2016. Forecasting the field performance of Austrian pine seedlings using morphological attributes. iFor. Biogeosci. For. 10, 99–107.
Jinks, R., Mason, B., 1998. Effects of seedling density on the growth of Corsican pine (*Pinus nigra* var. maritima Melv.), Scots pine (*Pinus sylvestris* L.) and Douglas-fir (*Pseudotsuga menziesii* Franco) in containers. Ann. de Sci. Forest. 55 (4), 407–423.
Jinks, R.L., Kerr, G., 1999. Establishment and early growth of different plant types of *Corsican pine* (*Pinus nigra* var. maritima) on four sites in Thetford Forest. Forestry 72 (4), 293–304.
Kaliniewicz, Z., Tylek, P., 2019. Aspects of the process of sorting European black pine seeds. Forests 10 (11), 966.
Kerr, G., 2000. Natural regeneration of Corsican pine (*Pinus nigra* subsp. *laricio*) in Great Britain. Forestry 73 (5), 479–488.
Kolevska, D.D., Dimitrova, A., Cokoski, K., Basova, M., 2020. Growth and quality of *Pinus nigra* (Arn.), *Pinus sylvestris* (L.) and *Pinus pinaster* (Aiton) seedlings in two container types. Reforesta (9), 21–36.
Kolevska-Pletikapic, B., Jelaska, S., Berljak, J., Vidakovic, M., 1983. Bud and shoot formation in juvenile tissue culture of *Pinus nigra*. Silvae Genet. 32, 115–119.
Lazarevic, J.S., Keca, N.D., Martinovic, A.B., 2012. Mycorrhization of containerized *Pinus nigra* seedlings with *Suillus granulatus* under open field conditions. Forest Syst. 21 (3), 498–507.
Lucas-Borja, M.E., Jing, X., Candel-Perez, D., Parhizkar, M., Rocha, F., Heydari, M., Muñoz-Rojas, M., Zema, D.A., 2022. Afforestation with *Pinus nigra* Arn. ssp *salzmannii* along an elevation gradient: controlling factors and implications for climate change adaptation. Trees 36, 93–102.
Mataruga, M., 1998. Interdependence of Characteristics and Development of Austrian Pine (*Pinus nigra* Arn.) Seedlings in Seed Orchard on Jelova Gora. Master thesis. Faculty of Forestry, University of Belgrade (in Serbian).
Mataruga, M., 2003. Genetic-selection Bases of Enhancing the Seedling Production of Different Provenances of Austrian Pine (*Pinus nigra* Arnold). PhD thesis. Faculty of Forestry, University of Belgrade (in Serbian).
Mataruga, M., Haase, D.L., Isajev, V., 2010. Dynamics of seed imbibition and germination of Austrian pine (*Pinus nigra* Arnold) from extreme habitat conditions within five Balkan provenances. N. For. 40, 229–242.
Mataruga, M., 2006. Black Pine on the Rocks Variability and Possibility of Use. Faculty of Forestry, University of Banja Luka, pp. 1–282 (in Serbian).
Mataruga, M., Haase, D., Isajev, V., Orlovic, S., 2012. Growth, survival, and genetic variability of Austrian pine (*Pinus nigra* Arnold) seedlings in response to water deficit. N. For. 43, 791–804.
Oršanić, M., Drvodelić, D., Matić, S., 2011. Seed production of some more important tree and shrub species in the Croatian Mediterranean region (in Croatian). In: Slavko Matić: Šume hrvatskoga Sredozemlja, Akademija šumarskih znanosti, pp. 427–464 (in Croatian).

Öner, N., Eren, F., 2008. The comparisons between root collar diameter and height growth of black pine (*Pinus nigra* Arnold.) and Scots pine (*Pinus sylvestris* L.) seedlings in Bolu Forest Nursery. J. Appl. Biol. Sci. 2 (1), 7–12.

Öner, N., Kondur, Y., Simşek, Z.İ.Y.A., Aslan, S., 2015. Evaluation of survival ratios and growth of the common plantation species (*Black pine* and *Taurus cedar*) on arid and semiarid sites in Turkey. Fresenius Environ. Bull. 24 (9a), 2906–2915.

Paitaridou, D., Ganatsas, P., Sotiriou, K., Varvarigos, G., 2005. Control of the seed quality of four native pine species. In: Proceedings of the 12th Panhellenic Forestry Conference, Drama, October 2–5, 2005, pp. 151–160 (in Greek).

Panagopoulos, T., 1995. Assessment of the afforestation potential in the lignite mines of Ptolemaida: contributions to environment and landscape improvement. PhD thesis. In: School of Forestry and Natural Environment, Aristotle University of Thessaloniki (In Greek). https://doi.org/10.12681/eadd/8975.

Peñuelas Rubira, J.L., Nicolás Peragón, J.L., Benito Matías, F., 2012. *Pinus nigra* subsp. *salzmannii*. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo Parques Nacionales. Serie Forestal. Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, Spain, pp. 881–899.

Seletković, I., Potočić, N., Topić, V., Butorac, L., Jelić, G., Jazbec, A., 2011. Influence of various container types and slow-release fertilizer doses on growth and physiological parameters of black Pine (*Pinus nigra* Arn.) seedlings. Šumarski List 135 (13), 90–101 (in Croatian).

Stilinović, S., 1987. Forest and Ornamental Seedlings Production. Faculty of Forestry, University of Belgrade – Textbook, pp. 1–454 (in Serbian).

Stiliniović, S., 1991. Afforestation. Faculty of Forestry, University of Belgrade – Textbook, p. 272 pp. (in Serbian).

Šerý, M., Zahoranova, A., Kerdik, A., Šerá, B., 2020. Seed germination of black pine (*Pinus nigra* Arnold) after diffuse coplanar surface barrier discharge plasma treatment. IEEE Trans. Plasma Sci. 48 (4), 939–945.

Taïbi, K., Del Campo, A.D., Aguado, A., Mulet, J.M., 2016. Early establishment response of different *Pinus nigra* ssp. *salzmanii* seed sources on contrasting environments: implications for future reforestation programs and assisted population migration. J. Environ. Manag. 171, 184–194.

Temel, F., Gülcü, S., Ölmez, Z., Göktürk, A., 2011. Germination of Anatolian black pine (*Pinus nigra* subsp. *pallasiana*) seeds from the lakes region of Turkey: geographic variation and effect of storage. Not. Bot. Horti Agrobot. Cluj-Napoca 39 (1), 267–274.

Tíscar, P.A., 2002. Capacidad reproductiva de *Pinus nigra* subsp. *salzmannii* en relación con la edadde la planta madre. Investigación Agraria: Sistemas y Recursos Forest. 2, 357–332.

Tíscar, P., Lucas, M., 2010. Seed mass variation, germination time and seedling performance in a population of *Pinus nigra* subsp. *salzamannii*. Forest Syst. 19 (3), 344–353.

Tsakaldimi, M., Giannaki, P., Ivetić, V., Kapsali, N., Ganatsas, P., 2021. Fertilization and shading trials to promote *Pinus nigra* seedlings' nursery growth under the climate change demands. Sustainability 13 (6), 3563.

Turna, I., Bilgili, E., 2006. Effect of heat on seed germination of *Pinus sylvestris* and *Pinus nigra* ssp. pallasiana. Int. J. Wildland Fire 15 (2), 283–286.

Villar-Salvador, P., Puertolas, J., Cuesta, B., Penuelas, J.L., Uscola, M., Heredia-Guerrero, N., Rey-Benayas, J.M., 2012. Increase in size and nitrogen concentration enhances seedling survival in Mediterranean plantations. Insights from an ecophysiological conceptual model of plant survival. N. For. 43, 755–770.

Van Haverbeke, D., 2018. Twenty-Year Performances of Scotch, European Black (Austrian), Red, and Jack Pines in Eastern Nebraska (Classic Reprint), pp. 1–22.

Vidaković, M., 1974. Genetics of European black pine (*Pinus nigra* Am.). Annales Forest. 6, 57–86.

Yildiz, O., Altundağ, E., Çetin, B., Teoman, Güner, Sarginci, M., Toprak, B., 2018. Experimental arid land afforestation in Central Anatolia, Turkey. Env. Monit. Assess. 190 (6), 1–17. https://doi.org/10.1007/S10661-018-6724-1/TABLES/5.

Pinus pinaster Aiton

Juan Majada[1], Juan A. Oliet Palá[2], María José Aroca-Fernández[2], Juan Ignacio García-Viñas[2], Armando Herrero[3], Antonio T. Monteiro[4] and Ana E. Barros[5]

[1]Fundación Cetemas, Asturias, Spain; [2]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [3]Vivero Central de Castilla y León, Junta de Castilla y León, Valladolid, Spain; [4]Institute of Geography and Spatial Planning, Associate Laboratory Terra, University of Lisbon, Lisbon, Portugal; [5]Viveiros Aliança, Herdade de Espirra, Portugal

Propagation

Seed propagation

Pinus pinaster (maritime pine) shows high genetic diversity (Climent et al., 2021), likely resulting from evolutionary events occurring before and throughout the Last Glacial Maximum and the Holocene. During this time, the distribution of the species was reduced to isolated populations across a wide variety of habitats from which it latter recolonized (Jaramillo-Correa et al., 2020). This genetic diversification has enabled the species to tolerate a wide range of environmental conditions, indicating significant evolutionary adaptation (Ruiz de la Torre, 2006). Diversification also affects seed production and germination patterns, although general trends can be described.

Cone length varies from 8 to 22 cm (Alía et al., 1996), with a pyramidal process, a rhomboid profile, and a navel (Ruiz de la Torre, 2006). Cones take 2 years to develop and ripen between early November and the end of March or later in colder locations. The best time for harvesting begins when the cones turn uniformly brown. Premature harvesting negatively influences seed germination potential (Catalán, 1977).

Cone production has three relevant characteristics:

(i) Masting frequency: this alternates between (0) 1 and 3 (4) years, depending on the origin. In Spain, for instance, masting occurs every 3 years in Sierra de Gredos (Central Spain) but almost annually in the Sierras Béticas (Andalusia).

(ii) Intraspecific variability in cone size and production: 1 hl can contain 30–38 kg of cones (Alía et al., 2009), with 40–70 (110) seeds per cone. Highly productive trees yield between 8 and 30 kg cones per tree corresponding to 15 and 55 kg seeds/ha depending on stand structure (Lafuente et al., 2012).

(iii) Serotiny: *P. pinaster* produces serotinous cones, with production varying among provenances. Populations in Andalusia, Morocco, Algeria, and Tunisia show high levels of serotiny, while populations in the Duero basin (Spain) and Portugal have almost none (Tapias and Gil, 2000). Seeds from serotinous cones have a germination potential of approximately 70%, although they exhibit increased dormancy (Molina, 1994).

Cone harvesting can be done manually with a pole -with or without climbing-, or with a self-propelled telescopic boom lift. The collected cones must be dried to complete their maturation, open scales, and release the seeds. When cones are harvested

wet, they need additional drying before ripening begins. Dehiscence is currently carried out in kilns or drying chambers under controlled conditions of temperature and humidity. In the past, it was common to dry the cones by spreading them out on smooth, well-aerated and covered surfaces, but this system was gradually abandoned because of irregular and unsatisfactory results in preserving seed viability. Temperature must be raised a little higher for a limited period when serotinous cones are to be treated. Cones should be stirred at regular intervals to promote even drying. Throughout this process, conditions must be monitored to avoid favoring fungal infections. After dehiscence, seeds must be dewinged either by hand or mechanically.

P. pinaster seeds are ovoid, laterally compressed, and bicolor: blackish on one side and gray on the other (Fig. 11.28). Size ranges from 4.5 to 11.8 mm long, 3.8–6.2 mm wide and 2.6–4.3 mm thick. They are orthodox (Catalán, 1977; Lafuente et al., 2012) and should be kept in airtight containers preferably between 2–10°C and a relative humidity between 7% and 10%. Cryopreservation does not significantly reduce viability (Pita et al., 1998), but thermal shocks delay germination (Álvarez et al., 2007). The average seed characteristics are summarized in Table 11.40.

A testa imposes seed dormancy; without pretreatment, germination tends to be slow and irregular. The most common procedure for breaking dormancy is cold stratification of seeds at 6–7°C for 20–45 days, which significantly improves germination homogeneity (Lafuente et al., 2012). Time of stratification affects germination speed in a complex way that differs between populations and even within the same population (Wahid et al., 2007). In the past, submerging seeds in water at 16–20°C for 10 days was commonly used to break dormancy.

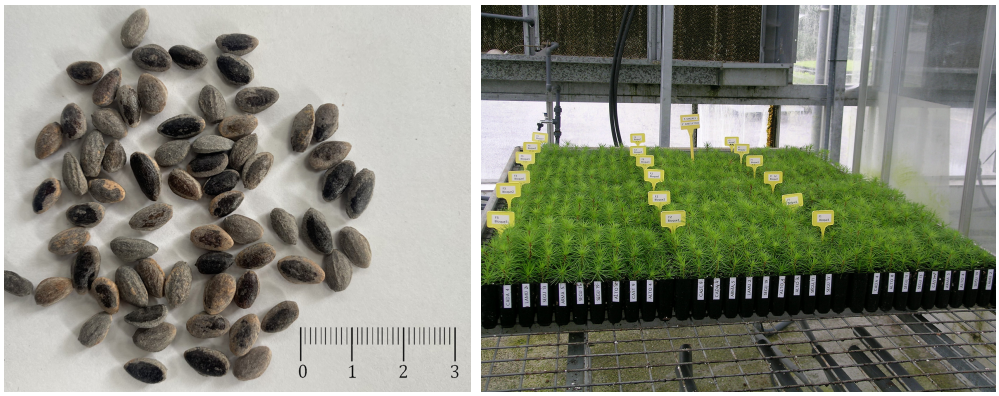


Figure 11.28 Wingless seeds (left) and plantlets from rooted minicuttings (right) of *Pinus pinaster*. *(Juan I. García-Viñas and Juan Majada, respectively.)*

Table 11.40 Characteristics of seed lots of *Pinus pinaster*.

Purity (%)	No. seeds/kg	Germination capacity (%)
95–99 (97)	13,000–28,300 (20,650)	65–90 (77)

Mean values in brackets.
Catalán (1977); Herrero and Nicolás (2012); Vivero Central de Castilla y León, Valladolid, Spain (2023, pers comm); Correia et al. (2014); Maia and Corticeiro (2022); Moraldi (2024).

Vegetative propagation

Vegetative propagation of *P. pinaster* has been developed on pilot and operational scales. It is based on small cuttings from axillary buds and brachyblasts. Moreover, grafting of resin-producing trees has also been carried out. Currently, rooted macro-, mini-, or microstem cuttings can only be used to propagate juvenile maritime pine material, typically seedlings. A high clonal heritability for shoot ontogenetic index and a moderately high heritability for rooting ability has been reported (Climent et al., 2013). These results indicate that both developmental phenomena are genetically controlled, but not strictly associated in this species.

The possibility for direct vegetative propagation of seed embryos (through somatic embryogenesis) and seedlings (through macro, mini and microcuttings) have a lot of interest for bulk propagation of best families in conifers (Rosvall et al., 2021) including *P. pinaster* (Majada et al., 2011; Martínez-Alonso et al., 2012). In addition, multivarietal forestry (Park et al., 2016), defined as the deployment of tested clones in plantation forestry, would be applicable by combining somatic embryogenesis with cryopreservation (Trontin et al., 2019).

Minicuttings 3–5 cm long and treated with IBA, placed at 25°C can achieve rooting rates between 68% and 97% (Majada et al., 2011). Recent studies show that the use of juvenile material, suitable environmental conditions, and IBA application benefits the rooting of clonal material and results in high rooting capacity (Martínez-Alonso et al., 2012). The propagation protocol developed in this study proved to be very effective and easy to implement on a large scale. Compared with the traditional rooting procedure by stem-cuttings, minicuttings have many advantages, leading to operational, technical, economic, environmental, and quality benefits. Overall, the minicutting technique for *P. pinaster* has great potential in mass scale cloning, providing high quality sprout production and well-formed new plants (Fig. 11.28). Labor and cost demands are markedly reduced, due to the elimination of labor-intensive treatment with growth substances; many field operations such as soil preparation, fertilization, cultivation, pest, and disease control are replaced by intensive activities in smaller indoor areas at much lower costs (Martínez-Alonso et al., 2012). In terms of cuttings produced per mother plant and rooting results, spring was the most favorable season for producing mini-cuttings.

Álvarez et al. (2009) developed an improved micropropagation protocol for *P. pinaster* using biotechnological methods with isolated cotyledons. Based on 86% rooting success, at least 70 plantlets can be expected per seed after 16 weeks. Somatic embryogenesis is the key technology for mass propagation of *P. pinaster*. As trees reach their adult vegetative or reproductive phase, the species is increasingly recalcitrant to vegetative propagation through conventional cuttings. Nevertheless, somatic embryogenesis initiation from mature conifers trees is still challenging. Therefore, the approach developed for *P. pinaster* is postponed propagation of tested trees by combining somatic embryogenesis initiation from immature zygotic embryos and stable cryopreservation of juvenile embryogenic tissue. Recent achievements and challenges toward efficient somatic embryogenesis are a key technology for *P. pinaster* multivarietal forestry (Trontin et al., 2016). Multivarietal forestry optimizes genetic gain at a selected level of genetic diversity in plantation forestry by combining the best varieties (for specific traits) in clonal mixtures. An important collection of over 2000 cryopreserved embryogenic lines from diverse elite families of the French breeding programs has been established by L'Institut Technologique Foret Cellulose Bois-Construction Ameublement (Trontin et al., 2016).

Seedling cultivation

Pinus pinaster can be cultivated as bareroot and container seedlings. In northern Spain, bareroot plants were traditionally used mainly due to tree stability issues observed in some plantations established with container stock. Instability was caused by four factors: (i) containers with inappropriate volume or design, (ii) excessively prolonged cultivation periods, (iii) inadequate soil preparation and planting, or (iv) soil or site limitations (Ocaña et al., 2001). Nursery practices and the quality of container seedlings have significantly improved, however, and container stock increasingly is used throughout the species distribution, particularly in Atlantic, continental and Mediterranean climates (Álvarez, 2004). Containers have important advantages including better control of cultivation variables, an extended planting period, and improved rooting and establishment after planting. Furthermore, container stock is easier to manage in the nursery during lifting, planting is faster, less labor demanding, and can be carried out by less specialized personnel. Therefore, we will focus only on container seedling production.

Container plants

Pinus pinaster is undemanding of environmental conditions for container cultivation. At sowing and during germination, however, extreme temperatures must be avoided by choosing an adequate sowing date and/or placing the plants in a greenhouse. *Pinus pinaster* can be cultivated in one season, around 30 weeks in total. Container-grown plants can be transferred to the field, especially in Atlantic zones, from just 16 weeks

as long as an appropriate fertigation is scheduled. At early stages, it is advisable to avoid high temperatures, excessive watering, and/or nitrogen application as they may increase the vulnerability of young plantlets to damping-off. It is important to synchronize seed sowing dates, management conditions, and planting-on dates (Navarro et al., 2006). Hardening-off of plants before delivery must always be considered. The best hardening procedures are restricting watering to stress the plants or exposing them to low temperatures. Importantly, container plants are more sensitive to frosts in the nursery than bare-root plants because roots are more exposed (Toca et al., 2018). When frosts are hard in winter, root systems need to be protected to protect the plug from freezing.

Container size is of particular importance in *P. pinaster* due to its fast growth and tendency to develop root deformations (Marcelli, 1989). Container specifications depend on the ecological conditions of planting sites. European regulations set a minimum container size of 120 mL (European Council, 2000), commonly used in Portuguese nurseries. In Atlantic environments, however, containers with an intermediate volume (200 mL) were associated with fewer stability issues (Ocaña Bueno et al., 2004). In Mediterranean climate, a volume of 300 mL is required because small containers have issues associated with limited growth, needle chlorosis, loss of dominance, and weak root systems.

Seedling quality requirements vary depending on planting conditions. Due to concerns in Atlantic environments about tree stability, containers of different sizes and shapes, varying cultivation periods, and root pruning methods (chemical or aerial) have been studied (Cabal et al., 2005; Krasowski, 2003; Lario et al., 2005). Stem straightness is most influenced by top dimensions, height, and volume of the container (Lario et al., 2005). Containers with perforated lateral walls to ensure air root pruning eliminates root spiraling. Conversely, shorter cultivation periods (4–6 months) allow a reduction in containers size (as little as 110 mL), with densities up to 500 plants/m^2. For longer cultivation periods, density needs to be reduced to 300–400 plants/m^2, with container volumes increased to 200–300 mL (Peñuelas and Ocaña, 1996). In Mediterranean zones, failures are mainly due to summer drought or late frosts. In contrast to Atlantic sites, stability problems are scarce as plants grow slower. The key to success on Mediterranean sites is to cultivate large, nutrient-rich, hardened-off plants with rapid root growth during establishment that ensures a deep root system capable of accessing moist soil horizons in summer (Toca et al., 2019).

Numerous growing media have been tested for *P. pinaster* (Rigueiro et al., 2001; Lazcano et al., 2010), with the best being those based on organic components such as moss (*Sphagnum*) peat, and coconut fiber. Generally, this is mixed with vermiculite or perlite to address aeration deficiencies of the organic materials and enriched with solid fertilizer. *P. pinaster* is sensitive to growing media compaction and low-aeration mixes should not be used. Pine bark was common 2 decades ago, but it is now little used, mainly due to diseases, particularly *Fusarium circinatum*. Growing media made from

composted urban waste or vermicompost have been evaluated with good results if mixed with peat (Mañas et al., 2009). Optimal pH should range between 4 and 5.5 to prevent the proliferation of fungal pathogens and ensure appropriate plant nutrition. In addition, porosity needs to be high with enough macropores to allow for root aeration, as well as micropores to ensure adequate water availability.

Fertilization is gaining importance to produce well-nourished seedlings of an appropriate size (Fraysse and Canlet 2017). Plant nutrient demand depends on the origin of the forest reproductive material, with improved material having higher nutrient demand (Ocaña Bueno et al., 1997). Nurseries can use different fertilization strategies such fertigation or incorporating solid fertilizer into the growing medium. Fertigation is more flexible, allowing nutrient supply to match seedling nutrient demand. Fertigation can be applied at different frequencies (every irrigation event, weekly, or biweekly) and at a constant (same rate applied at each fertigation event) or exponential rate (starting low and increasing as the plant grows) that produces seedlings with high tissue nutrient concentration. Exponential fertilization (Ingestad and Lund, 1986) is gaining popularity in forestry nurseries because it increases production control (Timmer, 1996) and is especially beneficial if planting site conditions are harsh, as nutrient-loaded seedlings are more competitive and drought resistant (Oliet et al., 2013). Exponential fertilization results in moderate shoot growth while increasing tissue nutrient concentration that can be remobilized for root and new stem growth of planted seedlings (Timmer, 1996). Exponential fertilization is being used for the production of *P. pinaster* in Atlantic regions, where the nutrition regime and the length of the cultivation are adjusted from sowing to lifting (Fraysse and Crémière, 1998).

Seedling nutrient loading can also be achieved by fall fertilization, when cold reduces growth but not nutrient uptake. Fall fertilization has been shown to significantly increase needle and root N concentrations without compromising frost tolerance in *P. pinaster* (Toca et al., 2018). N-rich seedlings have higher outplanting growth and survival (Oliet et al., 2013; Puértolas et al., 2012; Villar-Salvador et al., 2012). In particular, *P. pinaster* shows greater plasticity to N availability compared with other Mediterranean pines, with strong root growth response to higher N rates (150 mg) compared with lower rates (20 mg) after planting (Toca et al., 2019).

Pinus pinaster seedlings must be cultivated under full sun as it increases outplanting survival in Mediterranean conditions compared with seedlings grown under shade (Al Pavel et al., 2024). The use of mycorrhized plants may play an important role in the reforestation of degraded sites, such as those affected by forest fires, where natural regeneration is low or absent. Inoculation with ectomycorrhizal fungi of the genera *Lactarius* and *Suillus (Lactarius deliciosus, L. sanguifluus, Suillus collinitus, S. mediterranensis*) increased survival in semiarid areas of Spain (de las Heras et al., 2002; González-Ochoa et al., 2003). Bareroot and container plants inoculated in the nursery with several species of fungi (*Laccaria bicolor, L. laccata, Melanogaster ambiguus, Pisolithus tinctorius, Rhizopogon*

colossus, R. luteolus, R. roseolus, R. subareolatus, or *Scleroderma verrucosum*) across different land uses had improved growth of transplanted plants (Pera and Parladé, 2005). Similarly, under limiting soil conditions, growth of mycorrhized plants was more than double than that of non-inoculated plants (Alonso-Graña, 2010; Oliveira et al., 2012; Sanchez-Zabala et al., 2013).

Optimal N concentration in needles of *P. pinaster* should be above 12 mg/g and outplanting performance improved up to about 20 mg/g in Mediterranean regions and up to 40 mg/g in regions with an Atlantic climate where there is no drought or extreme cold (Lafuente et al., 2012). Some countries of southern Europe have seedling size specifications for Mediterranean areas (European Council, 2000). Apart from what is contained in the regulations, Rodríguez Soalleiro et al. (1997) proposed other morphological standards for this species (Table 11.41).

Field establishment

Planting has traditionally been preferred over direct seeding for establishing *P. pinaster*, despite the strong ability of this species to regenerate from seed. In commercial forests, planting has been used to create even-aged, homogeneous stands at lower densities, which simplifies mechanical removal of competing vegetation and delays the first precommercial thinning (Lafuente et al., 2012). Direct sowing is currently limited, in part due to the high seed requirements and associated costs. Nevertheless, direct sowing can be effective on good sites, such as those with loose, stone-free soils, mild summer droughts, and even rainfall distribution throughout the year (de la Cruz Sevilla et al.,

Table 11.41 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Pinus pinaster*.

Attributes	Observed values (min-max)	Recommended values
Shoot height (cm)	12–40	>20
Root collar diameter (cm) (cm)	0.2–0.5	>0.3
Shoot dry weight (g)	1.2–5.1	>3
Root dry weight (g)	0.4–2.0	>1.5
Plant dry weight (g)	1.6–7.2	>4.5
Shoot/root dry weight ratio	2.7–3.6	≈2
Needles [N] (mg/g)	17.1–21.5	>18
Needles [P] (mg/g)	1.8–4.4	>2
Needles [K] (mg/g)	3.2–8.9	>5
Root [N] (mg/g)	5.0–10.7	>7
Root [P] (mg/g)	1.2–2.1	>1.5
Root [K] (mg/g)	0.9–1.8	>1.5

European Council (2000); Lafuente et al. (2012); Rodríguez Soalleiro et al. (1997) for recommended values; Al Pavel et al. (2024); Mañas et al. (2009) *and unpublished results by Toca* et al. *(pers comm) for observed values.*

1989). For direct sowing, tilling or scarification are required to remove herbaceous vegetation and ensure good seed-to-soil contact. Shrubs must also be eliminated through slashing or burning, especially when required by pest management regulations, such as in Portugal (Soares et al., 2020). Sowing can be done in lines, strips, or broadcast, although the latter is not recommended. Soil preparation should be shallow (20–30 cm) as deeper preparation may impoverish the fine fertile topsoil of the seed bed upper horizon, reduce soil moisture, and hinder seedling emergence and survival, particularly if summer drought is intense (Lafuente et al., 2012). The sowing window depends on winter and spring temperatures and soil water retention, with waterlogging being particularly detrimental for germination and emergence. Seed application rates vary from 5–15 kg/ha depending on sowing methods (Catalán, 1991). In France's Landes, mechanical seeders sow rows 4 m apart, using mechanical seeders and applying 2–3 kg of seed/ha. Rates of 3 kg/ha are applied in Central Spain for broadcast seeding after fires (unpublished results by Iglesias).

Competing vegetation must also be removed before planting, especially on productive sites. We recommend mechanical soil preparation, especially for poorly drained soils because this species is particularly susceptible to low aeration. This can be done by subsoiling to a depth of 50 or 60 cm. Soil preparation also can be done by digging holes at least 40 × 40 × 40 cm. The planting window will depend on climate and meteorological conditions of the year and stock type. In Atlantic climates, spring planting is best for container plants, while planting bareroot plants is better suited to autumn to provide more time for the root system to regenerate before shoot growth begins in the spring. In Mediterranean climates, planting must be completed before the onset of summer drought, but also considering limitations due to the frost period. In Atlantic environments, with mild winters and wet summers, planting can be extended to late spring.

Initial planting densities for timber production vary from 1000 to 1670 seedlings/ha, depending on site quality; lower densities are preferred for poor sites (Soares et al., 2020) or genetically improved plantations. Typical quadrangular planting frames are 2.5 × 2.5 m to 3.0 × 3.0 m while rectangular frames range from 3.0 × 2.1 m to 4.0 × 2.0 m. On steep or uneven slopes, frame size can be modified, and planting can be done along contour lines. If densities exceed 1600 stems/ha, precommercial thinning will be necessary after 8 or 10 years to reduce competition. For low-density planting (1000–1350 stems/ha), typical for genetically improved material, it is important to minimize failures, promptly replacing dead seedlings to maintain the desired density.

During the fourth and fifth year, plantations may require release interventions depending on pine growth and the competitive ability of the spontaneous vegetation (Rodríguez Soalleiro et al., 1997). Herbaceous vegetation removal is particularly important, and spraying glyphosate has proven to be most effective method. On old agricultural land application of this herbicide 2 years after planting at 3 L/ha effectively reduces

competition. Care must be taken to avoid the herbicide directly contacting *P. pinaster* plants or the chemical being transferred by the wind or by dripping from faulty nozzles (Lafuente et al., 2012). Applying glyphosate prior to planting at a 1% v:v rate has also been beneficial for seedling survival and soil properties compared with mechanical vegetation removal (Fidan et al., 2022).

Fertilization during the early years after planting can speed seedling growth and improve competitive capacity against surrounding scrub, especially on low-to medium-fertility soils such as heathlands. Fertilization must be carried out in spring, within 2 months of planting. Typically, slow release N, P and K fertilizers (8-24-16) are applied directly around each plant or in mixture with the soil of the planting hole at a rate of 50–70 g/tree (Lafuente et al., 2012).

Irrigation is not usually applied for *Pinus pinaster*, due to moderate summer drought in most of its habitats. However, in marginal areas or specific limiting conditions such as sandy soils, postplanting watering during the first summer with 2 L every other week from June to September can significantly improve survival of *P. pinaster* (Al Pavel et al., 2024).

Chapter References

Al Pavel, M.A., Bravo, F., Ordóñez, C., Rodríguez-García, E., 2024. Linking the effects of nursery light and fertilization and post-transplant first summer irrigation on the field survival and growth of *Pinus pinaster* seedlings. N. For. 55, 251–271. https://doi.org/10.1007/s11056-023-09973-x.

Alía, R., García del Barrio, J.M., Iglesias, S., Mancha, J.A., De Miguel, J., Nicolás, J.L., Pérez Martín, F., Sánchez Ron, D., 2009. Regiones de Procedencia de las especies forestales en España. In: Organismo Autónomo Parques Nacionales. Madrid. Spain.

Alía, R., Martín, S., De Miguel, J., Galera, R.M., Agúndez, D., Gordo, J., Salvador, L., Catalán, G., Gil, L., 1996. Regiones de procedencia: *Pinus pinaster* Aiton. In: Organismo Autónomo Parques Nacionales, Madrid. Spain.

Alonso-Graña, M., 2010. Ectomicorrización inducida en vivero sobre *Pinus pinaster* Aiton. y Betula pubescens Ehrh.: especies fúngicas y métodos de inoculación. Escuela Politécnica de Mieres, Ingeniería Técnica Forestal. Universidad de Oviedo, Mieres.

Álvarez, J.M., Majada, J., Ordás, R.J., 2009. An improved micropropagation protocol for maritime pine (*Pinus pinaster* Ait.) isolated cotyledons. Forestry 82, 175–184. https://doi.org/10.1093/forestry/cpn052.

Álvarez, P., 2004. Viveros forestales y uso de planta en repoblación en Galicia. In: Escuela Politécnica Superior de Lugo. Universidad de Santiago de Compostela, Santiago de Compostela.

Álvarez, R., Valbuena, L., Calvo, L., 2007. Effect of high temperatures on seed germination and seedling survival in three pine species (*Pinus pinaster, P. sylvestris* and *P. nigra*). Int. J. Wildland Fire 16, 63–70. https://doi.org/10.1071/WF06001.

Cabal, A., Kidelman, A., Ortega, U., Duñabeitia, M., Majada, J., 2005. Influencia de la poda química en la biomasa y desarrollo radical de *Pinus pinaster* Ait. y *Pinus radiata* D. Don. Investigación agraria. Sistemas y recursos forestales 14, 52–63.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: ICONA. Colección Técnica., Madrid, Spain.

Catalán, G., 1977. Semillas de árboles y arbustos forestales. Instituto para la Conservación de la Naturaleza. Ministerio de Agricultura, Madrid. Spain.

Climent, J., Alizoti, P., Rodriguez-Quilón, I., Kurt, Y., Ducci, F., Fady, B., Alía, R., 2021. Conservation and breeding of mediterranean pines. In: Ne'eman, G., Osem, Y. (Eds.), Pines and Their Mixed Forest Ecosystems in the Mediterranean Basin. Springer.

Climent, J., Dantas, A.K., Alia, R., Majada, J., 2013. Clonal variation for shoot ontogenetic heteroblasty in maritime pine (*Pinus pinaster* Ait.). Trees Struct. Funct. 27, 1813–1819. https://doi.org/10.1007/s00468-013-0901-1.

Correia, I., Santos, L., Faria, C., No, C., Almeida, H., David, T., 2014. Cone to seedling variation between *Pinus pinaster* provenances from contrasting altitudes. For. Sci. 60, 724–732. https://doi.org/10.5849/forsci.12-044.

de la Cruz, Sevilla A.L., Medrano Rivas, J.M., Nicolás Zabala, J.M., 1989. Siembra. In: Molina, J.L., Navarro, M., Montero de Burgos, J.L., Herranz, J.L. (Eds.), Técnicas de forestación en países mediterráneos. Instituto para la Conservación de la Naturaleza. Ministerio de Agricultura, Pesca y Alimentación, Madrid, Spain, pp. 61–63.

de las Heras, J., Martínez-Sánchez, J.J., González-Ochoa, A.I., Ferrandis, P., Herranz, J.M., 2002. Establishment of *Pinus halepensis* Mill. saplings following fire: effects of competition with shrub species. Acta Oecol 23, 91–97.

European Council, 2000. Council Directive 1999/105/EC of 22 December on the Marketing of Forest Reproductive Material. Council Directive, Brussels.

Fidan, C., Eşen, D., Demirsu, N., Cabak, E., Güner, S., 2022. Effects of site preparation and postplanting cultural treatments on the establishment and early growth of maritime pine (*Pinus pinaster* Ait.). Turk. J. Agric. For. 46, 613–622. https://doi.org/10.55730/1300-011X.3030.

Fraysse, J.Y., Crémière, L., 1998. Nursery factors influencing containerized *Pinus pinaster* seedling's initial growth in the nursery. Silva Fenn. 32, 261–270.

Fraysse, J.Y., Canlet, F., 2017. Gestion de la qualite des plants de pin maritime en pepiniere. FCBA Info.

González-Ochoa, A.I., De Las Heras, J., Torres, P., Sánchez-Gómez, E., 2003. Mycorrhization of *Pinus halepensis* Mill. and *Pinus pinaster* Aiton seedlings in two commercial nurseries. Ann. For. Sci. 60, 43–48. https://doi.org/10.1051/forest.

Herrero, N., Nicolás, J.L., 2012. Anejo III: Datos característicos de lotes de semillas. In: Pemán, J., Navarro, R., Nicoás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo de Parques Nacionales. Serie Forestal, Madrid. Spain, pp. 736–739.

Ingestad, T., Lund, A.B., 1986. Theory and techniques for steady state mineral nutrition and growth of plants. Scand. J. For. Res. 1, 439–453. https://doi.org/10.1080/02827588609382436.

Jaramillo-Correa, J.P., Bagnoli, F., Grivet, D., Fady, B., Aravanopoulos, F.A., Vendramin, G.G., González-Martínez, S.C., 2020. Evolutionary rate and genetic load in an emblematic Mediterranean tree following an ancient and prolonged population collapse. Mol. Ecol. 29, 4797–4811. https://doi.org/10.1111/mec.15684.

Krasowski, M.J., 2003. Root system modifications by nursery culture reflect on post-planting growth and development of coniferous seedlings. For. Chron. 79, 882–891. https://doi.org/10.5558/tfc79882-5.

Lafuente, D., Rodríguez-Núñez, L., Majada, J., 2012. *Pinus pinaster* Aiton. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Tomo I. Organismo Autónomo Parques Nacionales, pp. 900–927 (Madrid. Spain).

Lario, F., Ocaña, L., Rodríguez, J.R., Gómez, J.A., 2005. Calidad de forma del fuste de *Pinus pinaster* Aiton en clima atlántico. Modelos radiculares de brinzales producidos en vivero según el sistema de cultivo. Primer paso para la predicción de calidad mecánica y de forma. In: Actas Del IV Congreso Forestal Español. Sociedad Española de Ciencias Forestales, pp. 1–8. Zaragoza.

Lazcano, C., Sampedro, L., Zas, R., Domínguez, J., 2010. Assessment of plant growth promotion by vermicompost in different progenies of maritime pine (*Pinus pinaster* Ait.). Compost Sci. Util. 18 (2), 111–118. https://doi.org/10.1080/1065657X.2010.10736943.

Maia, P., Corticeiro, S., 2022. From cone to seed and seedling—characterization of three Portuguese *Pinus pinaster* Aiton populations. Seeds 1, 126–135. https://doi.org/10.3390/seeds1020011.

Majada, J., Martínez-Alonso, C., Feito, I., Kidelman, A., Aranda, I., Alía, R., 2011. Mini-cuttings: an effective technique for the propagation of *Pinus pinaster* Ait. N. For. 41, 399–412. https://doi.org/10.1007/s11056-010-9232-x.

Majada, J., Martínez-Alonso, C., Feito, I., Kidelman, A., Aranda, I., Alía, R., 2011. Mini-cuttings: an effective technique for the propagation. N. For. 41, 399–412. https://doi.org/10.1007/s11056-010-9232-x.

Mañas, P., Castro, E., De Las Heras, J., 2009. Quality of maritime pine (*Pinus pinaster* Ait.) seedlings using waste materials as nursery growing media. N. For. 37, 295–311. https://doi.org/10.1007/s11056-008-9125-4.

Marcelli, A.R., 1989. Moderne tecnologie per la produccione vivaística di spacie forestali. In: SAF Istituto Sperimentale per la Pioppicoltura. Italy.

Martínez-Alonso, C., Kidelman, A., Feito, I., Velasco, T., Alía, R., Gaspar, M.J., Majada, J., 2012. Optimization of seasonality and mother plant nutrition for vegetative propagation of *Pinus pinaster* Ait. N. For. 43, 651–663. https://doi.org/10.1007/s11056-012-9333-9.

Molina, L., 1994. Estudio de la serotinidad de los conos en *Pinus pinaster* Aiton, BSc Thesis ETSI Montes. Universidad Politécnica de Madrid, Spain.

Moraldi, M., 2024. Vivaistica forestale - Conoscerla per praticarla con successo. Compagnia delle Foreste, Arezzo. Italia. 221pp.

Navarro, R.M., Del Campo, A.D., Cortina, J., 2006. Factores que afectan al éxito de una repoblación y su relación con la calidad de la planta. In: Cortina, J., Peñuelas, J.L., Puértolas, J., Vilagrosa, A., Savé, R. (Eds.), Calidad de Planta Forestal Para La Restauración En Ambientes Mediterráneos. Estado Actual de Conocimientos. Organismo Autónomo Parques Nacionales. Ministerio de Medio Ambiente, Madrid, pp. 31–46.

Ocaña Bueno, L., Dominguez Lerena, S., Carrasco Manzano, I., Peñuelas Rubira, L., Herrero, S.N., 1997. Influencia del tamaño de la semilla y diferentes dosis de fertilización sobre el crecimiento y supervivencia en campo de cuatro especies forestales. In: Actas II Congreso Forestal Español. Mesa 3. Sociedad Española de Ciencias Forestales. Granada, pp. 461–466.

Ocaña Bueno, L., Lario Leza, F.J., Gómez Fernández, J.A., Rodríguez Gallego, R., 2004. La mejora de la estabilidad en las repoblaciones con *Pinus pinaster* Aiton. en el norte de España. Cuadernos de la Sociedad Española de Ciencias Forestales 17, 211–215.

Ocaña, L., Santos, M.I., Gómez, J.A., Renilla, I., Cuenca, B., 2001. Comparación de siete modelos de contenedores y raíz desnuda en repoblaciones de *P. pinaster* en Galicia. In: Actas del III Congreso Forestal Español. Tomo II. Sociedad Española de Ciencias Forestales, Granada, pp. 736–741.

Oliet, J.A., Puértolas, J., Planelles, R., Jacobs, D.F., 2013. Nutrient loading of forest tree seedlings to promote stress resistance and field performance: a Mediterranean perspective. N. For. 44, 649–669. https://doi.org/10.1007/s11056-013-9382-8.

Oliveira, R.S., Franco, A.R., Castro, P.M.L., 2012. Combined use of *Pinus pinaster* plus and inoculation with selected ectomycorrhizal fungi as an ecotechnology to improve plant performance. Ecol. Eng. 43, 95–103. https://doi.org/10.1016/j.ecoleng.2012.01.021.

Park, Y.-S., Beaulieu, J., Bousquet, J., 2016. Multi-varietal forestry integrating genomic selection and somatic embryogenesis. In: Park, Y.-S., Bonga, J., Moon, H. (Eds.), Vegetative Propagation of Forest Trees. National Institute of Forest Science (NIFoS), Seoul, Korea.

Peñuelas, J.L., Ocaña, L., 1996. Cultivo de plantas forestales en contenedor. Principios y fundamentos. MAPA. Mundi Prensa, Madrid.

Pera, J., Parladé, J., 2005. Inoculación controlada con hongos ectomicorrícicos en la producción de planta destinada a repoblaciones forestales: estado actual en España. Investigación Agraria. Sistemas y Recur. Natur. 14, 419–433.

Pita, J.M., Sanz, V., Escudero, A., 1998. Seed cryopreservation of seven Spanish native pine species. Silvae Genet. 47, 220–223.

Puértolas, J., Jacobs, D.F., Benito, L.F., Peñuelas, J.L., 2012. Cost-benefit analysis of different container capacities and fertilization regimes in *Pinus* stock-type production for forest restoration in dry Mediterranean areas. Ecol. Eng. 44, 210–215. https://doi.org/10.1016/j.ecoleng.2012.04.005.

Rigueiro, A., Mosquera, M., Vila, T., 2001. Efecto de la proporción de distintos componentes del sustrato en el crecimiento de *Pinus pinaster* Aiton en envase en vivero. III Congreso Forestal Español. Mesa 3, 410–415.

Rodríguez Soalleiro, R., Álvarez González, J.G., Cela González, M., Mansilla Vázquez, P., Vega Alonso, P., González Rosales, .M., Ruiz Zorrilla, P., Vega Alonso, G., 1997. Manual técnico de selvicultura de *P. pinaster*. Universidad de Santiago de Compostela, 75 pp, Santiago de Compostela.

Rosvall, O., Bradshaw, R.H.W., Egertsdotter, U., Ingvarsson, P.K., Wu, H., 2021. Using Norway spruce clones in Swedish forestry: introduction. Scand. J. For. Res. 34, 333–335. https://doi.org/10.1080/02827581.2018.1562565.

Ruiz de la Torre, J., 2006. Flora mayor. In: Organismo Autónomo Parques Nacionales.

Sanchez-Zabala, J., Majada, J., Martín-Rodrigues, N., Gonzalez-Murua, C., Ortega, U., Alonso-Graña, M., Arana, O., Duñabeitia, M.K., 2013. Physiological aspects underlying the improved outplanting performance of *Pinus pinaster* Ait. seedlings associated with ectomycorrhizal inoculation. Mycorrhiza 23, 627–640. https://doi.org/10.1007/s00572-013-0500-4.

Soares, P., Calado, N., Carneiro, S., 2020. Manual boas praticas pinheiro-bravo. In: Centro PINUS Associação para a Valorização da Floresta de Pinho.

Tapias, R., Gil, L., 2000. Adaptación reproductiva de las especies forestales ante el fuego. In: La Defensa Contra Incendios Forestales. Fundamentos Y Experiencias. McGraw-Hill, Madrid, pp. 4–36.

Timmer, V.R., 1996. Exponential nutrient loading: a new fertilization technique to improve seedling performance on competitive sites. N. For. 13, 279–299. https://doi.org/10.1023/A:1006502830067.

Toca, A., Oliet, J.A., Villar-Salvador, P., Maroto, J., Jacobs, D.F., 2018. Species ecology determines the role of nitrogen nutrition in the frost tolerance of pine seedlings. Tree Physiol. 38, 96–108. https://doi.org/10.1093/treephys/tpx165.

Toca, A., Oliet, J.A., Villar-Salvador, P., Martínez Catalán, R.A., Jacobs, D.F., 2019. Ecologically distinct pine species show differential root development after outplanting in response to nursery nutrient cultivation. For. Ecol. Manag. 451. https://doi.org/10.1016/j.foreco.2019.117562.

Trontin, J.F., Rupps, A., Raschke, J., et al., 2019. Multiforever: Towards Intensification of Conifer Production through Multi-Varietal Forestry Based on Somatic Embryogenesis. Finland, Helsinki.

Trontin, J.F., Teyssier, C., Harvengt, L., Lelu-Walter, M.-A., 2016. Prospects for New Variety Deployment through Somatic Embryogenesis in Maritime Pine. National Institute of Forest Science Institute (NIFoS) ffhal-02795079f.

Villar-Salvador, P., Puértolas, J., Cuesta, B., Peñuelas, J.L., Uscola, M., Heredia-Guerrero, N., Rey Benayas, J.M., 2012. Increase in size and nitrogen concentration enhances seedling survival in Mediterranean plantations. Insights from an ecophysiological conceptual model of plant survival. N. For. 43, 755–770. https://doi.org/10.1007/s11056-012-9328-6.

Wahid, N., Hadrami, I.E., Boulli, A., 2007. Variabilité du potentiel germinatif des semences de certaines populations marocaines de pin maritime (*Pinus pinaster* Ait.) sous conditions contrôlées. Acta Bot. Gall. 154 (1), 7–19. https://doi.org/10.1080/12538078.2007.10516040.

Wahid, N., Hadrami, I.E., Boulli, A., 2007. Variabilité du potentiel germinatif des semences de certaines populations marocaines de pin maritime (*Pinus pinaster* Ait.) sous conditions contrôlées. Acta Bot. Gall. 154 (1), 7–19. https://doi.org/10.1080/12538078.2007.10516040.

Pinus pinea L.

Rafael M. Navarro Cerrillo[1], Marianthi Tsakaldimi[2], Antonio J. Molina[1] and Pedro Villar-Salvador[3]
[1]Department of Forestry, Faculty of Agriculture and Forestry, University of Córdoba, DendrodatLab-ERSAF, Laboratory of Dendrochronology, Silviculture and Climate Change, Córdoba, Spain; [2]Laboratory of Silviculture, Department of Forestry and Natural Environment, Aristotle University of Thessaloniki, Thessaloniki, Greece; [3]Universidad de Alcalá, Forest Ecology and Restoration Group, Departamento de Ciencias de la Vida, Alcalá de Henares, Madrid, Spain

Propagation

Seed propagation

Pinus pinea (Stone pine) is easily propagated by seeds. Cone production begins when individuals reach 15–20 years, which is older than other Mediterranean pines. Cones ripen in late April, about 3 years after pollination (Castro-García et al., 2011; Ganatsas et al., 2008; Ganatsas and Tsakaldimi, 2007; Gonçalves and Pommerening, 2012; Montero et al., 2004). Annual seed production is low, depends on tree density, and typically ranges from 103,000 to 219,000 seeds/ha (Ganatsas and Thanasis, 2010). Annual variation in cone production is significant. Production is positively related to crown size (Gonçalves and Pommerening, 2012). The number of cones per tree can vary from 2.4 to 57, cone weight from 100 to 474 g, while cone length and width vary between 75 and 182 mm and 63 and 145 mm, respectively (Balekoglu et al., 2020; Castro-García et al., 2011; Ganatsas et al., 2008; Loewe-Muñoz et al., 2023, 2024b).

Traditionally, cone harvesting was performed manually by climbing the trees. Currently, harvesting is mechanical, conducted mainly with mobile elevators and trunk shakers (Castro-García et al., 2011). After harvesting, cones easily open when placed under the sun or in an oven at 54°C with air circulation and a relative humidity of 20%. Long-term seed storage for conservation purposes can be accomplished at 4°C. Storage at room temperature strongly reduces long-term germination capacity (García de Castro and Martinez-Honduvilla, 1984). Seed moisture during storage varies between 5% and 10%.

Seed length and diameter vary between 13.8 and 20.0 mm and 7.1–9.2 mm, respectively (Balekoglu et al., 2020; Ganatsas et al., 2008; Ganatsas and Thanasis, 2010). Other main seed characteristics are shown in Table 11.42. Seed provenance may affect seed size

Table 11.42 Characteristics of seed lots of *Pinus pinea*.

Cone to seed yield (% in weight)	Number of developed seeds per cone	Purity (%)	No. Seeds/ kg	Germination Capacity (%)
7.5–18	25.0–115	90–100	1,200–2,400	65–100

Balekoglu et al. (2020); Castro-García et al. (2011); Catalán (1991); Court-Picon et al. (2004); Ganatsas et al. (2008); Ganatsas and Thanasis (2010); García-Fayos (2001); Gradi (1989); Herrero Sierra and Nicolás Peragón (2013); Kadri et al. (2015); Loewe-Muñoz et al. (2024b, 2023); Montero et al. (2004); Navarro-Cerrillo and Gálvez (2001); Nergiz and Dönmez (2004); Picher Morelló et al. (2013); Ribeiro et al. (2001)

and consequently germination capacity and initial juvenile height growth (Court-Picon et al., 2004).

Seeds do not need any pretreatment to germinate, which is high at 15 to 20°C for fresh seeds and does not depend on light conditions (Ganatsas and Tsakaldimi, 2007; Renaldi et al., 2003). Germination is reduced at a higher constant temperature of 25°C or at alternating temperatures between 15 and 25°C (Escudero et al., 2002).

Vegetative propagation

Vegetative propagation of *P. pinea* has been favored due to interest in it as a pinyon (pine nuts, piñón, pinoli, or pignoli, the edible seeds of pines) producer (Castaño et al., 2004). While vegetative propagation via cuttings is feasible, grafting remains the prevailing method. Grafting has been extensively promoted to increase the potential of this species as a fruit-bearing tree (Catalán and Catalán, 1996; Butler et al., 1997; Mutke and Díaz, 2001; Guàrdia et al., 2021; Pérez-Luna et al., 2020). Castaño et al. (2004) conducted a comprehensive review of grafting techniques used for *P. pinea*. Grafting allows the production of adult specimens from young (5- or 6-year-old) rootstocks. The most common grafting technique for *P. pinea* on *P. pinea* is cleft or terminal leader replacement grafting onto 5- or 6-year-old hypobionts (rootstock), with a height of 1.5 m (Cortizo et al., 2009). Grafts are also compatible with *Pinus halepensis* (Piqué et al., 2017). The optimal time for grafting in southwestern Spain is from early March to late April. Scions are extracted from the terminal whorls of the adult tree, where female flowers emerge, and from lateral shoots, when the plant is dormant in a vegetative state. Grafting is difficult and should be carried out by well-trained personnel. A comprehensive description can be found in Cortizo et al. (2009).

In vitro propagation techniques for the species have been devised from immature zygotic embryos (Cortizo et al., 2009). Recent research has been conducted on the in vitro propagation of stone pine, with most studies focusing on biochemical responses (Cortizo et al., 2009, 2010). However, these findings could potentially be applied in the mass production of plants from selected materials.

Seedling cultivation

Container plants

Currently, *P. pinea* is cultivated exclusively as container stock. *P. pinea* is typically sown at the same time as other Mediterranean pines such as *P. pinaster* and *P. halepensis* and cultivated for about 35 weeks. Seedlings are cultivated easily in the nursery. However, this species is susceptible to unbalanced height growth (high slenderness), limited development of secondary roots that limits forming a consistent plug, and it is vulnerable to frosts, requiring suitable cultivation. Variations in seedling morphological and physiological attributes can be affected by seed source (Amaro et al., 2000; Catalán et al.,

1997; Palacios and Navarro Cerrillo, 2001). Depending on seed quality, the number of seeds per container cell should range from one to two, exceptionally three and early thinning will be needed if all seeds germinate. Germination occurs 3–4 weeks after sowing. The cultivation calendar should be adjusted in cold locations to avoid risk of frost damage. Nevertheless, the sowing date should not be significantly delayed to prevent damping-off damage.

The container used largely determines the final seedling morphology, so it should be chosen considering other cultivation variables affecting plant growth such as the cultivation cycle, fertilization, partial shading, and irrigation management. The typical *P. pinea* seedling for forest restoration is cultivated in containers whose volume varies between 200 and 400 mL (Domínguez Lerena et al., 1997, 2000a, 2006; Villar-Salvador et al., 2013), at a density between 200 and 300 seedlings/m^2 (Fig. 11.29). Although large container volumes usually enhance field survival and growth (Dominguez-Lerena et al., 2006), the cost-effective recommendation for *P. pinea* is containers with 300 mL volume (Puértolas et al., 2012). Cultivation at low densities is possible and desirable with this container volume, resulting in seedlings with more balanced shoot to root dry weight ratios (Dominguez-Lerena et al., 2006). If cultivated properly, the height of 1 + 0 *P. pinea* seedlings is usually at least 20 cm and a root-collar diameter >0.3 cm. The effects of container type on the quality of *P. pinea seedlings* are similar to those observed in other pines, whereby plant development, especially roots increase with container volume (Domínguez-Lerena et al., 2006; Puértolas et al., 2012). If the objective of *P. pinea* plantations is pinyon production, grafted, large seedlings cultivated in large containers (1000 cm^3) can be used.

Except during sowing and the beginning of the germination phase, cultivation under shade is not recommended because it increases the shoot to root mass ratio and induces other morphological imbalances (personal experience of the authors). If some shading is needed, white meshes are preferred as they do not alter light quality and therefore do not promote etiolation. Cultivation can be carried out in greenhouses, although it is very uncommon and unnecessary in Mediterranean environments.

The type of growing media influences seedling characteristics as much as the container does. In *P. pinea*, substrate porosity is important, and it should be rather high to promote root system fibrosity (air volume at 10 cm water column >10%; easily available water >32%). The best growing medium in terms of plant quality has peat, humus peat, coconut fiber, or pine bark as the main organic component, mixed with an inorganic component such as vermiculite or perlite (Hammad, 2012). The most common mixture is peat and perlite (3:1 by volume), although coir is progressively replacing peat. Other formulations recommended for *P. pinea* include peat and pine bark (1:1 by volume), or peat:coir:perlite (6:3:1 by volume) is used in some commercial nurseries (Navarro Cerrillo et al., 2009a; 2009b; Ruano et al., 2001). Growing media pH must be slightly acidic, given the soil requirements of the species. Alkaline pH values can result



Figure 11.29 One-year old *Pinus pinea* seedlings in the nursery containers (left) and in a rooting experiment (right). *(Pedro Villar-Salvador.)*

in nutrient deficiencies, notably in Fe and P, to which *P. pinea* shows a high sensitivity (Navarro Cerrillo et al., 2001).

Fertilization is necessary despite that some nurseries that grow *P. pinea* with deficient fertilization can achieve seedlings with acceptable morphology. This species is less demanding in nutrients compared with other pines due to its large seed. However, nitrogen fertilization and proper management of macro and micronutrients significantly contribute to improved plant quality (Arduini et al., 1995; Fernández et al., 2000b; Villar-Salvador et al., 2013). The effects of nutritional deficiencies on the seedlings of *P. pinea* have been described by Navarro Cerrillo et al. (2001b). Generally, moderate fertilization rates are recommended with balanced NPK ratios, such as 120–50–100 mg/L (Domínguez Lerena et al., 2000b) during the fast growth stage and 40–30–120 mg/L during hardening. In a container comparison experiment, Dominguez-Lerena et al. (2006) fertigated each plant with 68 mg N, 50 mg P, and 40 mg K during the growth phase. The seedlings cultivated in 400 mL containers (container model ForestPot 400) attained a final height of 21 cm, a root collar diameter of 3.8 mm, a total dry mass of 4 g and a shoot N concentration of 10 mg/g. If hard water is used for irrigation, the addition of Fe chelates is necessary, along with substrate pH control.

Large and nutrient-rich *P. pinea* seedlings show superior outplanting performance over those with the opposite attributes (Cañellas et al., 1999; Villar-Salvador et al., 2000, 2013). This is in part due to their greater root growth capacity (Toca et al.,

2019) Fig. 11.29, a key attribute for plant establishment in arid climates (Padilla and Pugnaire, 2007). Maintaining high fertilization rates in the final stages of cultivation in autumn increases the survival and growth of *P. pinea,* at a very low cost (Puértolas et al., 2012).

An alternative to fertigation is supplying slow-release fertilizer to the growing medium. It is common to use rates of 2 g/L of an NPK 18-11-10 fertilizer or 2.5 g/L of an NPK 14-8-15 fertilizer, in both cases with a release time of 8–9 months. With these fertilization rates, no growth problems or nutritional deficiencies have been observed in 1 + 0 seedlings. For short-duration crops, a valid option is the use of fertilized peat, with a base fertilization of NPK 16-8-16 with a pH corrector (2 kg/m^3 dolomite, 5% Mg), supplemented with a maintenance fertilization according to the crop duration and plant type (Navarro Cerrillo et al., 2012).

A slight restriction of irrigation can be an effective tool to control excessive aboveground growth of *P. pinea* (Kildisheva et al., 2017), along with low cultivation density. To achieve this, it is advisable to use a growing medium that will not become hydrophobic when exposed to moderate to high desiccation as occurs with blonde peat. Several studies have addressed the effect of different irrigation regimes on plant quality (Fernández et al., 2000a). Drought conditioning at the end of the vegetative period is not necessary since it does not significantly increase outplanting performance (Villar-Salvador et al., 2013). In addition, its control at the operational scale is difficult.

Mycorrhization with ectomycorrhizal fungi can improve the survival of seedlings in forest plantations. Some morphotypes from natural and/or planted pine forests are known (Colón Flores et al., 2001; Delard et al., 2023; Loewe-Muñoz et al., 2024c; Lucas García et al., 2004; Parladé et al., 2007; Probanza et al., 2002). In a nursery trial, Rincon et al. (2001) observed that mycorrhization with *Hebeloma crustuliniforme* and *Rhizopogon* spp. showed the highest infective capacity, with results analogous to those observed by Parladé et al. (2004) in agricultural fields. The combined effect of ectomycorrhizal fungi (*Pisolithus tinctorius*) and bacteria of the genus *Bacillus* (*B. licheniformis* and *B. pumilus*) has also been studied, although the effects on pine performance were not pronounced (Colón Flores et al., 2001). Mycorrhization with the truffle (*Tuber borchii)* in nurseries is also possible, increasing the commercial value of *P. pinea* plantations (Delard et al., 2023). Similarly, mycorrhization of *P. pinea* in nurseries with other edible species, particularly *Lactarius edulis*, has been proposed (Rincon et al., 2001). Despite their positive effect and potential benefits, mycorrhization is not commonly used in commercial *P. pinea* nurseries due to operational difficulties and the challenge of integrating this practice with other cultivation practices such as fertilization (Honrubia et al., 1997), growing media (Rincón et al., 1997), and irrigation (Honrubia et al., 1992; Rincón et al., 1997; Parladé et al., 2007). However, natural mycorrhization occurs relatively easily in nurseries, which likely contributes to improved plant quality.

Table 11.43 Morphological and physiological attributes of 1 + 0 container seedlings of *Pinus pinea* and recommended values of high-quality seedlings.

Attributes	Observed values	Recommended values
Shoot height (cm)	9–32	25–35
Root collar diameter (cm)	0.31–0.56	>0.38
Shoot dry weight (g)	1.5–8	>3.7
Root dry weight (g)	1.1–4.0	>1.7
Plant dry weight (g)	2.5–11	>4.5
Shoot/root dry weight ratio	1.7–3.1	2.0–2.5
Leaf [N] (mg/g)	8.9–23	>13
Leaf [P] (mg/g)	1.8–3	>2.0
Leaf [K] (mg/g)	4.5–12	>6
Leaf [Ca] (mg/g)	2.4–5.5	>3
Leaf [mg] (mg/g)	1.4–3.8	>1.5
Shoot [N] (mg/g)	8.9–11.5	>10
Shoot [P] (mg/g)	0.84–0.97	>0.9
Shoot [K] (mg/g)	5.2–7.6	>5.5
Root [N] (mg/g)	7.7–8.8	>8.4
Root [P] (mg/g)	0.67–0.76	>0.70
Root [K] (mg/g)	1.2–2.0	>1.5

Cañellas et al. (1999); Delard et al. (2023); Dominguez-Lerena et al. (2006); Kildisheva et al. (2017); Navarro et al. (2006); Navarro Cerrillo et al. (2012); Palacios and Navarro Cerrillo (2001); Puértolas et al. (2012); Villar-Salvador et al. (2013, 2000); Villar-Salvador unpublished results on shoot P and K concentration.

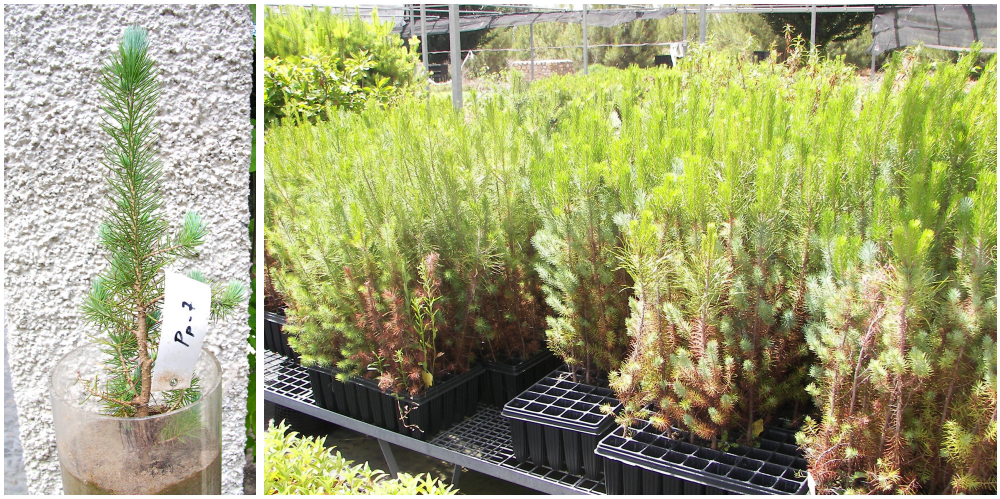


Figure 11.30 *Pinus pinea* high quality seedling (left) and unbalanced 2 + 0 seedlings (right). *(Pedro Villar-Salvador.)*

The typical and recommended values of morphological attributes and organ nutrient concentration for *P. pinea* are shown in Table 11.43. In general, it is a fast-growing species in the nursery, so cultivation should promote a balanced growth with moderate slenderness (height/root collar diameter) and lateral branches (or twigs), avoiding unbalanced, tall seedlings (height >35 cm) (Fig. 11.30). The biomass distribution between the shoot and root (shoot to root mass ratio) must be balanced, with average values clearly over 1.5.

Bareroot plants

As the plantations of this species are performed under dry Mediterranean climate conditions, we recommend using container plants as they are less sensitive to harsh planting conditions than bareroot plants (Grossnickle and El-Kassaby, 2016).

Field establishment

P. pinea is native to the Mediterranean basin. It has high ecological, esthetic and economic importance. It performs well in poor, eroded soils, and has high drought resistance, and its pinyons have high economic value. For these reasons, it has an important role in multipurpose forestry and nature conservation (Balekoglu et al., 2020; Ganatsas et al., 2008; Loewe-Muñoz et al., 2024b) (Fig. 11.31).

The most frequent method for establishing *P. pinea* forests, both for pinyon production and restoration, is planting. However, direct seeding has been recommended, particularly in sand dune restoration where high seedling densities are required. Both methods show comparable seedling survival and no single method is universally recommended for the species. Instead, the selection of the method should be based on site-specific conditions (Montero et al., 2004). Planting is typically performed manually. For direct seeding, two or three pinyons are placed in each spot, or seeds can be arranged in rows. In woodland areas, where implementing cultural practices is challenging, planting or seeding should be conducted in autumn. However, the establishment period can be extended to late winter on mid-mountain sites. Nevertheless, planting should be completed before February as later planting may compromise seedling survival, especially in rocky and shallow soils (Navarro Cerrillo et al., 2012).

The objective and the site conditions determine the type of plant used for planting. In most cases, we recommend using 1 + 0 container seedlings, cultivated in containers of 300 mL, with shoot height of 20–30 cm. For pinyon plantations, we recommend using 2 + 0 or 3 + 0 grafted plants with a shoot height of >50 cm, cultivated in large containers (>1000 mL) (Navarro Cerrillo et al., 2012).

Weed control before planting is done by clearing or tilling. Mechanized clearing using shrub cutters, including chain or hammer cutters, is one of the available clearing methods. Tilling with a harrow is the most common method used due its cost-effectiveness and ease of application. Shrub clearing treatments should be limited to areas with low-diversity thickets, such as *Cistus ladanifer* thickets. Spot clearing, whether done manually or with a brush cutter, is less common but offers the advantage of selective removal of competitors, without affecting shrub species that facilitate the establishment of tree species. This method is particularly beneficial in enrichment plantings (Navarro Cerrillo et al., 2012).

Soil preparation methods depend on forestation objectives. The most common techniques include subsoiling (single or preferably double) and ridging with deep plowing in cropland, the latter being used for cases where production is the main goal. In woodland areas, mechanical augers operated by tracked excavators may be used to create large holes (>50 cm deep). While manual drilling is occasionally employed, we do not recommend it due to significantly reduced seedling survival rates with low-intensity preparations (Navarro and Palacios, 2004).

Planting density recommendations for *P. pinea* vary widely among authors, reflecting the species adaptability to diverse ecological conditions. Planting densities typically range from 400 to 1200 (or even 1500) trees/ha, contingent upon the forestation objectives. In mixed plantings, such as *P. pinea*-*Quercus suber* combinations, densities can surpass 1000 trees/ha. For protective forestation projects, densities typically fall between 1200 and 1500 trees/ha. Optimal density for pinyon production in mature stands varies between 80 and 120 trees/ha (Montero et al., 2004). Several authors recommend higher initial densities (~300–400 trees/ha) for production plantations utilizing grafted plants (Loewe-Muñoz et al., 2020; Mutke et al., 2000, 2007), to facilitate tree selection for the future.

The functional response of seedlings during establishment has been studied by several authors (Amaro et al., 2000; Gordo Alonso et al., 2009; Puértolas et al., 2005; Sánchez-Gómez et al., 2009, 2011). The use of greenhouse tree shelters is quite uncommon in stone pine plantations, given the preference of the species for sunlight and dry conditions. The effect of tree shelters on rooting and growth of *P. pinea* has been studied (Carreras and Sánchez, 2001; Navarro Cerrillo et al., 2001a; Oliet et al., 2003). Findings from these studies together with field observations suggest that in regions with mild and cool winters, tree shelters are not necessary, seeming to be more suitable in highly xeric climates (Carreras and Sánchez, 2001).

Irrigation is generally not recommended for *P. pinea* reforestation, except under specific conditions such as intensive plantations, where approximately 50 L per tree supplied in presummer irrigations may be beneficial (Calama et al., 2007; Mutke et al.,

2007). Fertilization is also uncommon in *P. pinea* plantations. Cañellas et al. (1999) found no significant differences in 2-year survival with various fertilization treatments, although height growth was enhanced with Agroblen fertilizer (controlled release fertilizer, 9-13-18+3MgO + Fe, 12–14 months at 15 g/plant). This fertilizer is recommended for plantings on poor soils or areas with substantial herbaceous competition. Other studies reported no positive effects of fertilizers on survival (Cañellas et al., 2004; Loewe-Muñoz et al., 2020, 2024a), although some growth benefits were noted (Oliet et al., 2003). Higher growth and survival outcomes have been achieved with organic amendments (e.g., urban solid waste compost and plant residues) (Álvarez de la Puente et al., 2001; Larchevêque et al., 2006). Due to the ambiguous results and increased costs, fertilization is not done in most cases although in Chile it is a common practice. Greater emphasis is placed on high-quality nursery plants and appropriate site preparation. However, fertilization may be beneficial in intensive plantations, particularly with grafted plants, to regulate production (Calama et al., 2007).

Weed control is crucial during the one to 3 years following afforestation on former cropland (Cañellas et al., 1999; Navarro Cerrillo et al., 2009b). Plowing is the most used method for weed control in afforestation. Alternatives include shrub cutters, cultivators, or weeders. The application of herbicides has shown significant effects on survival, varying by type and dosage (Cañellas et al., 1999). Hexazinone was found to provide effective weed control at low dose (250 g/ha) although it is use in the EU is currently banned.

The most prominent pests affecting *P. pinea* plantations include the defoliating moth *Thaumetopoea pityocampa*, which causes issues in established stands. Other notable pests are borers such as *Tomicus piniperda*, *Pissodes castaneus*, *Hylobius abietis*, and *Retinia resinella* (all Coleoptera). In addition, the defoliating hymenopteran *Acantholyda hieroglyphica* primarily affects young stands. The primary diseases affecting young stands include *Lophodermium pinastri*, which causes defoliation of young branches; *Thyriopsis halepensis*,



Figure 11.31 *Pinus pinea* mixed plantation with the shrub *Retama sphaerocarpa* and *Quercus ilex*. *(Pedro Villar-Salvador.)*

leading to defoliation mainly in coastal sandy areas; and *Phellinus pini*, which causes trunk rot (Bertin et al., 2022).

Chapter References

Álvarez de la Puente, J., Del Campo, A., Sancho, F., 2001. Research and technological development of composting processes and its application in the agriculture and forestry fields. In: Proceedings of ORBIT 2001 on Biological Processing of Waste. Spanish Waste Club & ORBIT, Sevilla, Spain, pp. 107–113.

Amaro, J.A., Pardos, M., Agundez, D., Alia, R., 2000. Comportamiento de procedencias de *Pinus pinea* en cámara climática durante el primer año de crecimiento. 1er Simposio del Pino piñonero, Valladolid, Spain. Tomo II: 47–54 pp. In: Sociedad Española de Ciencias Forestales-Junta de Castilla y León.

Arduini, I., Godbold, D.L., Onnis, A., 1995. Influence of copper on root growth and morphology of *Pinus pinea* L. and *Pinus pinaster* Ait. seedlings. Tree Physiol. 15, 411–415. https://doi.org/10.1093/TREEPHYS/15.6.411.

Balekoglu, S., Caliskan, S., Dirik, H., 2020. Effects of geoclimatic factors on the variability in *Pinus pinea* cone, seed, and seedling traits in Turkey native habitats. Ecol. Process. 9, 1–13. https://doi.org/10.1186/s13717-020-00264-3.

Bertin, S., Ilardi, F., Scapini, C., Simoni, S., Roversi, P.F., 2022. Alien pest *Toumeyella parvicornis* (cockerell) (Hemiptera: Coccidae) on *Pinus pinea* L.: short time evaluation of endotherapic treatment. Redia 105, 11–16. https://doi.org/10.19263/REDIA-105.22.02.

Butler, I., Abellanas, B., Monteagudo, F., Bastida, F., López, J., 1997. Primeros resultados de una parcela de ensayo de técnicas de cultivo agronómico de injertos de pino piñonero en la finca experimental El Cebollar (Moguer, Huelva). In: Actas del II Congreso Forestal Español. Irati 97. Sociedad Española de Ciencias Forestales-Gobierno Foral de Navarra.

Calama, R., Madrigal, G., Candela, J.A., Montero, G., 2007. Effects of fertilization on the production of an edible forest fruit: stone pine (*Pinus pinea* L.) nuts in south-west Andalusia. Off. Syst. 16, 241–252. https://doi.org/10.5424/srf/2007163-01013.

Cañellas, I., Bachiller Bachiller, A., del Río Gaztelurrutia, M., Roig Gómez, S., Montero González, G., 2004. Efectos de la fertilización orgánica y mineral en el arraigo y desarrollo de especies mediterráneas durante los primeros años de la plantación. Cuadernos de la Sociedad Española de Ciencias Forest. 17, 139–143.

Cañellas, I., Bachiller Bachiller, A., Montero González, G., Finat, L., 1999. Comportamiento de planta de *Pinus pinea* en vivero y campo: ensayos de técnicas de cultivo de planta, fertilización y aplicación de herbicidas. Investigación agraria. Sistemas y recursos forestales 8, 335–360.

Carreras, C., Sánchez, J., 2001. Resultados de repoblación con especies de matorral en Vélez-Rubio (Almería), Actas del III Congreso Forestal Español. In: Sociedad Española de Ciencias Forestales-Junta de Andalucía, Granada, Spain, pp. 628–631.

Castaño, J., Estirado, M., Abellanas, B., Butler, I., Cosano, I., Luengo, J., García, J., Candela, J., 2004. Puesta en valor de los recursos forestales mediterráneos. El injerto de pino piñonero (*Pinus pinea* L.). In: Consejería de Medio Ambiente, Junta de Andalucía, Sevilla, Spain.

Castro-García, S., Blanco-Roldán, G.L., Gil-Ribes, J.A., 2011. Frequency response of *Pinus pinea* L. for selective cone harvesting by vibration. Trees Struct. Funct. 25, 801–808. https://doi.org/10.1007/s00468-011-0556-8.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: ICONA Madrid. 4a Ed, 4th Ed. Icona, Madrid, Spain.

Catalán, G., Catalán, G., 1996. Plantaciones mixtas de lavandín e injerto de pino piñonero en tierras agrícolas marginales. ITEA 17, 406–412.

Catalán, G., Dietl, B., Enciso, E., 1997. Selección de árboles sobresalientes, en cuanto a producción de fruto, en los pinares de pino piñonero en la región Valles del Tietar y del Alberche. Parámetros de selección. In: Actas del II Congreso Forestal Español. Irati 97, Tomo III: 141–145. Pamplona, Spain. Sociedad Española de Ciencias Forestales-Gobierno Foral de Navarra.

Colón Flores, J.J., Gutiérrez Mañero, F.J., Ruiz Palomino, M., Probanza Lobo, A., 2001. Incremento de parámetros biométricos y de la actividad biológica rizosférica de *Pinus pinea* mediante la utilización de baterias promotoras del crecimiento y ectomicorrizas, Actas del III Congreso Forestal Español. In: Sociedad Española de Ciencias Forestales and Junta de Andalucía, Granada, Spain, pp. 380–385.

Cortizo, M., Álvarez, J.M., Rodríguez, A., Fernández, B., Ordás, R.J., 2010. Cloning and characterization of a type-A response regulator differentially expressed during adventitious shoot formation in *Pinus pinea* L. J. Plant Physiol. 167, 1023–1026. https://doi.org/10.1016/J.JPLPH.2010.02.012.

Cortizo, M., Cuesta, C., Centeno, M.L., Rodríguez, A., Fernández, B., Ordás, R., 2009. Benzyladenine metabolism and temporal competence of *Pinus pinea* cotyledons to form buds in vitro. J. Plant Physiol. 166, 1069–1076. https://doi.org/10.1016/J.JPLPH.2008.12.013.

Court-Picon, M., Gadbin-Henry, C., Guibal, F., Roux, M., 2004. Dendrometry and morphometry of *Pinus pinea* L. in Lower Provence (France): adaptability and variability of provenances. For. Ecol. Manage. 194, 319–333. https://doi.org/10.1016/J.FORECO.2004.02.024.

Delard, C., Loewe, V., Río, R. Del, González, M., Gregori, G., Acevedo, M., Cartes, E., Balzarini, M., 2023. Effect of *Tuber borchii* inoculation on *Pinus pinea* seedling quality and initial growth in the field. New For. 55, 609–627. https://doi.org/10.1007/s11056-023-09992-8.

Domínguez Lerena, S., Carrasco, I., Herrero, N., Ocaña, L., Nicolás, J.L., Peñuelas, J.L., 2000a. Las características de los contenedores influyen en la supervivencia y crecimiento en campo de las plantas de *Pinus pinea* en campo, Actas del 1er Simposio del pino piñonero (*Pinus Pinea*). Sociedad Española de Ciencias Forestales-Junta de Castilla y León, Valladolid, Spain, pp. 203–209.

Domínguez Lerena, S., Oliet, J.A., Ruiz, P., Carrasco, I., Peñuelas, J.L., Serrada, R., 2000b. Influencia de la relación N-P-K en el desarrollo en vivero y campo de plantas de *Pinus pinea*. In: Actas del 1er Simposio del Pino Piñonero, pp. 195–202. Sociedad Española de Ciencias Forestales and Junta de Castilla y León, Valladolid, Spain.

Domínguez Lerena, S., Herrero-Sierra, N., Carrasco-Manzano, I., Ocaña Bueno, L., Peñuelas-Rubira, J.L., 1997. Ensayo de diferentes tipos de contenedores para *Quercus ilex, Pinus halepensis, P. pinaster* y *P. pinea*: resultados de vivero, Actas del II Congreso Forestal Español. In: Irati 97. Pamplona, Spain, pp. 189–194. Sociedad Española de Ciencias Forestales-Gobierno de Navarra.

Dominguez-Lerena, S., Herrero Sierra, N., Carrasco Manzano, I., Ocaña Bueno, L., Peñuelas Rubira, J.L., Mexal, J.G., 2006. Container characteristics influence *Pinus pinea* seedling development in the nursery and field. For. Ecol. Manage 221, 63–71. https://doi.org/10.1016/j.foreco.2005.08.031.

Escudero, A., Pérez-García, F., Luzuriaga, A.L., 2002. Effects of light, temperature and population variability on the germination of seven *Spanish pines*. Seed Sci. Res. 12, 261–271. https://doi.org/10.1079/SSR2002116.

Fernández, M., Martín, D., Masedo, F., Pardos, J.A., 2000a. Efecto del régimen de riego sobre la conductancia hidráulica en tallo hipocotilo de *Pinus pinea* L. In: Actas del 1er Simposio del Pino Piñonero, Valladolid, Spain. Sociedad Española de Ciencias Forestales-Junta de Castilla y León, pp. 177–181.

Fernández, M., Puertolas, J., Alonso, J., Rodríguez, M., Gil, L., Pardos, J.A., 2000b. Efecto de la fertilización durante el periodo de endurecimiento sobre las plantas de *Pinus pinea* L producidas en contenedor, Actas del 1er Simposio del Pino Piñonero, Valladolid, Spain. Sociedad Española de Ciencias Forestales-Junta de Castilla y León, pp. 183–190.

Ganatsas, P., Thanasis, G., 2010. *Pinus halepensis* invasion in *Pinus pinea* habitat in Strofylia forest (Site of NATURA 2000 network), southern Greece. J. Nat. Conserv. 18, 106–117. https://doi.org/10.1016/J.JNC.2009.04.006.

Ganatsas, P., Tsakaldimi, M., Thanos, C., 2008. Seed and cone diversity and seed germination of *Pinus pinea* in Strofylia site of the Natura 2000 network. Biodivers. Conserv. 17, 2427–2439. https://doi.org/10.1007/s10531-008-9390-8.

Ganatsas, P., Tsakaldimi, M.N., 2007. Effect of light conditions and salinity on germination behaviour and early growth of umbrella pine (*Pinus pinea* L.) seed. J. Hortic. Sci. Biotechnol. 82, 605–610. https://doi.org/10.1080/14620316.2007.11512280.

García de Castro, M.F., Martinez-Honduvilla, C.J., 1984. Ultrastructural changes in naturally aged *Pinus pinea* seeds. Physiol. Plant 62, 581–588. https://doi.org/10.1111/J.1399-3054.1984.TB02803.X.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal de la Comunidad Valenciana. Conselleria de Medi Ambient, Generalitat Valenciana, Alboraya, Valencia, Spain.

Gonçalves, A.C., Pommerening, A., 2012. Spatial dynamics of cone production in Mediterranean climates: a case study of *Pinus pinea* L. in Portugal. For Ecol. Manage. 266, 83–93. https://doi.org/10.1016/J.FORECO.2011.11.007.

Gordo Alonso, F.J., Álvarez Muñoz, D., Mutke, S., Gil, L., 2009. Selección y uso de materiales de base en el pino piñonero, Actas del V Congreso Forestal Español-Montes y Sociedad: Saber qué hacer. In: Ávila, Spain. Sociedad Española de Ciencias Forestales and Junta de Castilla y León.

Gradi, A., 1989. Raccolta di semi forestali e possibilità di meccanizzazione. Nel rispetto della sicurizza de glioperai. Economia montana Linea ecológica 21, 15–18.

Grossnickle, S.C., El-Kassaby, Y.A., 2016. Bareroot versus container stocktypes: a performance comparison. New For. 47, 1–51. https://doi.org/10.1007/s11056-015-9476-6.

Guàrdia, M., Teixidó, A., Sanchez-Bragado, R., Aletà, N., 2021. An agronomic approach to pine nut production by grafting stone pine on two rootstocks. Agriculture 2021 11, 1034. https://doi.org/10.3390/AGRICULTURE11111034.

Hammad, H.H., 2012. Impact of bio-composite on growth and chemical composition of three pine species seedlings as comparing with some growing media. J. Plant Product. Mansoura Univ. 3, 1005–1015.

Herrero Sierra, N., Nicolás Peragón, J.L., 2013. Anexo III Datos característicos de lotes de semillas. CNRGF El Serranillo. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Ministerio de agricultura, alimentación y medio ambiente., Madrid, Spain, pp. 734–739.

Honrubia, M., Torres, P., Díaz, G., Cano, A., 1992. Manual para micorrizar plantas en viveros forestales. In: Ministerio de Agricultura, Pesca y Alimentación. ICONA.

Honrubia, M., Carrillo, C., Peñuelas, J., Domínguez, S., Villar, P., Ocaña, L., 1997. Influencia de la fertirrigación en la micorrización controlada de *Pinus halepensis* en vivero, Actas del II Congreso Forestal Español. Irati 97. Pamplona, Spain, pp. 307–311. Sociedad Española de Ciencias Forestales-Gobierno de Navarra.

Kadri, N., Khettal, B., Aid, Y., Kherfellah, S., Sobhi, W., Barragan-Montero, V., 2015. Some physico-chemical characteristics of *pinus* (*Pinus halepensis* Mill., *Pinus pinea* L., *Pinus pinaster and Pinus canariensis*) seeds from North Algeria, their lipid profiles and volatile contents. Food Chem. 188, 184–192. https://doi.org/10.1016/J.FOODCHEM.2015.04.138.

Kildisheva, O.A., Aghai, M.M., Bouazza, K., Davis, A.S., 2017. Improving restoration success through research-driven initiatives: case studies targeting *Pinus pinea* reforestation stock development in Lebanon. Plant Ecol. 218, 39–53. https://doi.org/10.1007/s11258-016-0632-7.

Larchevêque, M., Ballini, C., Korboulewsky, N., Montès, N., 2006. The use of compost in afforestation of Mediterranean areas: effects on soil properties and young tree seedlings. Sci. Total Env. 369, 220–230. https://doi.org/10.1016/j.scitotenv.2006.04.017.

Loewe-Muñoz, V., del Río Millar, R., Delard Rodriguez, C., Balzarini, M., 2024a. Effects of fertilization on radial growth of *Pinus pinea* explored hourly using dendrometers. Ecol. Process. 13, 1–15. https://doi.org/10.1186/s13717-023-00479-0.

Loewe-Muñoz, V., del Rio, R., Delard, C., Balzarini, M., 2020. Irrigation and fertilization as tools to boost growth stability of stone pine (*Pinus pinea* L.) plantations. For Ecol. Manage. 463, 118017. https://doi.org/10.1016/J.FORECO.2020.118017.

Loewe-Muñoz, V., del Río, R., Delard, C., Balzarini, M., 2023. Effect of fertilization on *Pinus pinea* cone to seed and kernel yields. For Ecol. Manage. 545, 121249. https://doi.org/10.1016/J.FORECO.2023.121249.

Loewe-Muñoz, V., Delard, C., del Río, R., Balzarini, M., 2024b. Recommendations for increasing yield of the edible *Pinus pinea* L. pine nuts. PLoS One 19, e0300008. https://doi.org/10.1371/JOURNAL.PONE.0300008.

Loewe-Muñoz, V., Delard, C., del Río, R., Gregori, G., Balzarini, M., 2024c. Effects of *Tuber borchii* inoculation on *Pinus pinea* 3 years after establishment along a latitudinal gradient in the Southern Hemisphere. Agroforest. Syst. 98, 369–381. https://doi.org/10.1007/s10457-023-00915-2.

Lucas García, J.A., Domenech, J., Santamaría, C., Camacho, M., Daza, A., Mañero, F.J.G., 2004. Growth of forest plants (pine and holm-oak) inoculated with rhizobacteria: relationship with microbial community structure and biological activity of its rhizosphere. Env. Exp. Bot. 52. https://doi.org/10.1016/j.envexpbot.2004.02.003.

Montero, G., Ruiz-Peinado, R., Candela, J.A., Cañellas, I., Gutiérrez, M., Pavón, J., Alonso, A., Río, M. D., Bachiller, A., Calama, R., 2004. Silvicultura. In: Montero González, G., Candela Plaza, J.A., Rodríguez Navarro, A. (Eds.), El Pino Piñonero (*Pinus Pinea* L.) En Andalucía. Ecología, Distribución y Selvicultura. Consejería de Medio Ambiente. Junta de Andalucía, Sevilla, Spain, pp. 113–252.

Mutke, S., Díaz Balteiro, L., Gordo, J., 2000. Análisis comparativo de la rentabilidad comercial privada de plantaciones de *Pinus pinea* L. en tierras agrarias de la provincia de Valladolid. Investigación Agraria: Sistemas y Recur. Forest. 9, 269–304.

Mutke, S., Díaz, L., 2001. Plantaciones injertadas de *Pinus pinea*. Posible impacto de los programas de forestación de tierras agrarias. Actas del III Congreso Forestal Español-Granada, 2001. Tomo III. Sociedad Española de Ciencias Forestales-Juanta de Andalucía 186–193.

Mutke, S., Iglesias, S., Gil, L., 2007. Selección de clones de pino piñonero sobresalientes en la producción de piña. Investigación Agraria: Sistemas y Recursos Forestales 16, 39–51.

Navarro Cerrillo, R.M., Oliet, J., Contreras, O., 2001a. El uso de tubos protectores con cuatro especies forestales en Andalucía occidental: supervivencia y crecimiento. In: III Congreso Forestal Español Congreso.

Navarro Cerrillo, R.M., Gallegos Pérula, V., Alcántara Vara, E., 2001b. Deficiencias nutritivas en plantas de una savia de tres especies del género *Pinus* sp. en cultivo hidropónico. Off. Syst. 10, 43–58.

Navarro, R.M., Palacios, G., 2004. Efecto de la calidad de planta, el procedimiento de preparación y la fecha de plantación en la supervivencia de una repoblación de“ Pinus pinea” L. Cuad. Soc. Esp. Cienc. For. 17, 199–204.

Navarro Cerillo, R.M., Zapater, P., Sánchez Lancha, A., Arroyo Sauce, M., del Campo, A.D., 2009a. Comparación de sustratos para el cultivo de pino piñonero (*Pinus pinea* L.) y encina (*Quercus ilex* L.) en la Red de Viveros de Andalucía. In: Actas del V Congreso Forestal Español. Ávila, Spain. Sociedad Española de Ciencias Forestales-Junta de Castilla y León.

Navarro Cerrillo, R.M., Peman García, J., Del Campo García, A., Moreno Sánchez, J., Lara Gómez, M.A., Díaz Hernández, J.L., Pousa Salvador, F., Piñon Castillo, F., 2009b. Manual de especies para la forestación de tierras agrarias en Andalucía, Agricultura. Forestal. Junta de Andalucía. Consejería de Agricultura y Pesca, Sevilla, Spain.

Navarro Cerrillo, R.M., Sánchez Lancha, A., Arroyo Sauces, M., Plaza Arregui, L., Mallofret, C.E., Lara Gómez, M.A., Alejano Monge, R., 2012. *Pinus pinea* L. In: Pemán García, J., Navarro Cerrillo, R. M., Nicolás Peragón, J.L., Prada Sáez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio ambiente, Madrid, Spain, pp. 928–959.

Navarro-Cerrillo, R.M., Gálvez, C., 2001. In: Costa Pérez, J.C. (Ed.), Manual para la identificación y reproducción de semillas de especies autóctonas de Andalucía. Junta de Andalucía, Sevilla, España, pp. 292–296.

Nergiz, C., Dönmez, I., 2004. Chemical composition and nutritive value of *Pinus pinea* L. seeds. Food Chem. 86, 365–368. https://doi.org/10.1016/J.FOODCHEM.2003.09.009.

Oliet, J., Navarro Cerrillo, R.M., Contreras Atalaya, O., 2003. Evaluación de la aplicación de tubos y mejoradores en repoblaciones forestales. In: Dirección General de Gestión del Medio Natural, Junta de Andalucía, Córdoba, Spain.

Padilla, F.M., Pugnaire, F.I., 2007. Rooting depth and soil moisture control Mediterranean woody seedling survival during drought. Funct. Ecol. 21, 489–495.

Palacios, G., Navarro Cerrillo, R.M., 2001. Caracterización de la calidad de planta en vivero de siete procedencias de pino piñonero (*Pinus pinea* L.). Actas del III Congreso Forestal Español-Granada, Tomo III. Sociedad Española de Ciencias Forestales-Junta de Andalucía, pp. 854–860.

Parladé, J., Luque, J., Pera, J., Rincón, A., 2004. Field performance of *Pinus pinea* and *P. halepensis* seedlings inoculated with Rhizopogon spp. and outplanted in formerly arable land. Ann. For. Sci. 61, 507–514.

Parladé, J., Hortal, S., Pera, J., Galipienso, L., 2007. Quantitative detection of *Lactarius deliciosus* extraradical soil mycelium by real-time PCR and its application in the study of fungal persistence and interspecific competition. J. Biotechnol. 128. https://doi.org/10.1016/j.jbiotec.2006.09.010.

Pérez-Luna, A., Wehenkel, C., Prieto-Ruíz, J.Á., López-Upton, J., Solís-González, S., Chávez-Simental, J. A., Hernández-Díaz, J.C., 2020. Grafting in conifers: a review. Pak. J. Bot. 52, 1369–1372.

Picher Morelló, M., Ventimilla Antón, P., Campos Fuster, E., 2013. Anexo II Datos característicos de lotes de semillas. Banc de llavors forestals, Dirección General del Medio Natural, Generalitat Valenciana. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Ministerio de agricultura, alimentación y medio ambiente, Madrid, Spain, pp. 731–733.

Piqué, M., Coello, J., Ammari, Y., Aletà, N., Sghaier, T., Mutke, S., 2017. Grafted stone pine plantations for cone production: trials on *Pinus pinea* and *Pinus halepensis* rootstocks from Tunisia and Spain. Options Mediterraneennes, Options Méditerranéennes : Série A. Sémin. Méditerran. 122, 17–23.

Probanza, A., Lucas García, J.A., Ruiz Palomino, M., Ramos, B., Gutiérrez Mañero, F.J., 2002. *Pinus pinea* L. seedling growth and bacterial rhizosphere structure after inoculation with PGPR *Bacillus* (*B. licheniformis* CECT 5106 and *B. pumilus* CECT 5105). Appl. Soil Ecol. 20. https://doi.org/10.1016/S0929-1393(02)00007-0.

Puértolas, J., Sierra, R., Gil, L., Pardos, J.A., 2005. Comportamiento fisiológico en una plantación de *Pinus halepensis* y *Pinus pinea* en un antiguo terreno agrícola. In: Actas del IV Congreso Forestal Español. Zaragoza, Spain. Sociedad Española de Ciencias Forestales-Gobierno de Aragón.

Puértolas, J., Jacobs, D.F., Benito, L.F., Peñuelas, J.L., 2012. Cost-benefit analysis of different container capacities and fertilization regimes in *Pinus* stock-type production for forest restoration in dry Mediterranean areas. Ecol. Eng. 44, 210–215. https://doi.org/10.1016/j.ecoleng.2012.04.005.

Renaldi, F., Giachetti, E., Guerin, E., Bacci, S., Paoletti, E., Boddi, V., Vanni, P., 2003. Gravitational stress on germinating *Pinus pinea* seeds. C. R. Biol. 326, 553–564. https://doi.org/10.1016/S1631-0691(03)00151-3.

Ribeiro, D., Ribeiro, H., Louro, V., 2001. Produção Em Viveiros Florestais. Direcção-Geral de Desenvolvimento Rural, Lisboa.

Rincón, A., Álvarez, I.F., Parladé, J., Pera, J., 1997. Micorrización controlada de *Pinus pinea* en vivero. Actas del II Congreso Forestal Español. Irati 97. Tomo III. Sociedad Española de Ciencias Forestales-Gobierno Foral de Navarra 545–550 pp Pamplona, Spain.

Rincon, A., Alvarez, I.F., Pera, J., 2001. Inoculation of containerized *Pinus pinea* L. Seedlings with seven ectomycorrhizal fungi. Mycorrhiza 11. https://doi.org/10.1007/s005720100127.

Ruano, R., López, E., Martínez, A., Villaplana, R., Fos, M., Sanchís, E., 2001. Sustratos alternativos al empleo de la turba en el cultivo de brinzales de pino, Actas del III Congreso Forestal Español. Mesa 3. Granada, Spain. Sociedad Española de Ciencias Forestales- Junta de Andalucía, pp. 441–448.

Sánchez-Gómez, D., Cano, F.J., Cervera, M.T., Aranda, I., 2009. Variabilidad fenotípica en respuesta al estrés hídrico en una especie forestal genéticamente homogénea: Pinus pinea L., Actas del V Congreso Forestal Español. In: Ávila, Spain. Sociedad Española de Ciencias Forestales- Junta de Castilla y León.

Sánchez-Gómez, D., Velasco-Conde, T., Cano-Martín, F.J., Ángeles Guevara, M., Teresa Cervera, M., Aranda, I., 2011. Inter-clonal variation in functional traits in response to drought for a genetically homogeneous Mediterranean conifer. Env.. Exp. Bot. 70. https://doi.org/10.1016/j.envexpbot.2010.08.007.

Toca, A., Oliet, J.A., Villar-Salvador, P., Martínez Catalán, R.A., Jacobs, D.F., 2019. Ecologically distinct pine species show differential root development after outplanting in response to nursery nutrient cultivation. For. Ecol. Manage. 451, 117562. https://doi.org/10.1016/J.FORECO.2019.117562.

Villar-Salvador, P., Domínguez Lerena, S., Peñuelas Rubira, J.L., Carrasco, I., Herrero Sierra, N., Nicolás Peragón, J.L., Ocaña Bueno, L., 2000. Plantas grandes y mejor nutridas de *Pinus pinea* L. tienen mejor desarrollo en campo, Actas del 1er Simposio del Pino Piñonero (*Pinus pinea* L.). Valladolid, Spain. Sociedad Española de Ciencias Forestales-Junta de Castilla y León, pp. 219–227.

Villar-Salvador, P., Peñuelas, J.L., Jacobs, D.F., 2013. Nitrogen nutrition and drought hardening exert opposite effects on the stress tolerance of *Pinus pinea* L. seedlings. Tree Physiol. 33, 221–232. https://doi.org/10.1093/treephys/tps133.

Pinus sylvestris L.

Vladan Ivetić and Ljubica Mijatović
University of Belgrade, Faculty of Forestry, Belgrade, Serbia

Propagation

Seed propagation

The optimal period for *Pinus sylvestris* (Scots pine) cone harvesting is when seeds have reached full maturity but before deterioration begins, typically in late summer to early autumn, depending on local environmental conditions and site characteristics (Ivetić, 2023). Seeds collected in September show a germination rate of 95%–99.5% (Hurly et al., 1987). Cones must be harvested before natural seed dispersal, which is influenced by climatic factors, particularly by high temperatures that promote dispersal. For example, in central Sweden, peak seed fall occurs in early to mid-May, suggesting that seed collection should be completed before this period to prevent losses (Hannerz et al., 2002). Temperature and relative air humidity, and the presence of seed predators like squirrels, also influence the timing and method of harvesting.

P. sylvestris cones are usually harvested manually, a labor-intensive method that allows, however, careful selection of cones (Roth, 2000). It involves climbing trees or using mobile elevators and several tools and devices to reach and cut the cones (Ivetić, 2023).

Processing of *P. sylvestris* cones and seeds involves multiple steps to optimize seed extraction and quality (Ivetić, 2013): (1) Predrying of cones, (2) precleaning seeds of debris and large impurities, (3) drying cones, (4) extracting seed, (5) dewinging, (6) grading, (7) drying, and (8) storing seeds. Cone drying is a crucial step, with temperature and duration depending on initial moisture of the cones. The typical drying temperature for *P. sylvestris* cones varies between 45 and 55°C (Ivetić, 2023). In addition to the conventional method, new methods have been proposed such as microwave irradiation (Aniszewska et al., 2019; Aniszewska et al., 2020) and a two-stage extraction process that relies on interrupting extraction, sprinkling or soaking cones in water, and redrying (Aniszewska, 2013).

Seeds can be stored for up to 2 years at room temperature (Donald and Jacobs, 1990). However, for long-term preservation of valuable seeds, storage at low temperatures, such as −18°C (Nikolaeva et al., 2021), or in liquid nitrogen at ultralow temperatures (−182 to −196°C) is recommended (Ahuja, 1989; Pita et al., 1998; Nikolaeva et al., 2021). Seeds should be dried to a moisture content of 3%–7% before storage, and sealed in hermetic containers, such as aluminum foil bags, to protect against humidity and oxygen, which can deteriorate seeds. Regular monitoring of germination and infection levels is also recommended (van Treuren et al., 2013; Nikolaeva et al., 2021).

Table 11.44 Characteristics of seed lots of *Pinus sylvestris*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
1–2 (1.5)	90–100 (95)	75,000–240,000 (160,000)	0–99.5 (85)

Mean values in brackets.
Petrović (1952); Stilinović (1987); Wennström et al. (2002); Butorina et al. (2005); Hoffmann et al. (2007); Hilli et al. (2008); Peñuelas Rubira et al. (2012); Ivetić (2023); Mailybaeva (2024); Koba et al. (2024).

Although seeds of *P. sylvestris* are not dormant and do not need presowing treatment, cold stratification accelerates seed germination (Houšková et al., 2021). The quantitative characteristics of Pinus sylvestris seeds are presented in Table 11.44.

Vegetative propagation

Vegetative propagation of *P. sylvestris* is primarily achieved through somatic embryogenesis (SE), although existing protocols have not yet been developed for large-scale application (Aronen et al., 2009; Abrahamsson et al., 2018). The late phases of SE, including germination and acclimation, are critical, as a shortened germination protocol and controlled acclimation have shown improvements in the growth characteristics of somatic embryos (Reeves et al., 2023). Alternative propagation methods include organogenesis (Chávez and Feria, 2012), clonal propagation (Trontin et al., 2016) and grafting (Korczyk and Myszczyńska, 2012).

Seedling cultivation

Container plants

P. sylvestris seedlings in containers are usually grown as 1 + 0 (Fig. 11.32). The most effective seeding method involves automatic procedures by seed drills. To avoid empty containers or cells, one to three seeds per cell are sown, depending on seed quality.



Figure 11.32 *Pinus sylvestris* seedlings. Left: 1 + 0 container plants. Right: 2 + 0 bareroot plants. *(Vladan Ivetić.)*

Although seedlings are generally grown in open areas, in colder climates containers can remain in greenhouses during seed germination and early seedling emergence. Once the first needles develop, seedlings can be moved outdoors. If cultivation starts in an open area, seeds are sown in the second half of April. After watering and fungicide application to prevent damping-off, containers are moved outdoors and covered with agro textile to protect emerging seedlings from frost (Szabla and Pabian, 2009). The agro textile cover can be removed after the risk of late frost is over, usually in mid-May. In a greenhouse, seeds are usually sown from early March to late May, with the optimal period in the second half of April (Szabla and Pabian, 2009). Germination usually takes 6–8 days under an air temperature of 21–24°C and high relative humidity. Once the first needles develop, a weak frost will not damage seedlings, but if temperatures drop a few degrees below 0°C, seedlings should be protected by covering or spraying. Water spraying can be used to protect seedlings down to −8°C but should not be used if relative air humidity is below 20%. Spraying should start just before or right after reaching the freezing temperature and should end only when the ice starts melting, which is an indicator that there is no more danger of freezing injuries. The amount of applied water increases with decreasing temperature and increasing wind speed (Hansen et al., 1979).

P. sylvestris seedlings are mainly grown in rigid plastic containers of various cell volumes, from 30 to 50 cm^3 in the north (northern Sweden, Häggström et al., 2021) and from 100 to 200 cm^3 in the central and south of Europe (Szabla and Pabian, 2009; Peñuelas Rubira et al., 2012; Kolevska et al., 2020). Vertical slits in container cells improve root structure and stem straightness of outplanted seedlings (Rune, 2003).

During the rapid growth phase of *P. sylvestris*, Ingestad (1979) suggests maintaining optimal nutrient uptake with a nutrient solution containing 50 mg/L N and an element balance of NPKCaMg = 100:45:14:6:6. Optimal fertilization regimes include a constant supply of 4 g N/m/week to ensure nutrient surplus or variable regimes of 3.5 or 7 g N/m^2/week based on target growth rate (Troeng and Ackzell, 1988). These authors emphasize that low N-supply enhances root growth more than shoot growth, increasing root/shoot ratio, while higher N-supply has the opposite effect.

Irrigation at a growing media matric potential of −5 to −10 kPa balances water availability and aeration, promoting rapid seedling growth while keeping air porosity in the peat medium between 39% and 43%, which is considered optimal to slightly suboptimal (Heiskanen, 1995). A subirrigation regime with precise control can reduce water consumption by about 8% and improve shoot sturdiness and root development (Kormanek et al., 2023).

P. sylvestris seedlings can be cold hardened by shortening the photoperiod and adjusting the fertilization regime. Short day conditions with photoperiods between 6 and 12 h, combined with low temperatures during the night, are most effective for cold hardening (Aronsson, 1975). Ryyppö et al. (1998) recommends a two-stage hardening process: (1) a 6 h photoperiod with a gradual temperature decrease to 5°C over 4 weeks, followed by

(2) maintaining these conditions for an additional 4 weeks. Late-summer fertilization has been shown to enhance frost hardiness, with increased needle nitrogen, phosphorus, and potassium levels correlating with higher frost resistance (Rikala and Repo, 1997).

Mycorrhization of *P. sylvestris* container seedlings can be achieved by fungal inoculation or coinoculation with fungi and bacteria, for example, *Amanita rubescens* or *Hebeloma sinapizans* with fungi and ectomycorrhiza associated bacteria (EMAB), *Pseudomonas putida* or *Bacillus cereus* (Kozdrój et al., 2007). While mycorrhization provides potential benefits, its effectiveness varies depending on environmental conditions, fungal species, and additional treatments, such as soil preparation and fertilization (Klavina et al., 2017; Andreev et al., 2022; Bzdy et al., 2022; Mrak et al., 2024).

Fungi such as *Fusarium* spp., *Alternaria* spp., and *Botrytis cinerea* are prevalent in *P. sylvestris* nurseries, causing diseases like damping-off and needle cast (Lilja, 1986; Woodward et al., 2022). Notable pests that damage seedlings include pine weevils (Curculionidae) (Miezite et al., 2022) and pinewood nematode (*Bursaphelenchus xylophilus*), which causes pine wilt disease (da Silva et al., 2015).

Various storage methods, including cold storage and outdoor overwintering can be used. While cold storage at controlled temperatures (optimal at 2°C) generally supports better seedling recovery and growth, outdoor overwintering is less predictable and potentially more harmful for plants (Mattsson and Troeng, 1986). Cold-stored seedlings generally exhibit better growth performance postplanting, which is crucial for establishing robust forest stands (Mattsson, 1986).

After one growing season, seedlings typically reach an average height of 15–20 cm with a root collar diameter of 0.5 cm. Seedlings coming from May sowing or directly into cassettes on open surface are slightly smaller but are suitable to be used to plant even challenging sites.

Bareroot plants

Although container seedlings have gained most of the market share in Fennoscandia (and across the rest of Europe), bareroot seedlings often are still used, especially on fertile, vegetation-rich sites prone to pine weevil damage (Nilsson et al., 2010). The most common bareroot stock types of *P. sylvestris* are $2 + 0$, $3 + 0$ in southern regions (Ivetić, 2023) and $2 + 1$ plants in northern regions (Lilja et al., 1997). Bareroot seedling production generally involves the following steps: (1) preparing the soil and seedbed, (2) sowing, (3) culturing, (4) lifting for outplanting or transplanting; if transplants are produced, (5) transplanting in a growing area and (6) lifting for outplanting. Each operation varies depending on the nursery location, which strongly affects the environmental conditions, such as soil and water quality, and available equipment.

Soil preparation usually involves subsoiling and plowing. Subsoiling is recommended if soil is compacted below the plow layer or if water drainage is an issue. Plowing should

be performed in autumn after seedlings are lifted or when preparing new fields. During plowing, granular mineral fertilizer should be applied, and organic material, mainly manure, can be incorporated depending on soil characteristics. Phosphorus (P) and potassium (K) are the primary nutrients targeted with fertilization, with K applied in sulfate form to avoid chlorine's negative effects on conifers (Baule and Fricker, 1978). Aldhous (1972) recommends applying 55–110 kg/ha P, and 100–130 kg/ha K for seedbeds. Manure, applied at a rate of 10–40 T/ha depending on soil properties and origin, is commonly used to improve soil physical and chemical properties (Ivetić, 2023). If soil pH is too high, it can be lowered using ammonium-sulfate, sulfur, peat, humus, or crop rotation. After harrowing and rototilling, raised seedbeds should be formed. Seedbeds are typically 1–1.3 m wide, with the width adjusted to accommodate mechanized seedling cultivation.

Although seeds can be broadcast over seedbeds, drill sowing is preferable for mechanical cultivation and root culturing. *P. sylvestris* seeds should be sown in early spring, when soil has warmed enough and the risk of late frost is over, usually in April. Seeds do not need any pretreatment, although soaking seeds in water for 24 h prior to sowing increase germination rate and uniformity (Stilinović, 1987). Spacing between furrows in drill sowing is 15–20 cm, with a seeding rate of 2–3 g/m. Seeds are covered with 1.5–2.5 cm of loose soil, preferably with humus-rich soil and the seedbed rolled to press the soil around the seed (Petrović, 1952). Irrigation should begin immediately after sowing, with slow watering during germination to prevent crust formation. An air relative humidity of 95% and a soil moisture tension of 3 kPa significantly enhance germination success (Oleskog et al., 2000).

Cultivation usually includes shading, pest protection, irrigation, supplementary fertilization, weed control, root culturing, and transplanting. In the southern range of *P. sylvestris*, seedbeds with recently emerged seedlings should be shaded on sunny days and protected from birds until seedlings shed their seed coats. Pest protection measures are similar to those in container production. Sprinkler irrigation is commonly used, but recently drip and subirrigation systems are being used in some nurseries. If water pH > 6.8 or contains more than 150 mg/L $CaCO_3$, it should be acidified (Ivetić, 2023).

Supplementary fertilization mainly aims to supply Aldhous (1972) recommends applying 50–100 kg/ha N and 0–200 kg/ha K after sowing or transplanting divided into several smaller applications. Liquid fertilizers, such as urea, can be used for rapid N supplementation, with satisfactory results in conifers (Baule and Fricker, 1978). Weed control should be mechanical, or using herbicides. Root culturing includes undercutting, wrenching, and vertical or lateral, box or bench pruning (Grossnickle and Ivetić, 2022). Transplanting is needed for producing $2 + 1$ stock types. Only well-developed seedlings should be transplanted, with spacings 15–20 cm between rows and 8–10 cm within rows (Table 11.45).

Table 11.45 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 (container) and 2 + 0 (bareroot) seedlings of *Pinus sylvestris*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	2.5–30.5	25	7.4–49	15–43
Root collar diameter (cm)	0.2–0.55	>0.3	0.26–1.8	>0.4
Shoot dry weight (g)	3.52–5.83	>3.5	0.3–23.2	>5
Root dry weight (g)	1.42–3	>2	0.1–9.1	>2.5
Plant dry weight (g)	4.94–8.83	>5.5	0.4–32.3	>7.5
Shoot/root dry weight ratio	1.3–2	1.5–2.5	1.55–10.1	1.5–2.5
Shoot [N] (mg/g)	9–14.8	14–18.3	13.6–15.4	18–30
Shoot [P] (mg/g)	0.8–2.2	1.3–1.9	1.77–1.96	1.5–4
Shoot [K] (mg/g)	0.9–5.1	4.4–7	7.83–8.36	6–16
Root [N] (mg/g)	9.6–21.8	>17		
Root [P] (mg/g)	1–3.3	>1.5		
Root [K] (mg/g)	3.9–11.1	>5		

Aldhous (1972); Baule and Fricker (1978); Aldhous (1994); Jalkanen and Rikala (1995); Morgan (1999); Rikala (2000); Öner and Eren (2008); Zhang and George (2009); Mellert and Göttlein (2012); Memisoglu and Tilki (2014); Ivetić et al. (2017); Kolevska et al. (2020); Nosnikau et al. (2021); Pająk et al. (2022; 2022a).

Field establishment

P. sylvestris is a pioneer species with low shade tolerance, rapid initial growth, and early culmination of mean annual increment. It can grow in a wide range of soil conditions, including poor sites. Extreme weather conditions during the end of the winter are a significant cause of mortality in its northern range (Nilsson et al., 2010).

P. sylvestris can be established by both direct sowing and seedling planting. Spring sowing shows better results than summer sowing (de Chantal et al., 2004). Microsite preparation for seeding by 2 cm deep pyramidal indentations in the mineral soil, significantly improves emergence (Winsa and Bergsten, 1994). Direct sowing can result in germination rate of 26%–88% and establishment rates of 0%–45% (see Grossnickle and Ivetić, 2017).

P. sylvestris seedlings are usually planted during short windows in spring and autumn. Mechanical site preparation is recommended for successful establishment (Hjelm et al., 2019; Sikström et al., 2020) and includes bedding, plowing in furrows, and trenching (Węgiel et al., 2023), and use of double moldboard forest plows, active plowing, and forest milling (the machine used loosens the soil while grounds plant material). This latter method resulted in the best method for planting while active plowing was the optimal procedure for direct seeding (Aleksandrowicz-Trzcińska et al., 2017).

Protecting seedlings from pests like Curculionidae (weevils) is essential. While chemical pesticides are effective, alternative methods such as using wax and sand coatings have been explored, though they may incur higher costs (Miezite et al., 2022). In fact, sand coating is currently performed in Sweden before seedlings are stored or taken to the field.

Shoot browsing by moose (*Alces alces*) and other ungulates in young stands can cause severe regional economic losses (Nilsson, 2020). However, *P. sylvestris* shows strong recover ability after browsing, with significant family differences in growth traits. This suggests that selecting for herbivory tolerance traits could be a viable management strategy to mitigate browsing damage (O'Reilly-Wapstra et al., 2014). A recent study demonstrated that using *P. abies* bark extract as a repellent significantly reduced moose browsing on *P. sylvestris* (Lindmark et al., 2020).

Herbicides are used on vegetation-rich sites where permitted by local laws and certification rules. However, on most sites across northern Europe, effective vegetation control can be achieved through scarification (Nilsson et al., 2010).

Chapter References

Abrahamsson, M., Clapham, D.E., Arnold, S. von, 2018. Somatic embryogenesis in Scots pine (*Pinus sylvestris* L.). In: Jain, S., Gupta, P. (Eds.), Step Wise Protocols for Somatic Embryogenesis of Important Woody Plants. Forestry Sciences, vol 84. Springer, Cham. https://doi.org/10.1007/978-3-319-89483-6_9.

Ahuja, M.R., 1989. Storage of forest tree germplasm at sub-zero temperatures. In: Dhawan, V. (Ed.), Applications of Biotechnology in Forestry and Horticulture. Springer, Boston, MA. https://doi.org/10.1007/978-1-4684-1321-2_16.

Aldhous, J.R., 1994. Nursery policy and planning. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Bulletin 111 of the Forestry Commission. HMSO, pp. 104–111.

Aldhous, J.R., 1972. Nursery practice. In: Forestry Commission Bulletin No. 43, London, pp. 11–40.

Aleksandrowicz-Trzcińska, M., Drozdowski, S., Wołczyk, Z., Bielak, K., Żybura, H., 2017. Effects of reforestation and site preparation methods on early growth and survival of Scots pine (*Pinus sylvestris* L.) in south-eastern Poland. Forests 8 (11), 421. https://doi.org/10.3390/f8110421.

Andreev, E.M., Stetsenko, S.K., Hurshkainen, T.V., Terekhov, G.G., Kutchin, A.V., 2022. The mycorrhization of the root system of Scots pine with different granulometric composition of the soil under the influence of bio-stimulants in the Middle Urals. In: AIP Conference Proceedings 2390. https://doi.org/10.1063/5.0069142.

Aniszewska, M., 2013. Changes in humidity and temperature inside the pine cones (*Pinus sylvestris* L.) in two stages seed extraction. Leśne Prace Badawcze 74, 205–214. https://doi.org/10.2478/FRP-2013-0020. Forest Research Papers.

Aniszewska, M., Gendek, A., Tulska, E., Pęska, P., Moskalik, T., 2019. Influence of the duration of microwave irradiation of Scots pine (*Pinus sylvestris* L.) cones on the quality of harvested seeds. Forests 10, 1108. https://doi.org/10.3390/F10121108.

Aniszewska, M., Zychowicz, W., Gendek, A., 2020. The effectiveness of short-term microwave irradiation on the process of seed extraction from Scots pine cones (*Pinus sylvestris* L.). iFor. Biogeosci. For. 13, 73–79. https://doi.org/10.3832/IFOR3089-012.

Aronen, T., Pehkonen, T., Ryynänen, L., 2009. Enhancement of somatic embryogenesis from immature zygotic embryos of *Pinus sylvestris*. Scand. J. For. Res. 24, 372–383. https://doi.org/10.1080/02827580903228862.

Aronsson, A., 1975. Influence of photo- and thermoperiod on the initial stages of frost hardening and dehardening of phytotron-grown seedlings of Scots pine (*Pinus silvestris* L.) and Norway spruce (*Picea abies* (L.) Karst.). Stud. For. Suec. 128, 1–20. https://res.slu.se/id/publ/125469.

Baule, H., Fricker, C., 1978. Đubrenje Šumskog Drveća, Jugoslovenski Poljoprivredno-Šumarski Centar – Služba Šumske Proizvodnje, Beograd, Str, pp. 130–140.

Butorina, A.K., Cherkashina, O.N., Chernodubov, A.I., Avdeev, I.A., 2005. Variability of the cytological parameters of *Pinus sylvestris* L. seeds from the unique Hrenovskoy pine forest. Russ. J. Genet. 41, 630–634. https://doi.org/10.1007/S11177-005-0137-2.

Bzdy, R.M., Sikora, K.O., Studnicki, M., Aleksandrowicz-Trzcińska, M., 2022. Communities of mycorrhizal fungi among seedlings of Scots pine (*Pinus sylvestris* L.) growing on a clearcut in microsites generated by different site-preparation methods. Forests 13 (2). https://doi.org/10.3390/f13020353, 353–353.

Chávez, M., Feria, M. de, 2012. Aspectos básicos de la propagación in vitro del género *Pinus* por organogénesis. Biotecnol. Veg. 12, 131–142.

da Silva, V.I., Barbosa, P., Mota, M., Ascensão, L., 2015. Histological changes in stems of *Pinus sylvestris* seedlings infected with a virulent isolate of the pinewood nematode *Bursaphelenchus xylophilus*. Microsc. Microanal. 21, 97–98. https://doi.org/10.1017/S1431927615014294.

de Chantal, M., Leinonen, K., Ilvesniemi, H., Westman, C.J., 2004. Effects of site preparation on soil properties and on morphology of *Pinus sylvestris* and *Picea abies* seedlings sown at different dates. N. For. 27, 159–173. https://doi.org/10.1023/A:1025042632491.

Donald, D.G.M., Jacobs, C.B., 1990. The effect of storage time, temperature and container on the viability of the seed of four pine species. S. Afr. For. J. 154, 41–46. https://doi.org/10.1080/00382167.1990.9629050.

Grossnickle, S.C., Ivetić, V., 2022. Root system development and field establishment: effect of seedling quality. N. For. 53, 1021–1067. https://doi.org/10.1007/s11056-022-09916-y.

Häggström, B., Domevscik, M., Öhlund, J., Nordin, A., 2021. Survival and growth of Scots pine (*Pinus sylvestris)* seedlings in north Sweden: effects of planting position and arginine phosphate addition. Scand. J. For. Res. 36 (6), 423–433. https://doi.org/10.1080/02827581.2021.1957999.

Hannerz, M., Almqvist, C., Hörnfeldt, R., 2002. Timing of seed dispersal in *Pinus sylvestris* stands in central Sweden. Silva Fenn. 36, 518. https://doi.org/10.14214/SF.518.

Hansen, V.E., Israelsen, O.W., Stringham, G.E., 1979. Irrigation Principles and Practices. John Wiley and Sons, New York, USA, 417 p.

Heiskanen, J., 1995. Irrigation regime affects water and aeration conditions in peat growth medium and the growth of containerized Scots pine seedlings. N. For. 9, 181–195. https://doi.org/10.1007/BF00035486.

Hilli, A., Hokkanen, T., Hyvönen, J., Sutinen, M.L., 2008. Long-term variation in Scots pine seed crop size and quality in northern Finland. Scand. J. For. Res. 23 (5), 395–403. https://doi.org/10.1080/02827580802334217.

Hjelm, K., Nilsson, U., Johansson, U., Nordin, P., 2019. Effects of mechanical site preparation and slash removal on long-term productivity of conifer plantations in Sweden. Can. J. For. Res. 49, 1311–1319. https://doi.org/10.1139/cjfr-2019-0081.

Hoffmann, J., Chvalova, K., Palatova, E., 2007. Lesne semenarstvo na Slovensku. Itgamma, s.r.o Sliač 195. ISBN: 978-80-969717-0-1.

Houšková, K., Klepárník, J., Mauer, O., 2021. How to accelerate the germination of Scots pine and Norway spruce seeds? J. For. Sci. 67, 134–142.

Hurly, T.A., Robertson, R.J., Yeatman, C.W., 1987. Maturity and viability of seed from squirrel-cut pine cones. For. Chron. 63, 268–271. https://doi.org/10.5558/TFC63268-4.

Ingestad, T., 1979. Mineral nutrient requirements of *Pinus silvestris* and *Picea abies* seedlings. Physiol. Plant. 45, 373–380. https://doi.org/10.1111/j.1399-3054.1979.tb02599.x.

Ivetić, V., 2013. Praktikum iz Semenarstva rasadničarstva i pošumljavanja. Univerzitet u Beo¬gradu - Šumarski fakultet 213. ISBN: 978-86-7299-211-3.

Ivetić, V., 2023. Forest Reproductive Material – Biology and Technology of Production of Forest Trees Seed and Seedlings. University of Belgrade – Faculty of Forestry, Belgrade. 304 pg.

Ivetić, V., Maksimović, Z., Kerkez, I., Devetaković, J.R., 2017. Seedling quality in Serbia – results from a three-year survey. REFORESTA 4, 27–53. https://doi.org/10.21750/REFOR.4.04.43.

Jalkanen, A., Rikala, R., 1995. Foliar nutrient composition in bareroot *Pinus sylvestris* nursery crops. New Forest 10, 225–237. https://doi.org/10.1007/BF00027925.

Klavina, D., Muižnieks, I., Baumanis, I., Jansons, J., Gaitnieks, T., Menkis, A., 2017. Survival, growth and mycorrhization of containerised *Pinus sylvestris* and *Picea abies* seedlings of different provenances outplanted in a forest clear-cut. Proc. Latv. Acad. Sci. Sect. B Nat. Exact Appl. Sci. 71 (4), 293–297. https://doi.org/10.1515/PROLAS-2017-0049.

Koba, V.P., Krest'yanishin, I.A., Korenkova, O., 2024. Seed production and seed quality of *Pinus sylvestris* L. in forest plantations on the Crimean Yaylas. Russ. Forest. J. 4, 68–80. https://doi.org/10.37482/0536-1036-2024-4-68-80.

Kolevska, D.D., Dimitrova, A., Cokoski, K., Basova, M., 2020. Growth and quality of *Pinus nigra* (Arn.), *Pinus sylvestris* (L.) and *Pinus pinaster* (Aiton) seedlings in two container types. Reforesta 9, 21–36. https://doi.org/10.21750/REFOR.9.04.78.

Korczyk, A., Myszczyńska, I., 2012. A clone archive of old Scots pine *(Pinus sylvestris)* trees in the Augustowska Forest. For. Res. Pap. 1, 33–44. https://doi.org/10.2478/V10111-012-0004-6.

Kormanek, M., Małek, S., Banach, J., Durło, G., 2023. Effect of changing substrate density and water application method on substrate physical properties and container-grown seedling growth. Forests 14 (7), 1490. https://doi.org/10.3390/f14071490.

Kozdrój, J., Piotrowska-Seget, Z., Krupa, P., 2007. Mycorrhizal fungi and ectomycorrhiza associated bacteria isolated from an industrial desert soil protect pine seedlings against Cd(II) impact. Ecotoxicology 16, 449–456. https://doi.org/10.1007/S10646-007-0149-X.

Lilja, S., 1986. Disease and pest problems on *Pinus sylvestris* nurseries in Finland. EPPO Bull. 16 (3), 561–564. https://doi.org/10.1111/J.1365-2338.1986.TB00323.X.

Lilja, A., Lilja, S., Kurkela, T., Rikala, R., 1997. Nursery practices and management of fungal diseases in forest nurseries in Finland. A review. Silva Fenn. 31 (1), 79–100.

Lindmark, M., Sunnerheim, K., Jonsson, B.G., 2020. Natural browsing repellent to protect Scots pine *Pinus sylvestris* from European moose *Alces alces*. For. Ecol. Manag. 14, 118347. https://doi.org/10.1016/J.FORECO.2020.118347.

Mailybaeva, L.N., 2024. Comparative characteristics of cones and seeds of *Pinus sylvestris* L. from various biotopes of Northern Kazakhstan//Forests of Russia and economy in them, pp. 37–46, 1(88).

Mattsson, A., 1986. Planting site storage: effects on survival and growth of overwinter-stored Scots pine (*Pinus sylvestris*) containerized seedlings. Can. J. For. Res. 16 (1), 84–89. https://doi.org/10.1139/X86-014.

Mattsson, A., Troeng, E., 1986. Effects of different overwinter storage regimes on shoot growth and net photosynthetic capacity in *Pinus sylvestris* seedlings. Scand. J. For. Res. 1, 75–84. https://doi.org/10.1080/02827588609382402.

Mellert, K.H., Göttlein, A., 2012. Comparison of new foliar nutrient thresholds derived from van den Burg's literature compilation with established central European references. Eur. J. For. Res. 131, 1461–1472. https://doi.org/10.1007/s10342-012-0615-8.

Memisoglu, T., Tilki, F., 2014. Growth of Scots pine and silver birch seedlings on different nursery container media. Not. Bot. Horti Agrobot. Cluj-Napoca 42, 565–572. https://doi.org/10.15835/nbha4229551.

Miezite, O., Jansone, B., Brizga, D., Sisenis, L., 2022. Protection of *Pinus sylvestris* stands from curculionidae damage. In: Proceedings of 22nd International Multidisciplinary Scientific GeoConference SGEM 2022, pp. 405–414. https://doi.org/10.5593/sgem2022v/3.2/s14.47, 22.

Morgan, J.L., 1999. Forest Tree Seedlings - best practice in supply treatment and planting. For. Commun. Bull. 121, 43pp.

Mrak, T., Grebenc, T., Friedrich, S., Münzenberger, B., 2024. Description, identification, and growth of *Tuber borchii* Vittad. mycorrhized *Pinus sylvestris* L. seedlings on different lime contents. Mycorrhiza 34, 85–94. https://doi.org/10.1007/s00572-023-01135-3.

Nikolaeva, M.A., Varentsova, E.Y., Safina, G.F., 2021. The impact of temperature patterns during storage of Scots pine and Norway spruce seeds on their germination and fungal infection rates. Proceed. Appl.

Bot. Genet. Breed. 182 (1), 157–167. https://doi.org/10.30901/227-8834-2021-1-157-167 (In Russ.).
Nilsson, O., 2020. Establishment and growth of Scots pine and Norway spruce: a comparison between species, Swedish University of Agricultural Sciences. PhD THESIS Acta Universit. Agricult. Suec. 71. https://pub.epsilon.slu.se/18698/1/Oscar%20Nilsson.pdf.
Nilsson, U., Luoranen, J., Kolström, T., Örlander, G., Puttonen, P., 2010. Reforestation with planting in northern Europe. Scand. J. For. Res. 25 (4), 283–294. https://doi.org/10.1080/02827581.2010.498384.
Nosnikau, V., Kimeichuk, I., Rabko, S., Kaidyk, O., Khryk, V., 2021. Growth and development of seedlings of *Scots pine* and European spruce container seedlings using various materials to neutralise the substrate. Sci. Horiz. 24 (4), 54–62. https://doi.org/10.48077/scihor.24(4).2021.54-62.
O'Reilly-Wapstra, J.M., Moore, B.D., Brewer, M.J., Beaton, J.R., Sim, D.A., Wiggins, N.L., Iason, G.R., 2014. *Pinus sylvestris* sapling growth and recovery from mammalian browsing. For. Ecol. Manag. 325, 18–25. https://doi.org/10.1016/j.foreco.2014.03.038.
Oleskog, G., Grip, H., Bergsten, U., Sahlén, K., 2000. Seedling emergence of *Pinus sylvestris* in characterized seedbed substrates under different moisture conditions. Can. J. For. Res. 11, 1766–1777. https://doi.org/10.1139/x00-111.
Öner, N., Eren, F., 2008. The comparisons between root collar diameter and height growth of black Pine (*Pinus nigra* Arnold.) and Scots pine (*Pinus sylvestris* L.) seedlings in Bolu Forest Nursery. J. Appl. Biol. Sci. 2 (1), 07–12.
Pająk, K., Małek, S., Kormanek, M., Jasik, M., 2022. The effect of peat substrate compaction on the macronutrient content of Scots pine *Pinus sylvestris* L. container seedlings. Sylwan 166, 211–223. https://doi.org/10.26202/sylwan.2022023.
Pająk, K., Małek, S., Kormanek, M., Banach, J., 2022a. Effect of peat substrate compaction on growth parameters and root system morphology of Scots pine *Pinus sylvestris* L. seedlings. Sylwan 166, 537–550. https://doi.org/10.26202/sylwan.2022062.
Peñuelas Rubira, J.L., Nicolás Peragón, J.L., Benito Matías, L., 2012. Pinus sylvestris. In: Pemán García, J., Navarro Cerrillo, R.M., Nicolás Peragón, J.L., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de semillas y plantas forestales, vol I. Organismo Autónomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, Spain, pp. 632–646.
Petrović, D., 1952. Rad u šumskim rasadnicima. In: Izdanje Glavne Uprave Za Šumarstvo NRS.
Pita, J.M., Sanz, V., Escudero, A., 1998. Seed cryopreservation of seven Spanish native pine species. Silvae Genet. 47, 220–223.
Reeves, C., Tikkinen, M., Aronen, T., Krajňáková, J., 2023. Application of cold storage and short in vitro germination for somatic embryos of *Pinus radiata* and P. *sylvestris*. Plants 12, 2095. https://doi.org/10.3390/plants12112095.
Rikala, R., 2000. Production and quality requirements of forest tree seedlings in Finland. Tree Planters' Notes U.S. Dep. Agric. For. Serv. 49 (3), 56–60.
Rikala, R., Repo, T., 1997. The effect of late summer fertilization on the frost hardening of second-year Scots pine seedlings. N. For. 14, 33–44. https://doi.org/10.1023/A:1006505919556.
Roth, V., 2000. Contribution to the knowledge of seed processing of *Scots pine* (*Pinus sylvestris* L.). Radovi Sumarski Inst. Jastreb. 35 (2), 5–16.
Rune, G., 2003. Slits in container wall improve root structure and stem straightness of outplanted Scots pine seedlings. Silva Fenn. 37 (3), 333–342.
Ryyppö, A., Repo, T., Vapaavuori, E., 1998. Development of freezing tolerance in roots and shoots of Scots pine seedlings at nonfreezing temperatures. Canad. J. Forest 28 (4), 557–565. https://doi.org/10.1139/x98-022.
Sikstrom, € U., Hjelm, K., Holt Hanssen, K., Saksa, T., Wallertz, K., 2020. Influence of mechanical site preparation on regeneration success of planted conifers in clearcuts in Fennoscandia – a review. Silva Fenn 54 (2), 10172. https://doi.org/10.14214/sf.10172.
Stilinović, S., 1987. Proizvodnja Sadnog Materijala Šumskog I Ukrasnog Drveća I Žbunja. Univerzitet U Beogradu. Šumarski fakultet, Beograd, Serbia.

Szabla, K., Pabian, R., 2009. Szkółkarstwo kontenerowe - nowe technologie i techniki w szkółkarstwie leśnym. Centr. Informac. Lasów Pañstwow.

Troeng, E., Ackzell, L., 1988. Growth regulation of *Scots pine* seedlings with different fertilizer compositions and regimes. N. For. https://doi.org/10.1007/BF00027763.

Trontin, J.F., Aronen, T., Hargreaves, C., Montalban, I.A., Moncalean, P., Reeves, C., Quoniou, S., Lelu-Walter, M.A., Klimaszewska, K., 2016. International Effort to Induce Somatic Embryogenesis in Adult Pine Trees. Vegetative Propagation of Forest Trees. National Institute of Forest Science Institute (NIFoS) hal-02800018.

van Treuren, R., de Groot, E.C., van Hintum, T.J.L., 2013. Preservation of seed viability during 25 years of storage under standard genebank conditions. Genet. Resour. Crop Evol. 60, 1407–1421. https://doi.org/10.1007/S10722-012-9929-0.

Węgiel, A., Jakubowski, J., Molińska-Glura, M., Krzysztof, P., Węgiel, J., Gornowicz, R., 2023. Effect of logging residue removal and mechanical site preparation on productivity of the subsequent Scots pine (*Pinus sylvestris* L.) stands. Ann. For. Sci. 80 (5). https://doi.org/10.1186/s13595-023-01175-x.

Wennström, U., Bergsten, U., Nilsson, J.E., 2002. Effects of seed weight and seed type on early seedling growth of *Pinus sylvestris* under harsh and optimal conditions. Scand. J. For. Res. 17 (2), 118–130. https://doi.org/10.1080/028275802753626764.

Winsa, H., Bergsten, U., 1994. Direct seeding of *Pinus sylvestris* using microsite preparation and invigorated seed lots of different quality: 2-year results. Canadian J. For. Res. 24, 77–86. https://doi.org/10.1139/x94-012.

Woodward, S., Flores-Pacheco, J.A., Jordán Muñoz-Adalia, E., Martínez-Álvarez, P., Martín-García, J., Diez, J.J., 2022. Susceptibility of germinating seedlings of European and Eurasian populations of *Pinus sylvestris* to damping-off caused by *Fusarium circinatum*. For. Pathol. 52 (4), e12749. https://doi.org/10.1111/efp.12749.

Zhang, Jl., George, E., 2009. Rhizosphere effects on ion concentrations near different root zones of Norway spruce (Picea abies (L.) Karst.) and root types of Douglas-fir (Pseudotsuga menziesii L.) seedlings. Plant Soil 322, 209–218. https://doi.org/10.1007/s11104-009-9909-0.

Platanus orientalis L.

Petar Zhelev[1], Ina Aneva[2] and Nasko Iliev[1]
[1]University of Forestry, Sofia, Bulgaria; [2]Department of Plant and Fungal Diversity and Resources, Institute of Biodiversity and Ecosystem Research, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

Fruit harvest of *Platanus orientalis* (oriental plane) is done manually from October to April (Delkov, 1977), preferably in dry weather to minimize fungal infections. The spheroidal infructescence (capitulum) can be collected directly by hand, when necessary, with ladders. Seeds are extracted manually by threshing or with wooden or rubber hammers. Infructescences harvested in late winter or early spring decompose easily.

Each capitulum contains 600 to 800 seeds, which are small, narrowly conical, up to 8 mm long, with a basal cluster of short hairs, and a beaked apex. Seeds can be stored in dry conditions, ideally below 15°C, although storage at room temperature is possible. Seeds can retain their germination capacity for up to 3 years if stored in refrigeration (0–4°C) and up to 10 years if stored at – 18°C (Forest Seed Control Station, 2023). The average characteristics of seed lots are summarized in Table 11.46.

Germination varies significantly among individuals in natural populations, with low germination often due to a high proportion of empty seeds. No pretreatment is necessary, although some studies report that soaking seeds in running water for several hours can slightly increase germination (Delkov, 1975). Sowing in nurseries is recommended in rows at a depth of 1 cm, covered with sand. Most germination (>70%) occurs during the first 5 days.

Vegetative propagation

This species can be propagated using green (herbaceous) (Zencirkiran and Erken, 2012) and lignified (red-to-brown colored) (Delkov, 1979; Panetsos et al., 1994) cuttings, with the latter being preferred due to higher rooting capacity than green cuttings (Delkov, 1979). Rooting in the greenhouse or in hot beds is promoted when the growing media temperature ranges between 10 and 15°C.

Table 11.46 Characteristics of seed lots of *Platanus orientalis*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
50–65 (60)	90–98 (95)	200,000–300,000 (230,000)	20–80 (45)

Mean values in brackets.
Delkov (1977); Piotto et al. (2001); Milev et al. (2004); Suliman and Ahmed (2014); Forest Seed Control Station (2023)

Shoot cutting must be 20–30 cm long, preferably with three nodes. Like in most tree species, the basal part of the cutting must be beveled to increase rooting surface. Soaking cuttings for 24 h can increase rooting. Rooting media are usually sand or sandy soil and the use of growth regulators is generally unnecessary (Zencirkiran and Erken, 2012). Rooting success varies by cutting collection time and genotype, ranging between 30% and 50% (Panetsos et al., 1994; Arene et al., 2000; Sajid et al., 2012; Rezaei et al., 2023) but reaching up to 80% in some cases (Ciafi et al., 2018).

Successful in vitro propagation is feasible using node and internode explants (Guoqiang et al., 2003; Showkat et al., 2017; Ciafi et al., 2018).

Seedling cultivation

Container plants

Container seedling production for forest plantings is still in its early stages, and available information is scarce. Cultivation can start between February and April, with the first phase conducted in a greenhouse if the risk of frost damage outdoors is high. Automatic seed filling and seeding line are possible. Due to their small size, seeds should be sown at a depth of ~1 cm in sand, or in a mixture of sand and perlite (3:1). Under greenhouse conditions, seedlings emerge around 15–20 days after seeding. After emergence, only one seedling must be left in each container cell. While cultivation can be done in 200 mL containers, we recommend using 300 mL containers at low cultivation density since *P. orientalis* is a fast-growing species with big leaves that may limit keeping a homogenous irrigation distribution in the crop. Table 11.47 summarizes some morphological and physiological characteristics of 1 + 0 seedlings.

Bareroot plants

Cultivation can start between February and April, depending on the occurrence of late frosts. Soil and seedbed preparation follow standard bareroot nursery practices. Seeds can

Table 11.47 Morphological attributes observed in 1 + 0 high quality, container and bareroot seedlings of *Platanus orientalis* as well as recommended values of high quality plants.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	23–50	>40	30–60	>40
Root collar diameter (cm)	–	–	0.3–1.5	>0.5
Shoot dry weight (g)	–	–	0.2–5	>2
Root dry weight (g)	–	–	0.1–5.2	>2

Suliman and Ahmed (2014).

be sown manually in 1-cm-deep rows, spaced 20 cm between them at a rate of about 60 seeds/m. The seeds should be covered with a 1 cm layer of sand (Milev et al., 2004). For up to 1.5 months after germination, seedlings are highly sensitive to direct sunlight and may require partial shading with a 30% reduction in the irradiation. Irrigation is recommended 1–2 times a week as needed. The species is tolerant of pests and diseases and no special protective measures are necessary. Hardening is not required, though early autumn frosts may damage non-lignified shoot tips. The primary quality indicator is seedling height, which should be at least 40 cm. Seedlings can be stored outdoors in the nursery.

Field establishment

Drought stress due to insufficient soil humidity during the first 2–3 years is the main environmental limitation for seedling establishment after planting. *P. orientalis* can grow on poor and shallow soils, including on limestone, but water availability is a crucial factor for survival and growth. To improve survival, planting in Mediterranean and sub-Mediterranean regions should occur when soil moisture levels are adequate.

There are two suitable periods of planting, spring and autumn. The most appropriate period in spring is in February and not later than the end of March, depending on site conditions. In autumn, the best planting time is November until mid-December, after or during fall rain events. Autumn planting is recommended because the seedlings will have had enough time to acclimate to the slowly changing environmental conditions. We recommend using $1 + 0$ seedlings for plantings.

Although the species tolerates shallow soils, deeper soils support better growth. The suitable planting sites for *P. orientalis* are riverine habitats with sandy soil. Therefore, no specific soil preparation is recommended in these habitats. The seedlings can be planted in holes done by mechanical augers. Soil fertility and nutrient availability is not of primary concern, but irrigation is the most crucial measure and mobile water tanks can facilitate watering in accessible sites. Hydrogel applied into planting holes can facilitate seedling establishment and survival on dry sites.

Herbs can affect the seedling performance during the first year, and therefore, if possible, herbs within 0.5 m around seedlings must be removed either by manual mowing or by digging.

Chapter References

Arene, L., Cadic, A., Djulbic, M., Gros, A., Renoux, A., 2000. Multiplication du platane par bouturage hivernal sur couche et microbouturage *in vitro*. In: Verger, M. (Ed.), Multiplication Végétative des Ligneux Forestiers, Fruitiers et Ornementaux. CIRAD-INRA Editions, Orléans, France, pp. 30–37 (coordinateur).

Ciafi, M., Alicandri, E., Vettraino, A.M., Paolacci, A.R., Tamantini, M., Tomao, A., Agrimi, M., Kuzminsky, E., 2018. Conservation of veteran trees within historical gardens (COVE): a case study applied to *Platanus orientalis* L. in central Italy. Urban For. Urban Green. 34, 336–347.

Delkov, N., 1975. Dynamics of germination of eastern plane seeds. Gorsko Stopanstvo 6, 18–20 (In Bulgarian).
Delkov, N., 1977. Studies on the Biological and Ecological Characteristics of the Eastern Plane (*Platanus orientalis* L.) in Bulgaria. Ph.D. thesis. University of Forestry, Sofia (in Bulgarian).
Delkov, N., 1979. On the rooting of Eastern Plane (*Platanus orientalis* L.) cuttings. High. Inst. Forest. Res. Pap. Ser. Fores. 21, 61–67.
Forest Seed Control Station, 2023. Unpublished Results of Seed Testing Sofia.
Guoqiang, F., Jianping, J., Yaoqing, H., Feng, L., 2003. In vitro plant regeneration with *Platanus orientalis* leaves as explants. Acta Hortic. Sin. 30, 236–238 (in Chinese with English abstract).
Milev, M., Alexandrov, P., Petkova, K., Iliev, N., 2004. Sowing Materials of Broadleaved Species. Videnov & Son Publ, House, Sofia (in Bulgarian).
Panetsos, K., Scaltsoyiannes, A., Alizoti, P., 1994. Vegetative propagation of *Platanus orientalis x P. occidentalis* F1 hybrids by stem cuttings. For. Genet. 1 (3), 125–130.
Piotto, B., Bartolini, G., Bussotti, F., et al., 2001. Schede informative sulla propagazione per seme degli alberi e degli arbusti piu' diffusi della Flora Mediterranea. In: Piotto, B., Di Noi, A. (Eds.), Propagazione per Seme di Alberi e Arbusti Della Flora Mediterranea. Manuale ANPA, Rome, Italy, pp. 109–170.
Rezaei, Z., Nasr, S.M., Asadi, H., 2023. Vegetative propagation of *Platanus orientalis* L. by coppice shoot cuttings. Forestist Hoseini 73 (3), 264–269.
Sajid, M., Pervaiz, M., Rab, A., Jan, I., Haq, I., Wahid, F., Shah, S.T., Ali, I., 2012. Response of plane tree (*Platanus orientalis*) to cuttings and planting dates. J. Anim. Plant Sci. 22 (2), 420–424.
Showkat, W., Gangoo, S.A., Jabeen, N., Nazir, N., Hussain, K., Husaini, A.M., 2017. In vitro propagation of Chinar (*Platanus orientalis* L.) using node and internode explants. Appl. Biol. Res. 19 (2), 197–204.
Suliman, H.H., Ahmed, G.G., 2014. Effect of altitudinal variation on seed and seedling growth characters of *Platanus orientalis* L. in Duhok province. J. Univers. Duhok Agricult. Veterin. Sci. 17 (1), 108–120.
Zencirkiran, M., Erken, K., 2012. The effect of different times collecting cutting and auxin treatments of the rooting in *Platanus orientalis* L. (Oriental Plane tree - Cinar). J. Animal Plant Sci. 22 (3), 764–767.

Populus L. (*Populus alba* L. and *Populus nigra* L.)

Claudia Cocozza[1], Zvonimir Vujnović[2] and Gregor Božič[3]
[1]Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy; [2]Department of Genetics Forest Tree Breeding and Seed Science, Croatian Forest Research Institute (CFRI), Jastrebarsko, Croatia; [3]Department of Forest Physiology and Genetics, Slovenian Forestry Institute (SFI), Ljubljana, Slovenia

Propagation

Seed propagation

Populus alba (white poplar) and *Populus nigra* (European black poplar) are heliophilous (shade-intolerant) forest tree species. *P. alba* shows optimal growth in temperate climates, on well-drained, deep, silty or sandy-silty soils. *P. nigra* thrives on deep, nutrient rich, medium-textured soils with a pH of 5.5–7.5 (Richardson et al., 2014). Both species are dioecious thus requiring outcrossing. Their reproductive structures consist of inconspicuous flowers in pendulous catkins. Male catkins are reddish-purple, while female catkins are greenish; both primarily are located in the upper crown. Anthesis precedes foliar development, typically between February and April, depending on climate. Pollination is anemophilous. The fruits are oblong-conic capsules, approximately 4 mm in length, maturing from March to June. Seed are dispersed by wind and water, with mature trees producing seeds annually.

Seed collection is conducted from March to June, employing various methods including long-handled cutting tools, ground collection, or whole-tree harvesting. Timing is critical, as collection must coincide with the onset of capsule dehiscence, necessitating careful field monitoring due to rapid seed dispersal. Premature collection is contraindicated, as seeds must reach full maturation. A recommended practice involves harvesting branches bearing early-ripening catkins and rapidly transporting them to a greenhouse (Vujnović et al., 2023). The cut ends are placed in water-filled containers and the branches are covered with a breathable cotton sheet to contain the seeds following capsule opening. Under semicontrolled greenhouse conditions, where day temperature ranged between 15°C and 34°C, and adequate humidity was maintained, catkin dehiscence and seed release occurred within 24 h. A single meter-long branch may yield four to seven catkins, each containing an average of 15 seeds per capsule (Vujnović et al., 2023). Postharvest maturation of *P. nigra* catkins at room temperature for 3–5 days facilitates capsule dehiscence and seed release.

P. nigra seeds have white silky hairs (Fig. 11.33), facilitating wind dispersal over long distances (Barsoum, 2001). While removal of the silky hairs is optional, it simplifies seed handling. Seeds can be separated from the hairs through mechanical abrasion using a 2.38 × 2.38 mm mesh sieve and compressed air (Vujnović et al., 2023). Although *P. nigra* seeds exhibit high germination and vigor, their limited energy reserves restrict longevity to days or weeks (Moss, 1938; Muller et al., 1982; Johnson, 1994; Van Splunder et al., 1995; Braatne et al., 1996; Karrenberg and Suter, 2003). Due to their rapid loss



Figure 11.33 Upon maturation, *Populus nigra* capsules split open to disseminate their short-lived, pappus bearing seeds with cottony hairs. *(Zvonimir Vujnovič.)*

of viability at ambient temperature and humidity, seed processing should be completed within 1 week after collection.

Seeds of *Populus* spp. are classified as suborthodox (Bonner, 2008) or intermediate (Suszka et al., 2014; Michalak et al., 2015). Seed viability can be maintained for ~4 weeks at 4°C under non-drying conditions (Gosling, 2007). For long-term storage (several years), seed moisture should be reduced to 5%–8% and stored in airtight containers at 4–5°C. Ultra-long-term storage requires temperatures below 0°C (e.g., −18°C) with 6%–8% moisture content. Rehydration of long-term stored seeds must be gradual to prevent imbibitional damage. Germination under controlled conditions requires no pre-treatment, with an optimal temperature of 20–25°C. Seed characteristics of *P. nigra* and *P. alba* are summarized in Table 11.48.

Vegetative propagation

Populus species are easily propagated vegetatively using hardwood and root cuttings. Optimal cuttings should be harvested from healthy, vigorous mature branches located in the lower part of the tree crown or from new shoots. Cuttings are typically 10–15 cm in length, containing 10 internodes, or 10 cm in length with five internodes.

Table 11.48 Characteristics of seed lots of *Populus nigra* and *Populus alba*.

Species	Purity (%)	No. seeds/kg	Germination capacity (%)
P. nigra	85–99 (92)	1,000,000–1,100,000 (1,050,000)	77–95 (86)
P. alba	85–99 (92)	1,600,000–1,800,000 (1,700,000)	60–95 (78)

Mean values in brackets.
Piotto et al. (2001), Bonner (2008), Prada et al. (2008), Suszka et al. (2014), Michalak et al. (2015).

For *P. nigra*, cutting diameters should range from 8 to 20 mm (Sabatti et al., 2001), with a general length of 20–30 cm (Fig. 11.34). Cuttings can be harvested both in winter and summer. Winter cuttings are collected from December to March, with early spring collection (February–March) in Mediterranean climates. Cuttings are stored in a cool, humid environment, either in cold storage, buried in sand, or within sawdust to maintain moisture (Bonner, 2008) until planting in the ground or pots in March.

Auxin application is generally unnecessary for propagating *Populus* cuttings. For *P. alba*, propagation success is highly influenced by ortet genotype (Prada et al., 2008), with substantial mortality when propagating diverse clonal material (Sekawin, 1975). For recalcitrant genotypes, rhizogenesis can be enhanced by using internodal material immediately below a node. This material should be subjected to a water pretreatment at 16°C under darkness until root initiation (Phipps et al., 1983), followed by transplanting to a rooting bed at 20°C. The enhanced root development observed in nodal-proximal regions likely results from localized concentration of endogenous root-promoting phytohormones.

Propagation using current-year shoots (green cuttings) collected in July and August requires careful moisture management (Barsoum, 2001). These cuttings are highly prone to desiccation and root poorly if not handled properly. Immediate immersion in water is



Figure 11.34 Hardwood cuttings as a potential source for valuable *Populus nigra* genotype propagation. *(Zvonimir Vujnovič.)*

crucial to maintain turgor and prevent desiccation. Green cuttings show the highest rooting for *P. alba*, probably because mother trees are in optimal physiological condition for vegetative propagation at that time (Harfouche et al., 2007).

Populus species can also be propagated via root segments, which can form roots, be used directly for plant production, or serve as green cuttings.

In vitro propagation of *Populus* spp. is feasible using catkins, stem segments, and axillary leaf buds (Ahuja, 1987). This technique involves cultivating tissue explants under controlled laboratory conditions to generate new plants. While in vitro methods facilitate the mass production of genetically uniform individuals (Bonner, 2008), the high costs in time, labor, and resources limit its practicality for large-scale forestry (Suszka et al., 2014; Michalak et al., 2015).

Seedling cultivation

Vegetative propagation is the main method for *Populus* spp. cultivation in nurseries, as it is easier than seed propagation, ensures genetic uniformity (a crucial factor for timber production), and enhances plant survival. Where greenhouse facilities are available, cutting cultivation can start in early December, coinciding with the initiation of preparing cuttings from mother stock plants. In nurseries without heated greenhouses, cuttings can be stored in cold storage or buried outdoors in soil. To prevent desiccation during storage, cuttings should be wrapped in damp jute bags. Greenhouses should be maintained at optimal temperatures, with supplemental heating during cold, low-light periods, along with adequate moisture, to promote root development.

Bareroot plants

Cuttings should be planted vertically in rows, with one-third of the cutting (containing one to three buds) remaining above the ground. Care must be taken to avoid damage to the apical bud and bark. Spacing is critical for plant development. For establishing mother plant fields, high planting density with reduced inter-plant distances are recommended. Rueda Fernández (2013) suggest densities ranging between 25,000 and 50,000 plants/ha for timber with *Populus* clones coppiced up to 2 or 3 years. For *P. nigra*, Maestro-Tejada and Alba Monfort (2013) recommend a spacing of 2 × 3 or 3 × 3 m for mother plant fields coppiced up to 8 years. Wider spacing is recommended for plants used for forestation. Excessively high densities result in etiolated growth, characterized by slender stems with reduced mechanical strength, making plants more susceptible to bending and wind damage after transplanting. Row spacing commonly ranges from 1 to 2 m, with intra-row spacing of 30–40 cm.

Supplemental irrigation is essential, particularly during extended dry periods, to ensure timely plant development. Irrigation frequency, typically every few days, depends on the weather conditions and plant size. In greenhouse settings, selective pruning of

lateral branches may be implemented to promote apical dominance and enhance plant vigor, improving their resistance to stress in field conditions after transplanting.

Fertilization should be tailored to correct nutrient deficiencies or suboptimal growth. Specific formulation, application rate, and frequency must be determined based on visual diagnosis and site-specific soil analysis. For field-grown plants, an initial application of balanced NPK fertilizer (e.g., 10-10-10) at a rate of 50–100 g/plant is recommended for plant establishment. Annual spring applications of 100–150 g/plant should continue in subsequent years, with an optional mid-season fertilization in late spring or early summer to boost growth. Mature trees ($\geq$5 years) typically require fertilization every 2–3 years at a rate of 300–500 g/plant.

Following leaf emergence, management of defoliating insect populations may be necessary. Insecticide selection, application rate, and frequency should be determined based on local pest pressure and established recommendations. Pyrethroid insecticides, including Cypermethrin (0.5–1 L/ha), Permethrin (0.5–1 L/ha), and Deltamethrin (0.05–0.1 L active ingredient/ha; equivalent to a 0.05%–0.1% solution with 100 L/ha), demonstrate efficacy against a broad spectrum of defoliating insects, such as poplar leaf beetles, tent caterpillars, and aphids. Biological insecticides like *Bacillus thuringiensis* (2.5–5 L/ha) are specific to larvae. To prevent herbivore damage, nurseries should be enclosed with a robust fence.

During harvest, roots should be thoroughly buried in moist soil (Božič et al., 2021), and plants should be covered with a shading net to minimize desiccation. Typically, 1 + 2 or 2 + 3 plants are used for forest planting.

Seed propagation requires seed sowing immediately after collection, without pretreatment. Direct seeding uses approximately 3000 seeds/m^2 on mechanically prepared seedbeds to facilitate sowing. Seeds with intact white silky hairs are preferred, as these structures aid adhesion to the moist substrate. After germination, adequate hydration is crucial for seedling establishment. Initial nutrient requirements are relatively low, as seedlings can effectively take up soil nutrients. Seed size influences germination success and seedling development. Seeds should not be covered or, if pressed into the substrate, should not be buried deeper than 2–3 times their diameter. Germination typically occurs within 12–24 h after sowing. Seedlings are highly vulnerable to drought stress during the first month.

Container plants

Containerized cultivation provides advantages over bareroot methods, particularly for out-of-season planting or challenging site conditions. For container-grown cuttings, container size is a critical factor. Initially, planting in smaller containers may require subsequent transplanting to larger containers to facilitate robust root system development. The final container volume depends on the production objective. Generally, a minimum volume of 6 L is necessary to provide enough space for roots to grow in all directions. Container depth should be at least 30–40 cm to facilitate unrestricted root growth.

A suitable growing medium for cuttings can be a 1:1:1 mixture of peat (pH near 7), fine-grained sand, and perlite, which promotes moisture retention and aeration (Vujnović et al., 2023). Commercial growing media mixtures are also available. Consistent irrigation is essential for successful rooting of cuttings. If rooting hormones are employed, Indole-3-butyric acid (IBA) can be applied at concentrations of 1000–3000 mg/L to stimulate root primordia formation (Harfouche et al., 2007). As with bareroot stock production, cuttings should be planted vertically in the growing medium with approximately one-third of the cutting containing one to three buds remaining above the ground.

For seed propagation, starting cultivation in small pots (up to 70–75 cm^3) is recommended. Once established, seedlings can be transplanted into larger containers ranging from 300 mL to 3.5 L (Vujnović et al., 2023). Under semicontrolled conditions, seedlings grow rapidly and develop dense root systems. The same substrate used for cuttings can be employed for seedling cultivation. *P. nigra* seed germination typically occurs rapidly (within 24–48 h) given adequate moisture (Siegel and Brock 1990; Van Splunder et al., 1995). However, seedlings are shade-intolerant (Gage and Cooper 2005), making sufficient light crucial for successful germination. Postemergence, high humidity and temperature predispose seedlings to fungal diseases, necessitating timely fungicide applications.

Nutrient leaching from substrates is a concern; therefore, supplemental fertilization is recommended. During the early rooting stage, a balanced or moderate amount of N is necessary to support initial growth, with NPK (relative proportions of 1:1:1 or 2:1:2). During active vegetative growth, we recommend that the relative proportion of N, P, and K in the fertilizers to be 3:1:2 or 4:1:2, as higher N supports shoot development, and higher P promotes root growth. Finally at the plant maturity stage, the relative NPK proportion should be 2:1:3 or 1:1:2, where increased K enhances stress and disease resistance, and general plant health. When flowering and seed production occur, an NPK ratio of 1:2:2 or 1:3:2 supports reproductive processes, with increased P and K facilitating flowering and fruiting.

Controlled-release fertilizers offer a single-application approach for season-long nutrient provision in containerized production and are typically incorporated into the substrate at a rate of 3–5 g/L.

Field establishment

Preplanting hydration is crucial, particularly during dry winters. Soaking bare roots in water for 1–2 days prior to planting is recommended. Timing during spring planting should coincide as closely as possible with anticipated rainfall, ideally within a 15-day window. Briefly immersing seedling roots in a muddy slurry before planting can increase establishment (Božič et al., 2021). However, autumn planting is generally preferred due to the increased frequency and severity of dry springs in many regions. If immediate

planting is not feasible, plants must be protected from desiccation. Bareroot seedlings should be heeled in by covering the roots with soil, while container seedlings can be stored above ground, protected from direct sunlight, and watered as needed.

Populus alba is a heliophilous species, adapted to full sunlight, which thrives on neutral to alkaline substrates and is tolerant of heavy soils and moderate salinity. It forms dense stands or grows sparsely in lowland or swampy riparian zones. In cooler climates, it cooccurs with *P. nigra*, while in warmer or coastal regions, it becomes the dominant species due to its thermophilic nature. Weed competition management is crucial during the initial 1–2 years postplanting, particularly in specific natural site conditions. Rotary cutting and manual removal of climbing plants (e.g., wild hops and clematis) using hand shears can help to minimize damage to young plants (Sallmannshofer and Ruhm, 2021).

Before planting, it is advisable to analyze soil pH, nutrient levels, and texture (sand, silt, clay) to optimize site selection and preparation as poplars thrive better on loamy soils with optimal drainage and a pH between 5.5 and 7. Correction of nutrient imbalances by adding fertilizers or organic amendments based on test results is suggested Although poplars tolerate wet conditions, proper drainage is necessary to prevent root rot. In compacted soils, using a subsoiler to break up the soil to a depth of at least 30–40 cm is recommended. Fertilization should be tailored to different growth stages. Before planting, a balanced fertilizer (e.g., 10:10:10 NPK), typically, a rate of 50–100 kg/ha is recommended. After planting, during the first year, a fertilizer enriched in N can be especially beneficial for root and shoot growth, with a dose of 50–100 g/tree. Fertilization should take place in early spring (before or at planting) and additional fertilization can be supplied in early summer. We recommend avoiding late-season fertilization (late summer or fall), as it can promote late-season growth, increasing plant vulnerability to frosts. For mature trees, fertilization may not be necessary unless soil nutrients are low. However, periodic maintenance fertilization every 2–3 years can help maintain health and productivity.

Deer herbivory presents a significant threat to poplar establishment, with damage manifesting as bark stripping and foliage and bud browsing. Mitigation strategies include physical and chemical deterrents. Electric fencing, while effective in North America (Brenneman, 1982), requires maintenance to ensure integrity, including vegetation control to prevent grounding. High-tensile woven wire fencing, although offering a robust physical barrier (Dickman and Lantagne, 1997), entails high initial investment costs. In certain regions, such as Croatia, localized protection methods involve wrapping individual seedlings in reeds secured with wire. Although more affordable than woven wire fencing, this method demands increased labor for individual plant treatment.

Chapter References

Ahuja, M.R., 1987. *In vitro* propagation of poplar and aspen. In: Bonga, J.M., Durzan, D.J. (Eds.), Cell and Tissue Culture in Forestry. Forestry Sciences. Springer, Dordrecht, pp. 24–26. https://doi.org/10.1007/978-94-017-0992-7_16.

Barsoum, N., 2001. Relative contributions of sexual and asexual regeneration strategies in *Populus* nigra and *Salix alba* during the first years of establishment on a braided gravel bed river. Evol. Ecol. 15 (4–6), 343–361.

Bonner, F.T., 2008. Storage of seeds: potential and limitations for germplasm conservation. For. Ecol. Manag. 256 (4), 593–602.

Božič, G., Westergren, M., Konrad, H., Lanšćak, M., Nagy, L., Novčić, Z., Rukavina, S., Schüler, S., Stojnić, S., Železnik, P., 2021. Guidelines for the production of planting material for restoration of riparian forests. Stud. Forestal. Sloven. 170.

Braatne, J.H., Rood, S.B., Heilman, P.E., 1996. Life history, ecology, and conservation of riparian cottonwoods in North America. In: Stettler, R.F., Bradshaw, H.D., Heilman, J.P.E., Hinckley, T.M. (Eds.), Biology of Populus and its Implication for Management and Conservation. Part I, Chapter 3. National Research Council of Canada, Ottawa, Canada, pp. 57–85.

Brenneman, R., 1982. Electric fencing to prevent deer browsing on hardwood clearcuts. J. For. 80 (10), 660–661.

Dickmann, D.I., Lantagne, D., 1997. Planting oaks for timber and other uses. North Central Regional Extension Publication No. 605. Michigan State University Extension, East Lansing, p. 12.

Gage, E.A., Cooper, D.J., 2005. Patterns of willow seed dispersal, seed entrapment, and seedling establishment in a heavily browsed montane riparian ecosystem. Can. J. Bot. 83, 678–687. https://doi.org/10.1139/b05-042.

Gosling, P., 2007. Raising Trees and Shrubs from Seeds; Practice Guide. Forestry Commission, Edinburgh, pp. 1–28.

Harfouche, A., Baoune, N., Merazga, H., 2007. Main and interaction effects of factors on softwood cutting of white poplar (*Populus alba* L.). Silvae Genet. 56 (1–6), 287–294.

Johnson, W.C., 1994. Woodland expansion in the Platte river, Nebraska: patterns and causes. Ecol. Monogr. 64, 45–84. https://doi.org/10.2307/2937055.

Karrenberg, S., Suter, M., 2003. Phenotypic trade-offs in the sexual reproduction of *Salicaceae* from flood plains. Am. J. Bot. 90, 749–754. https://doi.org/10.3732/ajb.90.5.749.

Maestro Tejada, C., Alba Monfort, N., 2013. Populus nigra. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales, vol II. Organismo Autónomo Parques Nacionales. Serie Forestal, Madrid, Spain, pp. 75–95.

Michalak, M., Plitta, B.P., Tylkowski, T., Chmielarz, P., Suszka, J., 2015. Desiccation tolerance and cryopreservation of seeds of black poplar (*Populus nigra* L.), a disappearing tree species in Europe. Eur. J. For. Res. 134, 53–60.

Moss, E.H., 1938. Longevity of seed and establishment of seedlings in species of *Populus*. Botan. Gazz. 99, 529–542. https://doi.org/10.1086/334728.

Muller, C., Teissier du Cros, E., Laroppe, E., Duval, H., 1982. Conservation pendant 5 ans de graines de peupliers noirs (*Populus nigra* L.). Ann. For. Sci. 39, 179–185. https://doi.org/10.1051/forest:19820205.

Phipps, H.M., Hansen, E.A., Fege, A.S., 1983. Pre-plant soaking of dormant *Populus* hardwood cuttings. In: USDA Forest Service Research Paper NC-RP-241. North Central Forest Experiment Station, St. Paul, MN.

Prada, M.A., Arizpe, D., Añíbarro, J., Rueda, J., Albert, N., Campos, E., Picher, M., Ventimilla, P., Gálvez, C., Faria, C., Jiménez, C.P., 2008. In: Prada, M.A., Arizpe, D. (Eds.), Riparian Tree and Shrub Propagation Handbook. An Aid to Riverine Restoration in the Mediterranean Región. CIEF-Banc de Llavors Forestals, Generalitat Valenciana, Valencia, pp. 97–104.

Richardson, J., Isebrands, J.G., Ball, J.B., 2014. Ecology and physiology of poplars and willows. In: Isebrands, J.G., Richardson, J. (Eds.), Poplars and Willows: Trees for Society and the Environment. CAB International, FAO, pp. 92–123.

Rueda-Fernández, J., 2013. *Populus* spp. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales, vol II. Organismo Autónomo Parques Nacionales. Serie Forestal, Madrid, Spain, pp. 39–55.

Sabatti, M., D'Ovidio, R., Tanzarella, O.A., Scarascia Mugnozza, G., 2001. Assessment of geographic variation by RAPD markers among Italian open-pollinated progenies of *Populus alba* L. Genet. Resour. Crop Evol. 48, 423–428. https://doi.org/10.1023/A:1012095401517.

Sallmannshofer, M., Ruhm, W., 2021. Integrating biodiversity enrichment in poplar forest management. In: Sallmannshofer, M., Schueler, S., Westergren, M. (Eds.), Perspectives for Forest and Conservation Management in Riparian Forests, vol 169. Studia Forestalia Slovenia, pp. 106–110. https://doi.org/10.20315/SFS.169.

Sekawin, M., 1975. Genetics of *Populus alba*. Ann. Forest. 6 (6), 159–189.

Siegel, R.S., Brock, J.H., 1990. Germination requirements of key Southwestern woody riparian species. Desert plants 10, 3–8.

Suszka, J., Plitta, B.P., Michalak, M., Bujarska-Borkowska, B., Tylkowski, T., Chmielarz, P., 2014. Optimal seed water content and storage temperature for preservation of *Populus nigra* L. germplasm. Ann. For. Sci. 71, 543–549.

Van Splunder, I., Coops, H., Voesenek, L.A.C.J., Blom, C.W.P.M., 1995. Establishment of alluvial forest species in floodplains: the role of dispersal timing, germination characteristics and water level fluctuations. Acta Bot. Neerl. 44, 269–278. https://doi.org/10.1111/j.1438-8677.1995.tb00785.x.

Vujnović, Z., Bogdan, S., Lanšćak, M., Gavranović Markić, A., Zorić, N., Bogunović, S., Ivanković, M., 2023. Preliminary work on generative seedling production and clone selection of European black poplar (*Populus nigra* L.). South-East Europ. Forest. 14 (1), 101–109.

Quercus faginea Lam.

Pedro Villar-Salvador[1], Juan A. Oliet[2], Luna Morcillo[3], Alberto Vilagrosa[3] and Juan Luis Nicolás Peragón[4]

[1]Universidad de Alcalá, Forest Ecology and Restoration Group, Departamento de Ciencias de la Vida, Alcalá de Henares, Madrid, Spain; [2]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [3]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain; [4]Centro Nacional de Recursos Genéticos Forestales "El Serranillo", MITECO, Guadalajara, Spain

Propagation

Seed propagation

Quercus faginea Lam. (Portuguese oak) is a masting species like other oaks (Sánchez Humanes, 2011), which will make commercial-scale harvesting of acorns difficult in some years. Acorns mature between early October and early November of the same year the tree flowers (Milla et al., 2010) and they mature earlier than the acorns of *Quercus ilex* L. with which it often coexists. Frosts below −6.9°C severely damage the acorns of *Q. faginea*, which shows lower frost resistance than that of *Quercus coccifera* L. and *Q. ilex* (Esteso-Martínez and Gil Pelegrín, 2004).

Acorn collection should be done when they are about to fall or have recently fallen. Acorns can be collected directly from the ground or after gently shaking the branches with rods. It is advisable to use nets placed on the ground to increase collection efficiency and reduce contamination by soil-borne pathogens. The collection yield of *Q. faginea* is typically 30 kg/day from 10 to 15 trees (Villar-Salvador et al., 2013) with an average yield per tree ranging from 1 to 11 kg in agroforestry systems (Medina Blanco, 1956).

The acorns of *Q. faginea*, like other oak species, are recalcitrant requiring special conservation to prevent significant moisture loss before use in nurseries or forestation. Critical moisture levels that significantly inhibit germination for this species are currently unknown, but they are likely similar to those reported for *Q. ilex*. For processing, handling, and storage of acorns, we recommend following the same guidelines outlined in the *Q. ilex* section of this compendium.

Quercus faginea propagates easily using acorns, which usually have a high germination rate if kept hydrated. No germination pretreatment is necessary, although immersing the acorns in water for 24 h homogenizes the germination time (García-Fayos, 2001). Table 11.49 presents the average characteristics of *Q. faginea* acorn lots.

Vegetative propagation

Fernández-Galiano et al. (1997) showed that it is possible to obtain *Q. faginea* plants through somatic embryogenesis using zygotic embryos. Similarly, it is possible to graft one-year-old twigs of this oak on rootstocks of the same species for acorn production (Peñuelas Rubira, 2022).

Table 11.49 Characteristics of seed lots of *Quercus faginea*.

Purity (%)	No. seeds/kg	Germination capacity (%)
94–100 (97)	230–430 (325)	70–90 (80)

Mean values in brackets.
Aissi et al. (2019); Catalán (1991); García-Fayos (2001); González-Rodríguez et al. (2011); Navarro Cerrillo and Gálvez Ramírez (2001); Villar-Salvador et al. (2013).

Seedling cultivation

Container plants

Quercus faginea can be grown both in containers and as bareroot stock. We recommend using container plants (Fig. 11.35), especially on dry sites, as they usually perform better than bareroot plants under harsh planting conditions (Grossnickle and El-Kassaby, 2016). According to our experience, cultivation in container nurseries can be started in mid-autumn, immediately after collecting the acorns, or it can be delayed until late winter and early spring. However, autumn sowing should be avoided outdoors in locations with severe winter frosts and prolonged spring frosts. In such cases cultivation should begin inside a greenhouse. Due to their size, only one acorn should be seeded in each container cell and should be placed horizontally or slightly downward and no deeper than 3 cm. To ensure that all cells of the container trays are occupied, we recommend pregerminating the acorns in seed trays with moist peat. Once most acorns start to germinate and the radicle length is less than 2 cm, germinating acorns can be transferred to the final cultivation containers. This operation is particularly recommended for seed lots of uncertain quality. In other oak species it has been reported that if the radicle length exceeds 5 cm, it can be trimmed up to 3 cm to facilitate seeding without apparent negative consequences for seedling growth (Piotto and di Noi, 2003). Excising the radicle typically results in seedlings developing multiple taproots. The germination of *Q. faginea* is hypogeal and seedling emergence occurs after 0.5–2 months depending on environmental conditions (Villar-Salvador et al., 2013a).

We recommend cultivating this oak in containers with a volume of 300–400 cm^3 since large containers, especially deep containers, usually benefit the field establishment of oaks (Chirino et al., 2008; Domínguez Lerena, 1999; Tsakaldimi et al., 2005). Growing media can be blonde or black peat, fertilized or not, or mixtures of coconut coir with peat, in both cases with additions of vermiculite up to 20% of the total volume. Cultivation density of *Q. faginea* should be slightly lower than the density used for cultivating *Q. ilex*, since it has larger leaves and therefore needs more lateral space. We recommend a maximum cultivation density of 350 plants m^2 (Villar-Salvador et al., 2013a).

Due to its deciduous habit and faster growth, *Q. faginea* demands more nutrients for its growth than most Mediterranean evergreen oaks (Cornelissen et al., 1996, 1997). Fertilization can be conducted by fertigation, which is the method we recommend if the nursery infrastructure allows for it. Good quality plants with high outplanting growth in the field are obtained when seedlings are supplied with at least 75 mg of N with the best performance at 150 mg of N per plant, distributed over 20 weeks (once per week) (Villar-Salvador et al., 2013b). Low fertilization rates produce small, nutrient poor plants (Fig. 11.35) with low outplanting performance (Villar-Salvador et al., 2013b). Fertilization can also be supplied using a slow release fertilizer mixed with the growing medium. We suggest a dose of 3–4 g/L with a release time of 9–12 months.

Nurseries in very hot summer locations can shade the seedlings by reducing irradiation not more than 60% during the hottest months of the summer. This protects the plants from high radiation and reduces water consumption. During the rest of the cultivation season, however, seedlings should be cultivated under the full sun to produce hardened seedlings.

Quercus faginea is a suitable host for mycorrhization with *Tuber melanosporum*, the black truffle, adding extra-value to plantings of this species (Reyna Domenech, 2012). Mycorrhization increases P content and, to a lesser extent, N content of seedlings in the nursery without negative effects on growth or their water stress tolerance (Domínguez Núñez et al., 2008). In the field, mycorrhization increases growth, although not survival compared with plants that have not been inoculated with this fungus (Domínguez Núñez et al., 2006). Mycorrhization can be stimulated by simultaneously



Figure 11.35 Left: *Quercus faginea* seedlings cultivated with increasing N fertilizer rates. From left to right the plants received 0, 35, 75, 150 and 250 mg of N. The best field performance was observed in the 150 mg-N plants. Right: A tray with seedlings fertilized with 150 mg of N. Further information on cultivation conditions can be found in Villar-Salvador et al. (2013b). *(Pedro Villar-Salvador.)*

inoculating *T. melanosporum* and the bacteria *Pseudomonas fluorescens* at a dose of 1 × 108 colony forming units/mL (Piñuela et al., 2020).

Table 11.50 shows the morphological and nutrient concentration ranges together with the recommended values for high-quality 1 + 0 *Q. faginea* plants. Branching in 1 + 0 seedlings is infrequent but becomes common in 2 + 0 and older plants. Although deciduous, the leaves of *Q. faginea* normally remain green and photosynthetically active throughout the first winter in 1 + 0 seedlings, especially in well-fertilized plants, shedding older leaves during spring bud break (Villar-Salvador et al., 2013b). Conversely, leaves of older plants shed in autumn or remain brown, on the plant without activity.

Bareroot plants

We found no information on bareroot plants of *Q. faginea*.

Field establishment

Quercus faginea can be established either by planting or by direct sowing. For planting, we recommend 1 + 0 seedlings as they have shown higher field survival and growth compared to 2 + 0 plants (Nicolás Peragón et al., 2004). If 2 + 0 seedlings are used, we recommend seedlings to be cultivated in containers with volumes of at least 300 cm^3. When sowing directly, deep sowing is recommended (Carreras Egaña et al., 1997), with one acorn per spot. It is crucial to protect acorns from predators, and different methods have been developed for this purpose (Castro et al., 2015; Löf et al., 2019). No differences in short-term survival were observed in a field comparison

Table 11.50 Morphological and physiological attributes of 1 + 0 container seedlings of *Quercus faginea*. The root collar diameter is measured immediately above the cotyledon insertion point.

Attributes	Observed values	Recommended values
Shoot height (cm)	8–30	>18
Root collar diameter (cm)	0.4–0.5	>0.45
Shoot dry weight (g)	0.7–1.9	>1.2
Root dry weight (g)	1.6–4.0	>3.7
Plant dry weight (g)	3.5–6.2	>4.8
Shoot/root dry weight ratio	0.2–0.5	>0.35
Leaf [N] (mg/g)	14–20	>16.5
Leaf [P] (mg/g)	0.5–5	>3.0
Leaf [K] (mg/g)	2.0–5.0	>3.5
Root [N] (mg/g)	6–13	>11.0
Root [P] (mg/g)	0.4–2.7	>2.2
Root [K] (mg/g)	3.2–5.7	>3.5

Data sources: Villar-Salvador et al., (2013b) and Nicolás-Peragón and Villar-Salvador, unpublished data.

of both reforestation methods. However, planted seedlings grew faster in height than seedlings grown from direct seeded acorns (Pemán et al., 2017).

Under water stress, *Q. faginea* has higher transpiration rates and lower water use efficiency than evergreen oaks such as *Q. ilex* and *Quercus suber* L., cork oak (Alonso-Forn et al., 2021; Mediavilla and Escudero, 2003). *Q. faginea* has deep roots and consequently it should be established on sites with deep soils and on substrates with a high-water storage capacity such as loamy textures, and in locations that retain moisture in summer such as soil depressions and north-facing slopes. Planting outside these conditions can significantly increase mortality compared with other Mediterranean species (del Campo et al., 2020) We recommend intensive soil preparation to facilitate deep root system development, such as subsoiling along contour lines and mechanical digging to depths between 40 and 60 cm. A comparison between subsoiling and mechanical planting holes opened with a backhoe showed no differences in *Q. faginea* survival, although height growth was higher in the mechanical planting holes (Nicolás Peragón et al., 2004).

Q. faginea planted seedlings present high root elongation rates in autumn and spring, with root activity decreasing significantly during winter to only 7%–15% of the maximum spring elongation rate (Aguilar et al., 2005). Therefore, we suggest planting in the fall, preferably in November with available soil moisture, or at the end of February for cold winter sites. If sowing is the forestation method, we recommend seeding in mid-February on site with long and cold winters or in the mid-autumn on mild winter sites.

The maximum planting density for *Quercus faginea*, considering its large adult size and resprouting ability, should not exceed 1000 plants/ha for pure and mixed stands. The use of tree shelters speeds growth and slightly increases the survival of this oak particularly when using ventilated tubes. However, light transmission of the tree shelter lower than 30% reduces spring growth potential and thus the ability to overcome limitations in the summer establishment period (Pemán et al., 2010, 2017). It is prudent to keep the tree shelters in place until the plants' stems have sufficiently thickened to support themselves without external assistance. This typically occurs no earlier than the fifth or sixth year for this species, according to our experience.

Postplanting irrigation is usually unnecessary provided that planting was performed at the right time and with the adequate soil preparation. In exceptionally dry years, however, maintenance irrigation of 10–40 L per plant, applied up to three times throughout the first summer, may be necessary. Fertilization of *Q. faginea* plantations is not recommended, given the poor results observed in other oak species (Oliet et al., 2003). It is preferable for seedlings used in plantations to have a good nutritional status acquired in the nursery. Where vigorous herb competition is expected, such as cropland (Fig. 11.36), weeds need to be removed in a 0.5 m radius around the plants for the first two to 4 years (Rey Benayas et al., 2005). Weeds can be removed through manual or mechanical weeding, herbicides, and mulching (Coello et al., 2018), with better results



Figure 11.36 A young 12-year-old *Q. faginea* tree planted in a former cropland in Monreal del Campo, Teruel, Spain at 1000 m asl. Planting was done in mid-February 2003 using 1 + 0 seedlings cultivated in 300 cm^3 containers. Soil was subsoiled 60 cm deep before planting. Weeds were mechanically removed the first year. *(Pedro Villar-Salvador.)*

achieved through manual or mechanical interventions. Weed control efforts should be concentrated in the first half of spring.

The use of nurse plants for establishing *Q. faginea* has shown disparate results. While Spanish broom (*Retama sphaerocarpa* L.), a leguminous shrub is known for facilitating the establishment of the evergreen oak *Q. ilex* (Andivia et al., 2017; Cuesta et al., 2010; Rolo et al., 2013), it did not facilitate *Q. faginea* (Cruz-Alonso et al., 2020). Gómez-Aparicio et al. (2004) (refer to the supplementary material in this paper) observed increased *Q. faginea* survival when planted under *Ulex parviflorus*, a thorny legume shrub, compared with planting in open areas. Soil preparation methods that avoid damaging the shrub, such as using mechanical augers or hammers, or hand-dug holes should be selected when planting under nurse plants.

Granados et al. (2016) found early positive survival and growth of *Q. faginea* when planted inside 60-year-old *Pinus halepensis* Mill. plantations. This positive effect of pine canopies persisted after 10 years and *Q. faginea* establishment was maximized at pine densities ranging from 800 to 1400 trees/ha, with maximum survival rates of 54%, while the plantation under low pine density (100–250 trees/ha) resulted in survival rates of 32% (Morcillo et al., 2022).

Chapter References

Aguilar, A., Valle, G., Sardá, P., Villar-Salvador, P., Peñuelas Rubira, J.L., 2005. Crecimiento radical de plantones de *Quercus coccifera y Quercus faginea* durante el período húmedo del año. In: IV Congreso Forestal Español. SECF, Zaragoza.

Aissi, A., Beghami, Y., Heuertz, M., 2019. The Portuguese oak in Algeria (*Quercus faginea*, Fagaceae): germination potential and variability of acorn and seedling morphology. Plant Ecol. Evolut. 152, 437–449. https://doi.org/10.5091/plecevo.2019.1553.

Alonso-Forn, D., Peguero-Pina, J.J., Ferrio, J.P., Mencuccini, M., Mendoza-Herrer, Ó., Sancho-Knapik, D., Gil-Pelegrín, E., 2021. Contrasting functional strategies following severe drought in two Mediterranean oaks with different leaf habit: *Quercus faginea* and *Quercus ilex* subsp. rotundifolia. Tree Physiol. 41, 371–387. https://doi.org/10.1093/TREEPHYS/TPAA135.

Andivia, E., Villar-Salvador, P., Tovar, L., Rabasa, S., Rey Benayas, J.M., 2017. Multiscale assessment of woody species recruitment in Mediterranean shrublands: facilitation and beyond. J. Veg. Sci. 28, 639–648. https://doi.org/10.1111/jvs.12520.

Carreras Egaña, C., Sánchez Hoyos, J., Reche Pérez, P., Herrero Hinojo, D., Navarro Encinas, E., Navío Oliver, J.J., 1997. Siembras profundas con ayuda de tubos protectores. Resultados en ensayos comparativos de siembras y plantaciones bajo condiciones áridas en Vélez-Rubio. In: II Congreso Forestal Español. SECF, Pamplona, pp. 123–128.

Castro, J., Leverkus, A.B., Fuster, F., 2015. A new device to foster oak forest restoration via seed sowing. N. For. 46, 919–929. https://doi.org/10.1007/s11056-015-9478-4.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: ICONA Madrid. 4a Ed, 4th Ed. Icona, Madrid, Spain.

Chirino, E., Vilagrosa, a., Hernández, E.I.I., Matos, a., Vallejo, V.R.R., 2008. Effects of a deep container on morpho-functional characteristics and root colonization in *Quercus suber* L. seedlings for reforestation in Mediterranean climate. For. Ecol. Manag. 256, 779–785. https://doi.org/10.1016/j.foreco.2008.05.035.

Coello, J., Ameztegui, A., Rovira, P., Fuentes, C., Piqué, M., 2018. Innovative soil conditioners and mulches for forest restoration in semiarid conditions in northeast Spain. Ecol. Eng. 118, 52–65. https://doi.org/10.1016/J.ECOLENG.2018.04.015.

Cornelissen, J.H.C., Castro-Díez, P., Hunt, R., 1996. Seedling growth, allocation and leaf attributes in a wide range of woody plant species and types. J. Ecol. 84, 755–765.

Cornelissen, J.H.C., Werger, M.J.A., Castro-Díez, P., Van Rheenen, J.W.A., Rowland, A.P., 1997. Foliar nutrients in relation to growth, allocation and leaf traits in seedlings of a wide range of woody plant species and types. Oecologia 111, 460–469. https://doi.org/10.1007/s004420050259.

Cruz-Alonso, V., Villar-Salvador, P., Ruiz-Benito, P., Ibáñez, I., Rey-Benayas, J.M., 2020. Long-term dynamics of shrub facilitation shape the mixing of evergreen and deciduous oaks in Mediterranean abandoned fields. J. Ecol. 108, 1125–1137. https://doi.org/10.1111/1365-2745.13309.

Cuesta, B., Villar-Salvador, P., Puértolas, J., Rey Benayas, J.M., Michalet, R., 2010. Facilitation of *Quercus ilex* in Mediterranean shrubland is explained by both direct and indirect interactions mediated by herbs. J. Ecol. 98, 687–696. https://doi.org/10.1111/j.1365-2745.2010.01655.x.

del Campo, A.D., Segura-Orenga, G., Ceacero, C.J., González-Sanchis, M., Molina, A.J., Reyna, S., Hermoso, J., 2020. Reforesting drylands under novel climates with extreme drought filters: the importance of trait-based species selection. For. Ecol. Manag. 467, 118156. https://doi.org/10.1016/j.foreco.2020.118156.

Domínguez Lerena, S., 1999. Influencia de distintos tipos de contenedores en el desarrollo en campo de *Pinus pinea y Quercus ilex*. In: Reunión de Coordinación Del Programa I+D Forestal, Fundación CEAM. Fundación CEAM, Castellón de la Plana, pp. 81–88.

Domínguez Núñez, J.A., Planelles González, R., Rodríguez Barreal, J.A., Saiz De Omeñaca González, J.A., 2008. The effect of *Tuber melanosporum* Vitt. mycorrhization on growth, nutrition, and water relations of *Quercus petraea* Liebl., *Quercus faginea* Lamk., and *Pinus halepensis* Mill. seedlings. N. For. 35, 159–171. https://doi.org/10.1007/s11056-007-9069-0.

Domínguez Núñez, J.A., Serrano, J.S., Barreal, J.A.R., González, J.A.S.D.O., Domínguez, J.A., Selva, J., Rodríguez, J.A., Saiz de Omeñaca, J.A., 2006. The influence of mycorrhization with *Tuber melanosporum* in the afforestation of a Mediterranean site with *Quercus ilex* and *Quercus faginea*. For. Ecol. Manag. 231, 226–233. https://doi.org/10.1016/j.foreco.2006.05.052.

Esteso-Martínez, J., Gil Pelegrín, E., 2004. Frost resistance of seeds in Mediterranean oaks and the role of litter in the thermal protection of acorns. Ann. For. Sci. 61, 481–486. https://doi.org/10.1051/forest.

Fernández-Galiano, E., Mauri, P.V., Garcia, G., 1997. Somatic embryogenesis induction on *Quercus faginea* Lam. Acta Hortic. 447, 143–145. https://doi.org/10.17660/ACTAHORTIC.1997.447.21.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal de la Comunidad Valenciana. Conselleria de Medi Ambient, Generalitat Valenciana, Alboraya (Valencia).

Gómez-Aparicio, L., Zamora, R., Gómez, J.M., Hódar, J., Castro, J., Baraza, E., 2004. Applying plant facilitation to forest restoration: a meta-analysis of the use of shrubs as nurse plants. Ecol. Appl. 14, 1128–1138. https://doi.org/10.1890/03-5084.

González-Rodríguez, V., Villar, R., Navarro-Cerrillo, R.M., 2011. Maternal influences on seed mass effect and initial seedling growth in four *Quercus* species. Acta Oecol. 37, 1–9. https://doi.org/10.1016/j.actao.2010.10.006.

Granados, M.E.E., Vilagrosa, A., Chirino, E., Vallejo, V.R.R., 2016. Reforestation with resprouter species to increase diversity and resilience in Mediterranean pine forests. For. Ecol. Manag. 362, 231–240. https://doi.org/10.1016/j.foreco.2015.12.020.

Grossnickle, S.C., El-Kassaby, Y.A., 2016. Bareroot versus container stocktypes: a performance comparison. N. For. 47, 1–51. https://doi.org/10.1007/s11056-015-9476-6.

Löf, M., Castro, J., Engman, M., Leverkus, A.B., Madsen, P., Reque, J.A., Villalobos, A., Gardiner, E.S., 2019. Direct seeding to restore oak (*Quercus* spp.) forests and woodlands. For. Ecol. Manag. 48, 474–489. https://doi.org/10.1016/j.foreco.2019.06.032.

Mediavilla, S., Escudero, A., 2003. Stomatal responses to drought at a Mediterranean site: a comparative study of co-occurring woody species differing in leaf longevity. Tree Physiol. 23, 987–996. https://doi.org/10.1093/treephys/23.14.987.

Medina Blanco, M., 1956. Contribución al estudio del área de la encina en la provincia de Córdoba y de sus posibilidades alimenticias para el ganado. Arch. Zootec. 5, 101–104.

Milla, R., Castro-Díez, P., Montserrat-Martí, G., 2010. Phenology of Mediterranean woody plants from NE Spain: synchrony, seasonality, and relationships among phenophases. Flora 205, 190–199. https://doi.org/10.1016/j.flora.2009.01.006.

Morcillo, L., Turrión, D., Soliveres, S., Chirino, E., Vallejo, V.R., Vilagrosa, A., 2022. Moderate pine cover maximizes 10-year survival and growth in late-successional species of contrasting functional strategies. For. Ecol. Manag. 509, 120098. https://doi.org/10.1016/J.FORECO.2022.120098.

Navarro Cerrillo, R.M., Gálvez Ramírez, C., 2001. Manual para la identificación y reproducción de semillas de especies vegetales autóctonas de Andalucía. In: Consejería de Medio Ambiente. Junta de Andalucía, Sevilla.

Nicolás Peragón, J.L., Villar-Salvador, P., Peñuelas Rubira, J.L., 2004. Efecto de la edad de la planta y el tipo de preparación del suelo en la supervivencia y crecimiento de *Quercus faginea* Lam. cultivado en contenedor. Cuadernos de la Sociedad Española de Ciencias Forestales 17, 205–209.

Oliet, J., Navarro Cerrillo, R.M., Contreras Atalaya, O., 2003. Evaluación de la aplicación de tubos y mejoradores en repoblaciones forestales. In: Dirección General de Gestión del Medio Natural, Junta de Andalucía, Córdoba.

Pemán, J., Chirino, E., Espelta, J.M., Jacobs, D.F., Martín-GómezRafael, P., Navarro-Cerrillo, R., Oliet, J. A., Vilagrosa, A., Villar-Salvador, P., Gil-Pelegrín, E., 2017. Physiological keys for natural and artificial regeneration of oaks. In: Gil-Pelegrín, E., Peguero-Pina, J.J., Sancho-Knapik, D. (Eds.), Oaks Physiological Ecology. Exploring the Functional Diversity of Genus *Quercus* L. Springer, Cham, pp. 453–511. https://doi.org/10.1007/978-3-319-69099-5_14.

Pemán, J., Peguero-Pina, J.J., Valladares, F., Gil-Pelegrín, E., 2010. Evaluation of unventilated treeshelters in the context of Mediterranean climate: insights from a study on *Quercus faginea* seedlings assessed with a 3D architectural plant model. Ecol. Eng. 36, 517–526. https://doi.org/10.1016/j.ecoleng.2009.11.021.

Peñuelas Rubira, J.L., 2022. La domesticación de los *Quercus* xerofíticos con vistas a su potencial frutícola. Foresta 84, 34–41.

Piñuela, Y., Alday, J.G., Oliach, D., Bolaño, F., Colinas, C., Bonet, J.A., 2020. Use of inoculator bacteria to promote *Tuber melanosporum* root colonization and growth on *Quercus faginea* saplings. Forests 11 (8), 79. https://doi.org/10.3390/F11080792.

Piotto, B., di Noi, A., 2003. Seed propagation of mediterranean trees and shrubs. In: APAT - Agency for the Protection of the Environment and for Technical Services, Roma.

Rey Benayas, J.M., Navarro, J., Espigares, T., Nicolau, J.M., Zavala, M.A., 2005. Effects of artificial shading and weed mowing in reforestation of Mediterranean abandoned cropland with contrasting *Quercus* species. For. Ecol. Manag. 212, 302–314. https://doi.org/10.1016/j.foreco.2005.03.032.

Reyna Domenech, S., 2012. Truficultura: Fundamentos Y Técnicas, second ed. Mundi-Prensa, Madrid.

Rolo, V., Plieninger, T., Moreno, G., 2013. Facilitation of holm oak recruitment through two contrasted shrubs species in Mediterranean grazed woodlands. J. Veg. Sci. 24, 344–355. https://doi.org/10.1111/j.1654-1103.2012.01458.x.

Sánchez Humanes, B., 2011. Acorn Production and Growth in Mediterranean Oaks: Trade-Offs and the Role of Climate, Modular Organization and Phenology. Universitat Autònoma de Barcelona, Barcelona, Spain.

Tsakaldimi, M., Zagas, T., Tsitsoni, T., Ganatsas, P., 2005. Root morphology, stem growth and field performance of seedlings of two Mediterranean evergreen oak species raised in different container types. Plant Soil 278, 85–93. https://doi.org/10.1007/s11104-005-2580-1.

Villar-Salvador, P., Oliet Palá, J.A., Heredia-Guerrero, N., Uscola, M., Goicoetxea, P., 2013a. *Quercus faginea* Lam. y *Quercus humilis* mill. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo Parques Nacionales. Serie Forestal, pp. 206–225.

Villar-Salvador, P., Peñuelas, J.L., Nicolás-Peragón, J.L., Benito, L.F., Domínguez-Lerena, S., 2013b. Is nitrogen fertilization in the nursery a suitable tool for enhancing the performance of Mediterranean oak plantations? N. For. 44, 733–751. https://doi.org/10.1007/s11056-013-9374-8.

Quercus frainetto Ten.

Aytekin Ertas
İstanbul University-Cerrahpasa, Faculty of Forestry, Department of Silviculture, Istanbul, Türkiye

Propagation

Seed propagation

Quercus frainetto Ten. (Hungarian oak) acorns are oblong, egg-shaped (2–2.5 cm), glabrous, with big curly cups. Acorns need one growing season to grow and are dispersed in the first half of October. The peduncle is very short (<2 mm) or even absent with groupings of 2–7 acorns. The cup (involucre) is hemispherical, protecting a third to half of the acorn, scales are oblong, pubescent, appressed, and loosely imbricate (Bordács et al., 2019).

Acorn size shows strong population differences. For instance, in Türkiye the fresh weight of 1000 acorns can vary from 3375 kg, with a mean diameter and length of 1.34 and 28 cm, respectively in the provenance of Kuşadası in southern Türkiye, to 1794 kg with a mean diameter and length of 1.3 and 1.6 cm in the provenance of Demirköy in northern Türkiye (Tigli, 2009).

The acorns of *Q. frainetto*, like those of other white oak species, are recalcitrant, requiring special conservation to prevent significant moisture loss before use. Critical moisture levels that significantly inhibit germination for this species are currently unknown, but they are likely similar to those reported for *Q. ilex* (Villar-Salvador et al., 2013). For processing, handling, and storage of acorns, we recommend following the same guidelines outlined in the *Q. ilex* section of this compendium.

Quercus frainetto acorns usually have a high germination rate if they are kept hydrated. Pretreatment of acorns for germination is unnecessary yet immersing them in water for 24 h can synchronize their germination times. Table 11.51 presents the average characteristics of *Q. frainetto* acorn lots.

Under natural conditions, acorns germinate early in the fall when soil moisture is sufficient and temperatures range from 6.2 to 16.3°C. By November, 33% of viable acorns have germinated, with root lengths of 0.5–10 cm, increasing to 92% by December, with radicle lengths of 0.5–22 cm. Root growth and germination pause during winter but resume with spring rains. Water stress during early radicle growth causes 70% embryo

Table 11.51 Characteristics of seed lots of *Quercus frainetto*.

Purity (%)	No. seeds/kg	Germination capacity (%)
94–100 (97)	296–557 (426)	64–90 (77)

Mean values in brackets.
Tigli (2009).

mortality in dry soils. Thick litter can also hinder root penetration, leading to horizontal radicle elongation (Bercea, 2013).

Seedling emergence begins when soil temperature exceeds 10°C, provided moisture is adequate. By late May, seedlings typically reach 11 cm in height with a root collar diameter of 3 mm. Most seedlings develop a single shoot, though 0.1% produce two shoots from a single root, and a second flush occurs in only 0.3% of cases (Bercea, 2013).

Vegetative propagation

Somatic embryogenesis can be used for vegetative propagation in oaks and immature zygotic embryos from fully developed acorns can be successfully used for this purpose in *Q. frainetto* (Timofte et al., 2011).

Seedling cultivation

The areas covered by *Q. frainetto* forests were reduced significantly in the last century due to anthropogenic influences, which may have had a negative effect on their genetic diversity. Natural regeneration of *Q. frainetto* is difficult and slow. Currently, it is mainly regenerated vegetatively (coppicing) which, in addition, contributes to the reduction in genetic diversity (Popović et al., 2021).

Q. frainetto seedlings can be grown as both container and bareroot stock. However, container stock is recommended due to the higher survival in dry environments (Grossnickle and El-Kassaby, 2016) where this oak is frequently planted. In 1 + 0 bareroot seedling propagation, successful development was not achieved when pregerminated radicles longer than 2 cm were clipped, as radicle removal inhibited shoot formation (Aslan, pers. observations).

Container plants

One-year-old container seedlings (Fig. 11.37) can be cultivated in polyethylene bags (5 cm diameter, 20–30 cm depth, 2.85 L volume). Such bag size prevents root spiraling and promotes proper root development. The recommended growing medium consists of a 1:1:1 (by volume) mixture of peat, top forest soil, and well-decomposed farmyard manure, ensuring adequate aeration, water retention, and nutrient availability. Fertilization should follow a site-specific NPK supplementation program, based on the nutrient composition of the growing medium. Regular irrigation and manual weeding are necessary to maintain optimal growth. Radoglou et al. (2003) recommended using plastic containers, 4 cm in diameter and 15 cm in depth (190 mL volume), using a 3:1 peat-to-perlite growing medium for 2 + 0 container seedlings. The aforementioned 1-year-old seedling production using bags can also be effectively applied to 2-year-old seedlings.

Table 11.52 Morphological attributes of 1 + 0 and 2 + 0 bareroot and container seedlings of *Quercus frainetto*.

	Bareroot 1 + 0		Bareroot 2 + 0		Container 1 + 0	
Attributes	**Observed**	**Recommended**	**Observed**	**Recommended**	**Observed**	**Recommended**
Shoot height (cm)	12.1	10–15	30.8	25–30	13.9	10–15
Root collar diameter (cm)	0.61	0.5–0.8	0.9	1.0–1.5	0.49	0.5–0.8
Shoot dry weight (g)	0.7	0.7–1.0	8.0	2–3	0.8	0.7–1.0
Root dry weight (g)	3.0	>3.0	3.8	4–6	4.3	>4.0
Plant dry weight (g)	3.7	>4.0	11.8	10–12	5.0	>5.0
Shoot/root dry weight ratio	0.23	0.2–0.3	2.1	0.5	0.19	0.2–0.3
Sturdiness quotient SH/RCD	2.0	2.0–2.5	3.4	2.0–2.5	2.8	2.0–2.5
Dickson quality index	1.66	1.5–2.0	2.4	4.0	1.65	1.5–2.0

Ozder et al., (2022); Yucedag et al. (2019).

Bareroot plants

Bareroot seedlings (Fig. 11.37) are often preferred for large-scale, cost-sensitive projects in regions where conditions support their establishment. They are an excellent choice for climates with a predictable dormant season on well-prepared planting sites. However, container seedlings might be more suitable in harsher conditions or where flexibility in planting schedules is needed. The mean root collar diameter of 1-year-old bareroot seedlings is 3.7 mm, the average height of the seedlings is 11.4 cm, and the mean vigor index of the seedlings is 3.1 (Popović et al., 2021). In the study by Yucedag et al. (2019), which examined the effects of presowing phytohormone treatments on seedling growth, the application of 300 ppm IBA for 24 h to *Q. frainetto* acorns resulted in significant improvements in seedling growth. Reduction in shoot-to-root ratio and sturdiness quotient and increase in the Dickson Quality Index is usually related to increased outplanting performance. According seedling morphology (Table 11.52), 1 + 0 bareroot seedlings are preferred over 2 + 0 seedlings in forest plantings, especially on dry sites or with high weed competition (Fig. 11.37).

Field establishment

Quercus frainetto seedlings show higher tolerance to water stress and higher field survival compared with the other species with which it coexists such as *Acer pseudoplatanus* and *Castanea sativa* (Raftoyannis et al., 2006). Competition control involves mechanical clearing or selective herbicide application, followed by mulching and periodic thinning



Figure 11.37 *Quercus frainetto* seedlings in the nursery. Left: 1 + 0 bareroot plants. Right: 1 + 0 container seedlings. *(Aytekin Ertas.)*

to enhance resource allocation. Herbivore protection is essential, as young seedlings are highly susceptible to browsing by deer and rodents. Effective measures include tree shelters, fencing, repellents, and promoting natural predators.

Irrigation is particularly important during the early years, with deep watering every 1–2 weeks in dry periods. Mulching further improves moisture retention. Optimal planting seasons are autumn (September–November) that allows root establishment before winter, or early spring (February–April) in colder regions.

Chapter References

Bercea, I., 2013. Germination, upshot and growth of Hungarian and Turkey oak seedlings in the woodlands of the western part of the Getic Plateau. Oltenia 29, 145–150.

Bordács, S., Zhelev, P., Schirone, B., 2019. EUFORGEN Technical Guidelines for Genetic Conservation and Use for Hungarian Oak (*Quercus frainetto*). European Forest Genetic Resources Programme (EUFORGEN). European Forest Institute, 6 pages. ISBN 13, 978-952-5980-61-5.

Grossnickle, S.C., El-Kassaby, Y.A., 2016. Bareroot versus container stocktypes: a performance comparison. N. For. 2016 47, 1–51. https://doi.org/10.1007/s11056-015-9476-6.

Ozder, H., Tolunay, D., Çelik, O., Ozyürek, E., 2022. Use of Hungarian oak (*Quercus frainetto* Ten.) seedlings grown in different growing media in reforestation projects of inner Thrace. In: Marmara Forestry Research Institute Directorate (Nursery Phase) interim project report, Project No. 10.1303/2018-2025 (Unpublished).

Popović, V., Lučić, A., Rakonjac, L., Jovanović, S., Lazarević, I., 2021. Variability of Hungarian oak (*Quercus frainetto* Ten.) from the territory of Lipovica according to morphological traits of seedlings. Sustain. Forest. 83–84, 27–36.

Radoglou, K., Raftoyannis, Y., Halivopoulos, G., 2003. The effects of planting date and seedling quality on field performance of *Castanea sativa* Mill. and *Quercus frainetto* Ten. seedlings. Forestry 76, 569–578. https://doi.org/10.1093/forestry/76.5.569.

Raftoyannis, Y., Radoglou, K., Halivopoulos, G., 2006. Ecophysiology and survival of *Acer pseudoplatanus* L., *Castanea sativa* Miller. and *Quercus frainetto* Ten. seedlings on a reforestation site in northern Greece. N. For. 31, 151–163.

Tigli, T., 2009. Seed and Germination Characteristics in Some Hungarian Oak (*Quercus frainetto* Ten.) Provenances in Turkey. Istanbul University Institute of Science Master's Thesis, 42pp.

Timofte, A.I., Pamfil, D., Palada-nicolau, M., Timofte, C.S., 2011. The Effect of Different Factors upon the in Vitro Propagation in *Quercus Robur* and *Q. Frainetto* by Somatic Embryogenesis Notulae Botanicae Horti Agrobotanici Cluj-Napoca, vol 39, pp. 288–291. https://doi.org/10.15835/nbha3915481.

Villar-Salvador, P., Nicolás-Peragón, J.L., Heredia-Guerrero, N., Uscola, M., 2013. Quercus ilex. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales, vol II. Organismo Autónomo Parques Nacionales, Madrid, Spain, pp. 226–250.

Yucedag, C., Bilir, N., Ozel, H.B., 2019. Phytohormone effect on seedling quality in Hungarian oak. Forest Syst. 28, 1–7. https://doi.org/10.5424/fs/2019282-14604.

Quercus ilex L.

Pedro Villar-Salvador[1], Juan Luis Nicolás Peragón[2], Juan A. Oliet[3] and Marianthi Tsakaldimi[4]
[1]Universidad de Alcalá, Forest Ecology and Restoration Group, Departamento de Ciencias de la Vida, Alcalá de Henares, Madrid, Spain; [2]Centro Nacional de Recursos Genéticos Forestales "El Serranillo", MITECO, Guadalajara, Spain; [3]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [4]Laboratory of Silviculture, Department of Forestry and Natural Environment, Aristotle University of Thessaloniki, Thessaloniki, Greece

Propagation

Seed propagation

The maturation of *Quercus ilex* L. (holm oak) acorns occurs from mid-October to December, depending on weather conditions. Under optimal conditions, sexual maturity is attained at the age of 8–10 years and regular crops start occurring when individuals reach 15 to 20-year-old, with maximum acorn production between 50 and 100-year-old (Ruiz de la Torre, 2006). It is a masting species that typically has high acorn yields every 2–3 years in agroforestry systems (Carbonero-Muñoz, 2011).

Acorns can be harvested from late October, mostly in November, collected directly from the ground after their natural fall or by spreading nets or tarps under the tree canopy and shaking tree branches. This maximizes profitability and reduces contamination with soil microbes. Collection is done by hand or with rakes and occasionally machines are used. The first acorns to fall should be avoided since they are often damaged or underdeveloped. Under typical natural conditions, *Q. ilex* acorns take 20 days to reach moisture content levels that make them unviable (Vázquez Pardo, 1998). Frost damage is frequent in acorns not protected by leaf litter or herbaceous cover; the first damage symptoms appear in the cupule scar. Oak seeds collected before they naturally ripen cannot be artificially matured (Bonner, 1979). We have observed, however, that *Q. ilex* acorns collected about 10 days before their final maturation (while they still show a greenish to yellowish color) have high germination potential (Villar-Salvador, unpublished data).

Acorn collection yield is 40–100 kg per day and worker, with a yield per tree of 15–60 kg in highly productive trees (Nicolás-Peragón, pers. comm.). Acorns transported from the field to processing sites must be in containers that prevent moisture loss but also avoid their overheating. The cupules, and other debris such as leaves and twigs, must be removed by sieving, winnowing, and flotation in water. The latter operation also allows for the partial removal of damaged or parasitized acorns. We recommend leaving the acorns in water for 16–24 h to raise their moisture content and check if they sink. This avoids discarding healthy acorns that float because they have slightly dehydrated before being collected, during temporary storage, or in transportation. A small amount of bleach can be added to the water (1%) to disinfect acorns, but we recommend leaving acorns not more than 2 h in the bleach water and rinsing them twice with tap water.

Damaged acorns must often be removed manually. It is important to cut some acorns to support the decision of rejecting acorn lots. Classification of *Q. ilex* acorns by size is not a common practice. Exceptionally small acorns can be separated when removing debris and for commercial presentation purposes. Although acorn sizing can improve the germination and uniformity of produced seedlings (Bonner, 2008) it can potentially lead to genetic biases (Burgarella et al., 2007).

Quercus ilex propagates well by seed. The average characteristics of *Q. ilex* acorn lots are shown in Table 11.53. The length of the acorns usually ranges from 22–45 mm. The mean width varies from 9.9–16 mm, while the mean seed fresh weight can vary between 1.3 and 5.5 g with a mean value of 3.1 g (Bonito et al., 2011; Gómez, 2004; Spanos and Gaitanis, 2016).

As with other oak species, *Q. ilex* acorns are recalcitrant seeds, that is, their viability depends on maintaining a high moisture content. Due to high moisture levels, the respiration rate remains high. When the moisture content is < 32%–30%, viability is reduced by half, and when it reaches 20%, acorns are unviable (Aroca et al., 2005; Joët et al., 2013; Villar-Salvador et al., 2013; Zulueta and Montoto, 1992). Small acorns lose viability due to faster desiccation than big acorns (Peñuelas-Rubira, 2001). Short-to-medium-term acorn conservation should avoid drying out the acorns while allowing for acorn respiration and preventing fermentation.

Storage must not be done in airtight containers and the temperature should be kept between −4 and 3°C. This temperature prevents cold damage and radicle growth, which would hinder subsequent acorn sowing. When stored only for the winter, the temperature can be set between 0 and 3°C. For longer periods, it should be between −4 and −1°C, but not at lower temperatures because *Q. ilex* acorns are severely damaged when frozen at −9°C (Esteso-Martínez and Gil Pelegrín, 2004). It is important to store acorns with their surface dry to avoid rotting and germination. To prevent rotting, we recommend treating acorns with a fungicide rather than applying thermal shock treatments, which are commonly used with deciduous oaks in central Europe and other parts of the world (Lacroix, 1990; Muller, 1990).

Insects that parasitize acorns can be eliminated by immersion in hot water at 41°C and left to cool for 180 min (Nicolás-Peragón, unpublished data) or with insecticides. However, we do not recommend the heat treatment due to the risk of damage to the acorns. Weevil larvae usually do not hinder germination or seedling growth if the

Table 11.53 Characteristics of seed lots of *Quercus ilex*.

Purity (%)	No. seeds/kg	Germination capacity (%)
94–100 (97)	182–500 (294)	73–91 (81)

Mean values in brackets.
Bonito et al. (2011); Catalán (1991); García-Fayos (2001); Gómez (2004); Gradi (1989); Louro and Pinto (2011); Navarro-Cerrillo and Gálvez (2001); Pemán et al. (2013); Piotto and Falleri (2001); Ribeiro et al. (2001); Spanos and Gaitanis (2016)

embryo has not been damaged, and cotyledons have only been partially damaged. Weevil larvae do not attack other unparasitized acorns during storage, and most will emerge from acorns during cold storage and die in the storage container (Nicolás-Peragón, unpublished data).

Quercus ilex acorns can be preserved in two ways:

(1) *Storage in plastic bags*. Clean and surface-dry acorns can be stored in polyethylene bags with a wall thickness of 40–70 μm, sealed with a zip-lock or thermally, and arranged in perforated, stackable plastic boxes to allow for air circulation. Polyethylene plastic prevents moisture loss from the bag but allows some diffusion of O_2 and CO_2 to maintain acorn respiration (Bonner, 2008; Rink and Williams, 1984). Thicker polyethylene plastic bags usually result in the death of oak acorns.

(2) *Stratification in a substrate*. Acorns are placed between layers of a porous substrate or mixed with a substrate in a 1:1 volume ratio. The mixture can be placed in 30–50 L containers in which a perforated 10 cm diameter plastic tube is placed vertically in the middle and along the container. Finally, the container must be sealed with plastic, leaving the aeration tube protruding from the lid to facilitate inside aeration (Villar-Salvador et al., 2013). The substrate can be peat, sand, sawdust, vermiculite, or coconut fiber, and should be dry to absorb excess moisture released by the acorns and prevent their germination. The acorns should be separated from each other to reduce fungi spread. For storage longer than 1 year, acorn moisture content should be 4–8% lower than the maximum moisture level (Aroca et al., 2005).

Stored acorns must be periodically checked to assess vigor (García-Fayos, 2001; Piotto and di Noi, 2003). If preserved properly, small batches of *Q. ilex* acorns for research purposes can be stored for up to 2–3 years without significantly hindering vigor (Nicolás-Peragón, unpublished data). However, at operational scale, long-term conservation (i.e., beyond the winter for sowing next spring) has not been considered due to the intrinsic difficulties of storing large quantities of acorns.

Acorns should be germinated as soon as possible after removal from storage to avoid viability loss. *Quercus ilex* acorns do not require any germination pretreatment as they show no or very moderate dormancy, since the time required for germination and emergence is reduced with the duration of cold storage. Immersing the acorns in water for 24 h helps to homogenize germination (Villar-Salvador et al., 2013). As some of the acorns can germinate during storage, handling acorns with radicles longer than 2 cm must be done carefully, because breaking the radicle at the insertion point to the acorn strongly inhibits shoot emergence (Nicolás-Peragón, pers. observations). To the best of our knowledge, no studies have addressed the impact of medium- and long-term storage of acorns on the quality of their seedlings.

Vegetative propagation

There is little experience with vegetative propagation of *Q. ilex*. However, rooting of stem segments has been obtained both in vitro and ex vitro with the aid of IBA solutions. Mother plant age had major influence on the percentage of rooting of the cuttings as the younger plants showed greater ability to root (Liñán et al., 2011). Recent research demonstrates the possibility of grafting *Q. ilex* twigs on rootstock of *Q. ilex*, *Q. faginea*, *Q. suber* and *Q. suber* × *Q. ilex* hybrids for the early production of highly productive acorn trees (Peñuelas Rubira, 2022). Similarly, it is possible to produce somatic embryos from tissues from mature *Q. ilex* oak trees (Barra-Jiménez et al., 2014).

Seedling cultivation

Quercus ilex seedlings can be grown as container and bareroot stock. Currently almost all the production of this species in Europe is container stock. We recommend container production given the greater survival of container stock in dry environments, the usual environment for establishing plantations of this species (Grossnickle and El-Kassaby, 2016; Tsakaldimi et al., 2005). Therefore, the cultivation information on *Q. ilex* will focus only on container production.

Container plants

Nursery sowing can be carried out in autumn, immediately after the acorn harvest, or at the end of winter and beginning of spring. Autumn sowing should be avoided for outdoor cultivation in locations where winter frosts are strong and prolonged and spring frosts are common. Autumn cultivation should start in a greenhouse. To ensure that all container cavities or cells are occupied, acorns can be pregerminated in trays with moist growing medium. Once germinated, acorns with radicles no longer than 2 cm can be transferred to the final cultivation containers. This is especially recommended when using batches with low germination. If the length of the radicle is > 5 cm, it can be cut up to 3 cm to facilitate sowing, without negative consequences for the seedling (Piotto and di Noi, 2003). Seedlings with excised radicles will produce multiple taproots rather than one taproot. Only one acorn should be sown per container cell, preferably placed horizontally and no more than 3 cm deep. Seedling emergence occurs 0.5–2 months under greenhouse conditions (Villar-Salvador et al., 2013) and 40 days outdoors under cool winter conditions (Tsakaldimi et al., 2011).

We recommend cultivating *Q. ilex* in containers of 300–400 mL. Large-volume, especially deep containers usually increases the success of field establishment of oak species (Chirino et al., 2008; Domínguez-Lerena, 1997; Jelić et al., 2016; Pemán et al., 2006, 2017; Tsakaldimi et al., 2005). The most common substrates are light and black peat, and mixtures of coconut fiber with peat, either fertilized or not (Mariotti et al.,

2020). We recommend adding vermiculite, up to 20% of the total volume, to facilitate organic growing medium hydration during irrigation and to improve cation exchange capacity when using coconut fiber (Landis et al., 2014). Due to the relatively small leaves and absence of lateral twigs in one-year-old seedlings, cultivation density for *Q. ilex* may be 300–400 plants/m^2.

The lack or low fertilization of *Q. ilex* seedlings during nursery cultivation increases their mortality and notably reduces their growth capacity in the field (Cortina et al., 2009; Villar-Salvador et al., 2004a). Most of the N demand of *Q. ilex* seedlings during early growth is met by the acorn, while little N is taken up from the growing medium (i.e., supplied by the fertilizer) from germination to the end of the first flush of shoot growth (Villar-Salvador et al., 2010). Therefore, to increase fertilization efficiency, we recommend, if possible, to supply fertilizer in the irrigation water rather than using fertilized growing medium and to postpone fertilization until most seedlings are in the second flush of shoot growth. The highest outplanting survival and growth rates are achieved when seedlings are fertilized with a total of 100–150 mg N/plant, distributed over 16–20 weeks, supplied weekly (Oliet et al., 2009; Uscola et al., 2015), while higher rates can reduce hardening (Heredia-Guerrero et al., 2014). Fertilization at an exponential or a constant rate does not make a great difference in growth and nutrient status (Oliet et al., 2009). However, fertilization from 1 to 2 months in the nursery in the fall results in seedling nutrient loading, increasing root growth potential and postplanting drought resistance, while not compromising hardening (Heredia-Guerrero et al., 2014; Oliet et al., 2011). Similarly, maintaining moderate autumn fertilization enhances cold acclimation compared with plants whose fertilization is ceased at the end of summer (Andivia et al., 2011). Foliar fertilization by spraying 40 mM of urea has been shown as an effective method to increase tissue N concentration in *Q. ilex* seedlings (Uscola et al., 2014).

Quercus ilex seedlings can be cultivated under full sun if the substrate remains hydrated. However, in locations with very hot summers, seedlings can benefit from shading during the summer months, reducing incident light by no more than 60%. This protects the plants from excessive sunlight and extreme temperatures at mid-day while it reduces irrigation needs (Puértolas et al., 2009). Moderate shade (up to 60% irradiation reduction) has little effect on the nursery growth and hardening of *Q. ilex* seedlings and subsequent field establishment (Puértolas et al., 2009; Villar-Salvador et al., 2004a).

Mycorrhization of *Q. ilex* in the nursery with *Tuber melanosporum* increases field survival and growth (Domínguez Núñez et al., 2006). Currently, the cultivation of *Q. ilex* mycorrhized with truffles of the genus *Tuber*, predominantly *T. melanosporum* is quite common. The production of such plants requires rigorous control of inoculation to ensure the identity of the initial fungal material and to verify the correct maturity and quality of the carpophores. The fungal material can be used fresh or dried in slices at around 40°C for 2–3 days and stored in cold conditions until use, in which case it

must be rehydrated before inoculation. Although various inoculation methods exist, the most recommended is injection, which involves preparing an aqueous suspension by gently blending the carpophores and incorporating the chosen dose with a syringe into one or more depths in the plug of each plant after transplanting (Reyna Domenech, 2012). Generally, the minimum recommended dose is 1 g of fresh truffle per plant. It is common to use soil from natural truffle grounds as a substrate, but the practice is discouraged due to negative effects on plant quality and the impact on truffle grounds caused by soil extraction.

Nursery location affects hardening and seedling quality. *Quercus ilex* seedlings cultivated in mild winter locations such as those near the Mediterranean coast, show reduced frost and drought tolerance during the fall and early spring, and lower new root growth capacity (Fig. 11.38) and field growth than plants cultivated in a location with cold winters (Heredia-Guerrero et al., 2014; Mollá et al., 2006). Drought hardening during the last phase of cultivation increases plant resistance to drought (Villar-Salvador et al., 2004b), but there is no clear evidence that it increases field survival and growth (Villar-Salvador unpublished results). Because hardening irrigation is a nursery practice difficult to implement, this cultivation practice is not recommended for *Q. ilex* except in nurseries located in mild winter areas to stop seedling growth in the mid-fall (Vilagrosa, pers. comm.).

Observed and recommended morphological and physiological characteristics of container *Q. ilex* stock are shown in Table 11.54, separated by the two subspecies of *Q. ilex*. According to our nursery experience and published literature, *Q. ilex* subsp. *ilex* grows faster than *Q. ilex* subsp. *ballota*.

A herringbone root system with an elongated taproot should be the optimal root architecture for *Q. ilex* seedlings to survive under Mediterranean conditions. However, container *Q. ilex* seedlings typically develop one thick taproot, while growth of secondary roots is much smaller (Fig. 11.38). Containers restrict taproot length resulting in a thick taproot with many higher order lateral roots (Tsakaldimi et al., 2009). Shoot branching in 1-year seedlings is rare, but not in two-year-old and older seedlings.

Seedling size at the end of nursery cultivation is strongly and positively related to acorn size, due to the increased amount of nutrients and carbohydrates available for the initial growth of the seedling (Navarro et al., 2006). Field survival and growth of one-year-old *Q. ilex* seedlings increases with shoot and root size at planting (Cortina et al., 1997; Cuesta et al., 2010a, 2010b; Tsakaldimi et al., 2012; Villar-Salvador et al., 2004a). Soil preparation and planting date have greater impact on the outplanting performance of *Q. ilex* seedlings than seedling quality. However, the importance of seedling quality becomes more relevant when combined with early and intermediate plantings dates and strong, deep soil preparation (Palacios et al., 2009).

Table 11.54 Morphological and physiological attributes recommended for 1 + 0 high quality *Q. ilex* container seedlings grown outdoors, with separate information on observed values for the two subspecies, *Q. ilex* subsp. *ballota* (most Iberian Peninsula and north Africa) and *Q. ilex* subsp. *ilex* (rest of the northern Mediterranean basin).

Attributes	Observed values (min-max)		Recommended values
	subsp. ilex	subsp. ballota	
Shoot height (cm)	12–42	11–24	>20
Root collar diameter (cm)	0.31–0.85	0.27–0.76	>0.5
Shoot dry weight (g)	0.9–8.5	1.1–5.1	>3
Root dry weight (g)	0.2–6.7	1.1–4.8	>4
Shoot/root dry weight ratio	0.59–0.77	0.34–1.27	≈0.7–0.8
DQI (Dickson's quality index)	0.85–1.41	0.81–2.76	>1.5
Shoot [N] (mg/g)	7.7–8.3	7.5–15.0	>13
Shoot [P] (mg/g)	0.48–0.57	0.30–1.3	>0.5
Shoot [K] (mg/g)	3.4–4.9	2.8–6.1	>5.5
Root [N] (mg/g)	3.7–5.3	5.0–12.5	>9
Root [P] (mg/g)	0.48–1.5	0.19–2.0	>0.9
Root [K] (mg/g)	2.62–5.5	6.5–8.5	>7

Andivia et al. (2012); Cuesta et al. (2010a); Cuesta et al. (2010b); de Castro et al. (2014); del Campo et al. (2022); del Campo et al. (2010); Heredia-Guerrero et al. (2014); Jelić et al. (2016); Mariotti et al. (2020); Mollá et al. (2006); Navarro et al. (2024); Oliet et al. (2009); Oliet et al. (2011); Palacios et al. (2009); Puértolas et al. (2009); Rodríguez-García et al. (2003); Tsakaldimi (2001); Tsakaldimi et al. (2005); Villar-Salvador et al. (2004a); Villar-Salvador et al. (2004b).

Field establishment

Due to its anisohydric water use behavior and lower control over leaf water potential (Gil-Pelegrín et al., 2017; Mediavilla and Escudero, 2004) and the development of a deep root system, field establishment of *Q. ilex* should be prioritized to deep soils with clay-loam texture in dry locations to ensure long-term viability, especially in the context of increasing aridity linked to climate change (Gavinet et al., 2016). Plantations on marls show higher survival than plantations on limestones (Serrasoles and Alloza, 2004) indicating the importance of soil texture.

Quercus ilex can be established by planting or sowing. Unfortunately, few comparative studies exist on the performance of both methods in *Q. ilex* to make solid recommendations. Some evidence shows greater survival of planted seedlings than sown seedlings (González-Rodríguez et al., 2011; Romero Esteban et al., 2005; Seva et al., 2004), while other workers have not observed differences in survival between the two reforestation methods (Nicolás et al., 1997; Oliet et al., 2015). Survival of seeded acorns can be strongly increased if acorns are germinated before sowing and protected with a tree shelter (Oliet et al., 2015; Seva et al., 2004). Besides, seedlings derived from acorns sown in the field have less root deformation than container seedlings (Löf

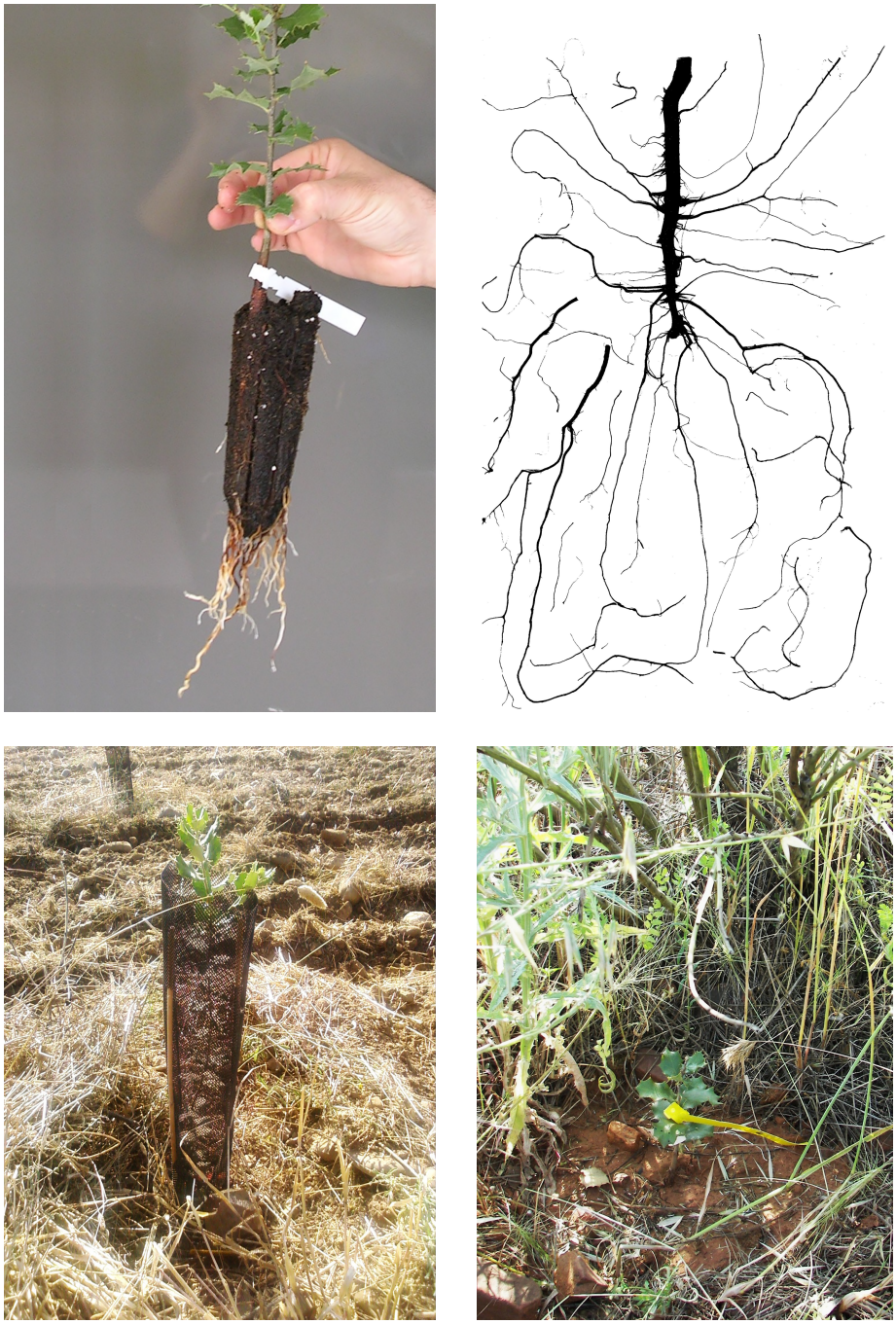


Figure 11.38 Upper left: Production of new roots after a root growth capacity test; upper right: Root structure of a 1 + 0 *Quercus ilex* container seedling; lower left: Protected seedling protruding out of a 60% light transmission mesh-type tree shelter 1 year after planting; lower right: 1 + 0 container seedling planted under the shade of a retama shrub and with weeds removed. *(Juan A. Oliet, Marianthi Tsakaldimi, and Pedro Villar-Salvador.)*

et al., 2019), but the implications of these root structure differences for field performance of *Q. ilex* are unknown as far as we know. If acorn sowing is chosen, we recommend using large acorns (Navarro et al., 2006, 2023), placed deep in the soil using the method of Carreras Egaña et al. (1997) and protected from predators with tree or seed shelters (Castro et al., 2015; Oliet et al., 2015).

We recommend using 1 + 0 plants for plantations (Fig. 11.38). In southern Spain 1 + 0 seedlings performed better than 3 + 0 saplings (González-Rodríguez et al., 2011), while better performance of 2 + 0 seedlings than 1 + 0 seedlings has also been reported (Siles et al., 2010). If seedlings older than 1 + 0 are used, it is important to cultivate plants in containers of at least 300 mL.

In dry climate zones, intense, deep soil preparation facilitates deep root growth and enhanced outplanting performance (Bocio et al., 2004; Löf et al., 2012; Nicolás et al., 1997; Palacios et al., 2009). We recommend subsoiling and mechanical hole digging, mainly with a backhoe, with depths >40 cm. In semiarid, south-facing slopes, the construction of micro basins associated with the planting holes significantly increases *Q. ilex* outplanting performance (Bocio et al., 2004; de Navarrete-Simón et al., 2006).

Experimental plantations of *Q. ilex* have shown that planting seedlings under shrubs (Fig. 11.38) can increase establishment compared with planting in open spaces by mitigating high irradiation and air temperature, increasing soil fertility, and protecting against large herbivores (Cuesta et al., 2010b; Gavinet et al., 2016; Gómez-Aparicio et al., 2005; Rolo et al., 2013). This facilitative effect of shrubs depends on the shrub species, with legume species showing the highest performance. The disadvantage of shrub underplanting is that it limits intense soil preparation, as it can damage nurse shrubs. This reduces plantation productivity because establishment is more complex and survival rates are frequently moderate to low. Therefore, we recommend restricting this technique to sites where intense soil preparation and field irrigation cannot be performed, high final plant densities are not the aim, or the use of tree or tube shelters is not authorized.



Figure 11.39 *Quercus ilex* afforestation in Quintos de Mora (Center of Spain) on an abandoned cropland and pastureland. *(Juan A. Oliet.)*

Underplanting *Q. ilex*, a resprouter tree, under the canopy of planted forests such as pine plantations has been suggested as a tool to promote species diversification and resilience of Mediterranean pine plantations (Puerta-Piñero et al., 2012). The canopy of these plantations can be more effective in facilitating *Q. ilex* than do shrubs (Mendoza et al., 2009). The most effective canopy openness of pine plantations to facilitate *Q. ilex* underplanted seedlings response is site specific. Seedling survival decreases under high-density pine stands on semiarid sites, while moderate to low density stands increases survival on mild and wet sites (Gavinet et al., 2015; Granados et al., 2016; Morcillo et al., 2022).

The establishment of planted *Q. ilex* seedlings is tightly linked to the production of an abundant system of new roots (Fig. 11.38) prior the beginning of summer (Villar-Salvador et al., 2012). In cold winter sites, planting in early November and at the beginning of winter did not show greater root production at the end of spring than planting in late winter (mid-February). In contrast, planting in early spring (mid-March) showed a comparatively significant reduction in new root growth (Corchero de la Torre et al., 2002). Therefore, fall planting in cold areas does not seem to have a greater advantage in terms of root production than planting in late winter. This is likely because *Q. ilex* shows little root elongation when the soil temperature $<10°C$. In brief, in mild winter sites, planting should preferably be carried out in the fall as soon as soils rewet after summer drought (Navarro et al., 2023; Palacios et al., 2009), while on cold winter sites (elevation >900–1000 m asl), they can be postponed to mid-February but not beyond mid-March. These suggestions will likely need to be revisited in the future due to increased winter temperature linked to climate change. If seeding is the establishment method, we recommend acorn seeding to be done in the fall and keeping the acorns in water for 12–24 h prior to sowing.

Quercus ilex frequently shows high mortality in forestation. Irrigating and shading in the field increase *Q. ilex* establishment (Puértolas et al., 2010; Rey Benayas et al., 2005). Shading can be achieved using tree shelters (Fig. 11.38), which have a strong positive effect on outplanting performance (De Castro et al., 2014; Navarro Cerrillo et al., 2021; Nicolás et al., 1997; Oliet et al., 2003). Shelters should be ventilated and have moderate light transmissivity, especially in areas with high water stress. Tree shelters also protect seedlings from herbivory. The length of the tube will depend on the type of herbivores on planting sites. If the predominant herbivores are rabbits and hares, 60–70 cm long tubes work well (Navarro-Cerillo et al., 2021). We recommend keeping the tubes until the plant stems have thickened and the plants can stand upright by themselves after elongating out of the tree shelter. In most cases, this can take 5–8 years depending on how fast plants grow.

Field irrigation strongly reduces mortality, especially on sites that are remarkable dry and hot in summer or in years with a dry spring. We suggest watering 10–20 L/plant, applied three times throughout the first summer after planting or seeding. Field

fertilization is not recommended for *Q. ilex* reforestation, as it does not seem to improve plant development (Oliet et al., 2003). Likewise, the use of composted urban solid waste and sewage sludge has been shown to reduce survival (Fuentes et al., 2007; Jiménez et al., 2007). It is preferable to develop a suitable nursery fertilization program and outplant seedlings with high tissue nutrient content.

In cropland afforestation (Fig. 11.39), herbs strongly inhibit *Q. ilex* performance (Cuesta et al., 2010a; Navarro et al., 2023; Rey Benayas et al., 2005). Therefore, herbs 1 m around oak seedlings must be removed during the first 2–3 years after planting to minimize competition, which can be accomplished by weeding, herbicides, or mulching (Coello et al., 2018; Coello and Piqué, 2016; Navarro Cerrillo et al., 2005; Navarro Cerrillo et al., 2021). Weed removal should be accomplished in the middle of spring.

Chapter References

Andivia, E., Fernández, M., Vázquez-Piqué, J., 2011. Autumn fertilization of *Quercus ilex* ssp. *ballota* (Desf.) Samp. nursery seedlings: effects on morpho-physiology and field performance. Ann. For. Sci. 68, 543–553. https://doi.org/10.1007/s13595-011-0048-4.

Andivia, E., Fernández, M., Vázquez-Piqué, J., Alejano, R., 2012. Two provenances of *Quercus ilex* ssp. *ballota* (Desf) Samp. nursery seedlings have different response to frost tolerance and autumn fertilization. Eur. J. For. Res. 131, 1091–1101. https://doi.org/10.1007/s10342-011-0578-1.

Aroca, M.J., Serrada, R., Calderón Guerrero, C., 2005. Influencia del contenido de humedad y del tiempo de almacenaje sobre la capacidad germinativa en bellotas de encina (*Quercus ilex* subsp. *ballota* L.) sometidas a congelación. In: Resultados preliminares, Actas del IV Congreso Forestal Español. p. 5. Sociedad Española de Ciencias Forestales.

Barra-Jiménez, A., Blasco, M., Ruiz-Galea, M., Celestino, C., Alegre, J., Arrillaga, I., Toribio, M., 2014. Cloning mature holm oak trees by somatic embryogenesis. Trees Struct. Funct. 28, 657–667. https://doi.org/10.1007/S00468-014-0979-0/TABLES/4.

Bocio, I., Navarro, F.B., Ripoll, M.A., Jiménez, M.N., de Simón, E., 2004. Holm oak (*Quercus rotundifolia* Lam.) and Aleppo pine (*Pinus halepensis* Mill.) response to different soil preparation techniques applied to forestation in abandoned farmland. Ann. For. Sci. 61, 171–178. https://doi.org/10.1051/forest.

Bonito, A., Varone, L., Gratani, L., 2011. Relationship between acorn size and seedling morphological and physiological traits of *Quercus ilex* L. from different climates. Photosynthetica 49, 75–86. https://doi.org/10.1007/S11099-011-0014-2/METRICS.

Bonner, F.T., 2008. *Quercus*. In: Bonner, F.T., Karrfalt, R.P. (Eds.), Woody Plant Seed Manual. USDA Forest Service, vol 727. Agriculture Handbook, pp. 928–996.

Bonner, F.T., 1979. Fruit maturation in hardwoods. In: Seed Collection Workshop. USDA Forest Service, Macon, GA, pp. 6–10.

Burgarella, C., Navascues, M., Soto, Á., Lora, Á., Fici, S., Soto, A., Lora, A., Fici, S., 2007. Narrow genetic base in forest restoration with holm oak (*Quercus ilex* L.) in Sicily. Ann. For. Sci. 64, 757–763. https://doi.org/10.1051/forest:2007055.

Carbonero-Muñoz, M.D., 2011. Evaluación de la producción y composición de la bellota de encina en dehesas (PhD). In: Universidad de Córdoba, Córdoba, Spain.

Carreras Egaña, C., Sánchez Hoyos, J., Reche Pérez, P., Herrero Hinojo, D., Navarro Encinas, E., Navío Oliver, J.J., 1997. Siembras profundas con ayuda de tubos protectores. Resultados en ensayos comparativos de siembras y plantaciones bajo condiciones áridas en Vélez-Rubio. In: Actas del II Congreso Forestal Español. SECF, Pamplona, pp. 123–128.

Castro, J., Leverkus, A.B., Fuster, F., 2015. A new device to foster oak forest restoration via seed sowing. N. For. 46, 919–929. https://doi.org/10.1007/S11056-015-9478-4/TABLES/2.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: ICONA Madrid. 4a Ed, 4th Ed. Icona, Madrid, Spain.

Chirino, E., Vilagrosa, a., Hernández, E.I.I., Matos, a., Vallejo, V.R.R., 2008. Effects of a deep container on morpho-functional characteristics and root colonization in *Quercus suber* L. seedlings for reforestation in Mediterranean climate. For. Ecol. Manag. 256, 779–785. https://doi.org/10.1016/j.foreco.2008.05.035.

Coello, J., Piqué, M., 2016. Soil Conditioners and Groundcovers for Sustainable and Cost-Efficient Tree Planting in Europe and the Mediterranean. Technical Guide. Solsona, Spain.

Coello, J., Ameztegui, A., Rovira, P., Fuentes, C., Piqué, M., 2018. Innovative soil conditioners and mulches for forest restoration in semiarid conditions in northeast Spain. Ecol. Eng. 118, 52–65. https://doi.org/10.1016/J.ECOLENG.2018.04.015.

Corchero de la Torre, S., Gozalo-Cano, M., Villar-Salvador, P., Peñuelas-Rubira, J.L., 2002. Crecimiento radical en campo de *Pinus halepensis* y *Quercus ilex* plantados en diferentes momentos. Rev. Mont. 68, 5–11.

Cortina, J., Monerris, J., Disante, K., Fuentes, D., Valdecantos, A., 2009. Fertilización tardía y establecimiento de *Quercus ilex* subsp. *ballota*, Actas del V Congreso Forestal Español. SECF, Ávila, pp. 1–11.

Cortina, J., Valdecantos, A., Seva, J.P., Vilagrosa, A., Bellot, J., Vallejo, V.R., 1997. Relación tamaño-supervivencia en plantones de especies arbustivas y arbóreas mediterráneas producidas en vivero, Actas del II Congreso Forestal Español (IRATI 97). Gobierno de Navarra, Pamplona, pp. 22–27.

Cuesta, B., Villar-Salvador, P., Puértolas, J., Jacobs, D.F., Rey Benayas, J.M., 2010a. Why do large, nitrogen rich seedlings better resist stressful transplanting conditions? A physiological analysis in two functionally contrasting Mediterranean forest species. Forest Ecolol. Manage. 260, 71–78. https://doi.org/10.1016/j.foreco.2010.04.002.

Cuesta, B., Villar-Salvador, P., Puértolas, J., Rey Benayas, J.M., Michalet, R., 2010b. Facilitation of *Quercus ilex* in Mediterranean shrubland is explained by both direct and indirect interactions mediated by herbs. J. Ecol. 98, 687–696. https://doi.org/10.1111/j.1365-2745.2010.01655.x.

de Castro, A.V., Oliet, J.A., Puértolas, J., Jacobs, D.F., 2014. Light transmissivity of tube shelters affects root growth and biomass allocation of *Quercus ilex* L. and *Pinus halepensis* Mill. Ann. For. Sci. 71, 91–99. https://doi.org/10.1007/s13595-013-0335-3.

de Simón-Navarrete, E., Ripoll-Morales, M., Fernández-Ondoño, E., Navarro-Reyes, F.B., Jiménez-Morales, M.N., Gallego-Teruel, E., 2006. Eficacia de las microcuencas en la supervivencia del pino carrasco (*Pinus halepensis* Mill.) y de la encina (*Quercus ilex* L. subsp. *ballota* (Desf.) Samp.) en distintos ambientes mediterráneos. Invest Agrarias: Sistemas y Recursos Forestales 15, 218–230.

del Campo, A.D., Navarro, R.M., Ceacero, C.J., 2010. Seedling quality and field performance of commercial stocklots of containerized holm oak (*Quercus ilex*) in Mediterranean Spain: an approach for establishing a quality standard. N. For. 39, 17–37.

del Campo, A.D., Segura-Orenga, G., Molina, A.J., González-Sanchis, M., Reyna, S., Hermoso, J., Ceacero, C.J., 2022. On the need to further refine stock quality specifications to improve reforestation under climatic extremes. Forests 13, 168. https://doi.org/10.3390/f13020168.

Domínguez Núñez, J.A., Serrano, J.S., Barreal, J.A.R., González, J.A.S.D.O., Domínguez, J.A., Selva, J., Rodríguez, J.A., Saiz de Omeñaca, J.A., 2006. The influence of mycorrhization with Tuber melanosporum in the afforestation of a Mediterranean site with *Quercus ilex* and *Quercus faginea*. For. Ecol. Manag. 231, 226–233. https://doi.org/10.1016/j.foreco.2006.05.052.

Domínguez-Lerena, S., 1997. La importancia del envase en la producción de plantas forestales. Quercus 134, 34–37.

Esteso-Martínez, J., Gil Pelegrín, E., 2004. Frost resistance of seeds in Mediterranean oaks and the role of litter in the thermal protection of acorns. Ann. For. Sci. 61, 481–486. https://doi.org/10.1051/forest.

Fuentes, D., Valdecantos, A., Cortina, J., Vallejo, V.R., 2007. Seedling performance in sewage sludge-amended degraded Mediterranean woodlands. Ecol. Eng. 31, 281–291. https://doi.org/10.1016/j.ecoleng.2007.09.001.

García-Fayos, P., 2001. Bases ecológicas para la recolección, almacenamiento y germinación de semillas de especies de uso forestal de la Comunidad Valenciana. In: Conselleria de Medi Ambient, Generalitat Valenciana, Alboraya (Valencia).

Gavinet, J., Prévosto, B., Fernandez, C., 2016. Do shrubs facilitate oak seedling establishment in Mediterranean pine forest understory? Forest Ecolol. Manage. 381, 289–296. https://doi.org/10.1016/j.foreco.2016.09.045.

Gavinet, J., Vilagrosa, A., Chirino, E., Granados, M.E., Vallejo, V.R., Prévosto, B., 2015. Hardwood seedling establishment below Aleppo pine depends on thinning intensity in two Mediterranean sites. Ann. For. Sci. 72, 999–1008. https://doi.org/10.1007/s13595-015-0495-4.

Gil-Pelegrín, E., Saz, M.Á., Cuadrat, J.M., Peguero-Pina, J., Sancho-Knapik, D., 2017. Oaks physiological ecology. In: Exploring the Functional Diversity of Genus *Quercus* L. Springer.

Gómez, J.M., 2004. Bigger is not always better: conflicting selective pressures on seed size in *Quercus ilex*. Evolution 58, 71–80. https://doi.org/10.1111/j.0014-3820.2004.tb01574.x.

Gómez-Aparicio, L., Gómez, J.M., Zamora, R., Boettinger, J.L., 2005. Canopy vs. soil effects of shrubs facilitating tree seedlings in Mediterranean montane ecosystems. J. Veg. Sci. 16, 191–198.

González-Rodríguez, V., Navarro-Cerrillo, R.M., Villar, R., 2011. Artificial regeneration with *Quercus ilex* L. and *Quercus suber* L. by direct seeding and planting in southern Spain. Ann. For. Sci. 68, 637–646. https://doi.org/10.1007/s13595-011-0057-3.

Gradi, A., 1989. Vivaistica Forestale. Edagricole, Bologna, Italia.

Granados, M.E.E., Vilagrosa, A., Chirino, E., Vallejo, V.R.R., 2016. Reforestation with resprouter species to increase diversity and resilience in Mediterranean pine forests. Forest Ecolol. Manage. 362, 231–240. https://doi.org/10.1016/j.foreco.2015.12.020.

Grossnickle, S.C., El-Kassaby, Y.A., 2016. Bareroot versus container stocktypes: a performance comparison. N. For. 47, 1–51. https://doi.org/10.1007/s11056-015-9476-6.

Heredia-Guerrero, N., Oliet, J.A., Villar-Salvador, P., Benito, L.F., Peñuelas, J.L., 2014. Fertilization regime interacts with fall temperature in the nursery to determine the frost and drought tolerance of the Mediterranean oak *Quercus ilex* subsp. *ballota*. Forest Ecolol. Manage. 331, 50–59. https://doi.org/10.1016/j.foreco.2014.07.022.

Jelić, G., Topić, V., Butorac, L., Jazbec, A., Oršanić, M., Đurđević, Z., 2016. The impact of the container size and soil preparation on afforestation success of one year old holm Oak (*Quercus ilex*) seedlings in Croatian mediterranean area. Period. Biol. 117, 493–503. https://doi.org/10.18054/pb.2015.117.4.3532.

Jiménez, M.N., Fernández-Ondoño, E., Ripoll, M.A., Navarro, F.B., Gallego, E., De Simón, E., Lallena, A.M., 2007. Influence of different post-planting treatments on the development in Holm oak afforestation. Trees Struct. Funct. 21, 443–455. https://doi.org/10.1007/s00468-007-0136-0.

Joët, T., Ourcival, J.M., Dussert, S., 2013. Ecological significance of seed desiccation sensitivity in *Quercus ilex*. Ann. Bot. 111, 693–701. https://doi.org/10.1093/aob/mct025.

Lacroix, P., 1990. La conservation des glands à l'échelle industrielle à l'office national des forêts en 1989. Rev. For. Fr. 42, 215–219.

Landis, T.D., Jacobs, D.F., Wilkinson, K.M., Luna, T., 2014. Growing media, in: Tropical nursery manual-A guide to starting and operating a nursery for native and traditional plants. USDA forest service. Agricult. Handb. 732, 101–122.

Liñán, J., Cantos, M., Troncoso, J., García, J.L., Fernández, A., Troncoso, A., 2011. Some propagation methods for cloning holm oak (*Quercus ilex* L.) plants. Cent. Eur. J. Biol. 6, 359–364. https://doi.org/10.2478/S11535-011-0007-Y/METRICS.

Löf, M., Castro, J., Engman, M., Leverkus, A.B., Madsen, P., Reque, J.A., Villalobos, A., Gardiner, E.S., 2019. Direct seeding to restore oak (*Quercus* spp.) forests and woodlands. For. Ecol. Manag. https://doi.org/10.1016/j.foreco.2019.06.032.

Löf, M., Dey, D.C., Navarro, R.M., Jacobs, D.F., 2012. Mechanical site preparation for forest restoration. N. For. 43, 825–848. https://doi.org/10.1007/s11056-012-9332-x.

Louro, V., Pinto, G., 2011. Sementes, uma ponte entre o passado e o futuro da floresta. In: Ministério da Agricultura, Mar, Ambiente e Ordenamento do Territorio. CENASEF.

Mariotti, B., Martini, S., Raddi, S., Tani, A., Jacobs, D.F., Oliet, J.A., Maltoni, A., 2020. Coconut coir as a sustainable nursery growing media for seedling production of the ecologically diverse *Quercus* species 2020. Forests 11, 522. https://doi.org/10.3390/F11050522.

Mediavilla, S., Escudero, A., 2004. Stomatal responses to drought of mature trees and seedlings of two co-occurring Mediterranean oaks. For. Ecol. Manag. 187, 281–294.
Mendoza, I., Zamora, R., Castro, J., 2009. A seeding experiment for testing tree-community recruitment under variable environments: implications for forest regeneration and conservation in Mediterranean habitats. Biol. Conserv. 142, 1491–1499.
Mollá, S., Villar-Salvador, P., García-Fayos, P., Peñuelas Rubira, J.L.J.L., 2006. Physiological and transplanting performance of *Quercus ilex* L. (holm oak) seedlings grown in nurseries with different winter conditions. For. Ecol. Manag. 237, 218–226. https://doi.org/10.1016/j.foreco.2006.09.047.
Morcillo, L., Turrión, D., Soliveres, S., Chirino, E., Vallejo, V.R., Vilagrosa, A., 2022. Moderate pine cover maximizes 10-year survival and growth in late-successional species of contrasting functional strategies. For. Ecol. Manag. 509, 120098. https://doi.org/10.1016/J.FORECO.2022.120098.
Muller, C., 1990. Problèmes posés par la conservation des glands. Rev. For. Fr. 42, 212–214. https://doi.org/10.4267/2042/26064ï.
Navarro-Cerrillo, R.M., Gálvez, C., 2001. In: Costa Pérez, J.C. (Ed.), Manual para la identificación y reproducción de semillas de especies autóctonas de Andalucía. Junta de Andalucía, Sevilla, Spain, pp. 292–296.
Navarro Cerrillo, R.M., Ceacero Ruiz, C.J., Coello, J., del Campo García, A., 2021. Control de la competencia en repoblaciones forestales. In: Pemán García, J., Navarro Cerrillo, R.M., Prada Sáez, M.A., Serrada Hierro, R. (Eds.), Bases Técnicas y Ecológicas Del Proyecto de Repoblación Forestal. Ministerio para la Transición Ecológica y el Reto Demográfico (MITECO), Madrid, Spain, pp. 884–935.
Navarro Cerrillo, R.M., del Campo García, A., Pemán García, J., Ceacero Ruiz, C.J., 2021. Métodos de protección de la repoblación forestal. In: Pemán García, J., Navarro Cerrillo, R.M., Prada Saez, M.A., Serrada Hierro, R. (Eds.), Bases técnicas y ecológicas del proyecto de repoblación forestal. Ministerio para la Transición Ecológica y el Reto Demográfico (MITECO), Madrid, Spain, pp. 823–883.
Navarro Cerrillo, R.M., Fragueiro, B., Ceaceros, C., del Campo, A., De Prado, R., 2005. Establishment of *Quercus ilex* L. subsp. ballota [Desf.] Samp. using different weed control strategies in southern Spain. Ecol. Eng. 25, 332–342. https://doi.org/10.1016/j.ecoleng.2005.06.002.
Navarro, F.B., Jiménez, N., Ripoll, M.A., Fernández-Ondoño, E., Gallego, E., De Simón, E., 2006. Direct sowing of holm oak acorns: effects of acorn size and soil treatment. Ann. For. Sci. 63, 961–967. https://doi.org/10.1051/forest:2006079.
Navarro, F.B., Caño, A.B., Gálvez, C., Kazani, A., Carbonero, M.D., Jiménez, M.N., 2023. Key factors in direct acorn seeding for the successful restoration of open oak woodlands. For. Ecol. Manag. 546, 121314. https://doi.org/10.1016/J.FORECO.2023.121314.
Navarro, F.B., Caño, A.B., Gálvez, C.R., Kazani, A., Carbonero, M.D., Ripoll, M.Á., Jiménez, M.N., 2024. Interaction between weeding, outplanting date and site in open oak woodland plantations. For. Ecol. Manag. 554, 121649. https://doi.org/10.1016/J.FORECO.2023.121649.
Nicolás, J.L., Domínguez-Lerena, S., Herrero-Sierra, N., Villar-Salvador, P., 1997. Plantación y siembra de *Quercus ilex* L. Efectos de la preparación del terreno y de la utilización de protectores en la supervivencia de plantas, Actas del II Congreso Forestal Español. Irati 97, Pamplona, Spain. In: Sociedad Española de Ciencias Forestales-Gobierno de Navarra, pp. 449–454.
Oliet, J., Navarro Cerrillo, R.M., Contreras Atalaya, O., 2003. Evaluación de la aplicación de tubos y mejoradores en repoblaciones forestales. In: Dirección General de Gestión del Medio Natural, Junta de Andalucía, Córdoba, Spain.
Oliet, J.A., Tejada, M., Salifu, K.F., Collazos, A., Jacobs, D.F., 2009. Performance and nutrient dynamics of holm oak (*Quercus ilex* L.) seedlings in relation to nursery nutrient loading and post-transplant fertility. Eur. J. For. Res. 128, 253–263. https://doi.org/10.1007/s10342-009-0261-y.
Oliet, J.A., Salazar, J.M., Villar, R., Robredo, E., Valladares, F., 2011. Fall fertilization of Holm oak affects N and P dynamics, root growth potential, and post-planting phenology and growth. Ann. For. Sci. 68, 647–656. https://doi.org/10.1007/s13595-011-0060-8.
Oliet, J.A., Vázquez de Castro, A., Puértolas, J., 2015. Establishing *Quercus ilex* under Mediterranean dry conditions: sowing recalcitrant acorns versus planting seedlings at different depths and tube shelter light transmissions. N. For. 46, 869–883. https://doi.org/10.1007/S11056-015-9495-3/METRICS.

Palacios, G., Navarro Cerrillo, R.M., del Campo, A., Toral, M., 2009. Site preparation, stock quality and planting date effect on early establishment of Holm oak (*Quercus ilex* L.) seedlings. Ecol. Eng. 35, 38–46. https://doi.org/10.1016/j.ecoleng.2008.09.006.

Pemán, J., Chirino, E., Espelta, J.M., Jacobs, D.F., Martín-GómezRafael, P., Navarro-Cerrillo, R., Oliet, J. A., Vilagrosa, A., Villar-Salvador, P., Gil-Pelegrín, E., 2017. Physiological keys for natural and artificial regeneration of oaks. In: Gil-Pelegrín, E., Peguero-Pina, J.J., Sancho-Knapik, D. (Eds.), Oaks Physiological Ecology. Exploring the Functional Diversity of Genus *Quercus* L. Springer, Cham, pp. 453–511. https://doi.org/10.1007/978-3-319-69099-5_14.

Pemán, J., Navarro-Cerrillo, R., Nicolás, J.L., Prada, M.A., Serrada, R., 2013. Producción y Manejo de Semillas y Plantas Forestales. Volumen I and II. In: Organismo Autónomo Parques Nacionales. Serie Forestal, Madrid, Spain.

Pemán, J., Voltas, J., Gil-Pelegrín, E., 2006. Morphological and functional variability in the root system of *Quercus ilex* L. subject to confinement: consequences for afforestation. Ann. For. Sci. 63, 425–430.

Peñuelas-Rubira, J.L., 2001. El Centro Nacional de Mejora "El Serranillo": diez años buscando la calidad de la planta forestal para las actuaciones en ámbito mediterráneo. In: Actas del III Congreso Forestal Español "Sierra Nevada 2001", Sociedad Española de Ciencias Forestales, Granada, Spain.

Peñuelas Rubira, J.L., 2022. La domesticación de los *Quercus* xerofíticos con vistas a su potencial frutícola. @RevForesta 84, 35–41.

Piotto, B., di Noi, A., 2003. Seed propagation of mediterranean trees and shrubs. In: APAT - Agency for the Protection of the Environment and for Technical Services, Roma.

Piotto, B., Falleri, E., 2001. La conservazione dei semi. In: Piotto, B., Di Noi, A. (Eds.), Propagazione per Seme Di Alberi e Arbusti Della Flora Mediterranea. ANPA, Roma, Italia.

Puerta-Piñero, C., Brotons, L., Coll, L., González-Olabarría, J.R., 2012. Valuing acorn dispersal and resprouting capacity ecological functions to ensure Mediterranean forest resilience after fire. Eur. J. For. Res. 131, 835–844. https://doi.org/10.1007/S10342-011-0557-6/TABLES/3.

Puértolas, J., Benito, L.F., Peñuelas, J.L., 2009. Effects of nursery shading on seedling quality and post-planting performance in two Mediterranean species with contrasting shade tolerance. N. For. 38, 295–308. https://doi.org/10.1007/s11056-009-9148-5.

Puértolas, J., Oliet, J.a., Jacobs, D.F., Benito, L.F., Peñuelas, J.L., 2010. Is light the key factor for success of tube shelters in forest restoration plantings under Mediterranean climates? For. Ecol. Manag. 260, 610–617. https://doi.org/10.1016/j.foreco.2010.05.017.

Rey Benayas, J.M., Navarro, J., Espigares, T., Nicolau, J.M., Zavala, M.A., 2005. Effects of artificial shading and weed mowing in reforestation of Mediterranean abandoned cropland with contrasting *Quercus* species. For. Ecol. Manag. 212, 302–314.

Reyna Domenech, S., 2012. Truficultura: Fundamentos Y Técnicas, second ed. Mundi-Prensa, Madrid.

Ribeiro, D., Ribeiro, H., Louro, V., 2001. Produção Em Viveiros Florestais. Direcção-Geral de Desenvolvimento Rural, Lisboa.

Rink, G., Williams, R.D., 1984. Storage technique affects white oak acorn viability. Tree Plant. Notes 35, 3–5.

Rodríguez-García, B., Villar-Salvador, P., SerraDA, R., 2003. Estudio del momento óptimo de plantación de brinzales de *Quercus ilex* y *Pinus halepensis* en función del clima de la estación de plantación. In: EUIT Forestal. Madrid.

Rolo, V., Plieninger, T., Moreno, G., 2013. Facilitation of holm oak recruitment through two contrasted shrubs species in Mediterranean grazed woodlands. J. Veg. Sci. 24, 344–355. https://doi.org/10.1111/j.1654-1103.2012.01458.x.

Romero Esteban, A.M., Aramburu Maqua, M.P., Heranz Ortega, J., 2005. Estudio comparado de la repoblación forestal mediante la técnica de "Siembra profunda superficial" frente a otras de plantación y siembra, Actas del IV Congreso Forestal Español. SECF, Zaragoza, Spain.

Ruiz de la Torre, J., 2006. Flora mayor. Organismo Autónomo Parques Nacionales. Ministerio de Medio Ambiente y Medio Rural y Marino, Madrid, Spain.

Serrasoles, I., Alloza, J.A., 2004. Condicionantes edáficos en la restauración forestal mediterránea. In: Vallejo, V.R., Alloza, J.A. (Eds.), Avances en el Estudio de la gestión del monte mediterráneo. Fundación CEAM, Valencia, pp. 133–160.

Seva, J.P., Valdecantos, A., Cortina, J., Vallejo, R., 2004. Diferentes técnicas de introducción de *Quercus ilex* ssp. *ballota* (Desf.) Samp. en zonas degradadas de la Comunidad Valenciana. Cuadernos de la Sociedad Española de Ciencias Forestales 17, 233–238.

Siles, G., Rey, P.J., Alcántara, J.M., Bastida, J.M., Herreros, J.L., 2010. Effects of soil enrichment, watering and seedling age on establishment of Mediterranean woody species. Acta Oecol. 36, 357–364.

Spanos, K., Gaitanis, D., 2016. Genetic diversity of seed characteristics of *Quercus ilex* L. and relationship with biometric parameters of the descendants (in Greek). In: Proceedings of 16th Scientific Conference of Hellenic Genetic Plant Breeding Society. Florina, Greece, pp. 16–22.

Tsakaldimi, M., 2001. Research on the Production and Quality Assessment of the Container-Planting Stock Used in the Afforestations (PhD). Faculty of Forestry and Natural Environment, Aristotle University of Greece, Thessaloniki.

Tsakaldimi, M., Zagas, T., Tsitsoni, T., Ganatsas, P., 2005. Root morphology, stem growth and field performance of seedlings of two Mediterranean evergreen oak species raised in different container types. Plant Soil 278, 85–93.

Tsakaldimi, M., Tsitsoni, T., Ganatsas, P., Zagas, T., 2009. A comparison of root architecture and shoot morphology between naturally regenerated and container-grown seedlings of *Quercus ilex*. Plant Soil 324, 103–113. https://doi.org/10.1007/S11104-009-9974-4/FIGURES/5.

Tsakaldimi, M., Papadopoulou, M., Prodofikas, C., Chatzichristaki, C., Tsitsoni, T., 2011. Germination capacity and rate of five Mediterranean wood species sown outdoors. In: 15th Scientific Conference of Hellenic Forestry Society, Karditsa, Greece.

Tsakaldimi, M., Ganatsas, P., Jacobs, D.F., 2012. Prediction of planted seedling survival of five Mediterranean species based on initial seedling morphology. N. For. 44, 327–339.

Uscola, M., Villar-Salvador, P., Oliet, J., Warren, C.R., 2014. Foliar absorption and root translocation of nitrogen from different chemical forms in seedlings of two Mediterranean trees. Environ. Exp. Bot. 104, 34–43. https://doi.org/10.1016/j.envexpbot.2014.03.004.

Uscola, M., Salifu, K.F., Oliet, J.A., Jacobs, D.F., 2015. An exponential fertilization dose-response model to promote restoration of the Mediterranean oak *Quercus ilex*. N. For. 46, 795–812. https://doi.org/10.1007/s11056-015-9493-5.

Vázquez Pardo, F.M., 1998. Semillas del género *Quercus*. Biología, Ecología y Manejo. Consejería de Agricultura y Comercio. Junta de Extremadura. Mérida, Extremadura, Spain.

Villar-Salvador, P., Planelles, R., Enríquez, E., Rubira, J.P., 2004a. Nursery cultivation regimes, plant functional attributes, and field performance relationships in the Mediterranean oak *Quercus ilex* L. For. Ecol. Manag. 196, 257–266. https://doi.org/10.1016/j.foreco.2004.02.061.

Villar-Salvador, P., Planelles, R., Oliet, J., Peñuelas-Rubira, J.L., Jacobs, D.F., González, M., 2004b. Drought tolerance and transplanting performance of holm oak (*Quercus ilex*) seedlings after drought hardening in the nursery. Tree Physiol. 24, 1147–1155. https://doi.org/10.1093/treephys/24.10.1147.

Villar-Salvador, P., Heredia, N., Millard, P., 2010. Remobilization of acorn nitrogen for seedling growth in holm oak (*Quercus ilex*), cultivated with contrasting nutrient availability. Tree Physiol. 30, 257–263. https://doi.org/10.1093/treephys/tpp115.

Villar-Salvador, P., Puértolas, J., Cuesta, B., Peñuelas, J.L., Uscola, M., Heredia-Guerrero, N., Rey Benayas, J.M., 2012. Increase in size and nitrogen concentration enhances seedling survival in Mediterranean plantations. Insights from an ecophysiological conceptual model of plant survival. N. For. 43, 755–770. https://doi.org/10.1007/s11056-012-9328-6.

Villar-Salvador, P., Nicolás-Peragón, J.L., Heredia-Guerrero, N., Uscola, M., 2013. Quercus ilex. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo Parques Nacionales. Serie Forestal. Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, Spain, pp. 226–250.

Zulueta, J. de, Montoto, J.L., 1992. Efectos de la temperatura y humedad en la germinación de bellotas de encina (*Quercus ilex* L.) y alcornoque (*Quercus suber* L.). Investigación Agraria - Sistema y Recursos Forestales 1, 65–71.

Quercus petraea (Matt.) Liebl.

Valeriu-Norocel Nicolescu[1], Sanja Perić[2] and Martina Đodan[2]
[1]Faculty of Silviculture and Forest Engineering, Transilvania University of Braşov, Braşov, Romania; [2]Croatian Forest Research Institute, Department for Silviculture, Jastrebarsko, Croatia

Propagation

Seed propagation

Quercus petraea (sessile oak) is a monoecious species with unisexual flowers. Female flowers appear in groups of two or three on 2–7 mm long stalks. Male flowers form slender, pendulous catkins arising from buds next to the abscission scars of previous year's leaves or from terminal buds (Fletcher, 1992; Stănescu et al., 1997). Flowering coincides with early leaf development in April or May (Negulescu and Săvulescu, 1957, 1965; Stănescu, 1979; Stănescu et al., 1997; Fernández-López et al., 2013). Ripe acorns are barrel-shaped, 15–25 mm long, which occur in groups of one to five and begin dispersal in October, with a peak drop after a frost (Negulescu and Săvulescu, 1957, 1965; Fletcher, 1992; Florescu, 1996; Şofletea and Curtu, 2007).

During development, acorns are attacked by the knopper gall wasp (*Andricus quercusalicis*), which significantly reduces fecundity. In addition, acorns are frequently infested by weevils of the genus *Curculio* whose larvae consume part of the cotyledon and can damage the developing embryos, which can render acorns non-viable. Infestation rates by *Curculio* spp. vary from year to year but commonly exceed 20% of the crop (Gordon and Faulkner, 1992).

Q. petraea starts producing substantial acorn crops at ages between 40 and 50 years (Evans, 1988; Gordon and Faulkner, 1992; Kerr and Evans, 1993) to 60–70 (80) years (Negulescu and Săvulescu, 1957, 1965; Haralamb, 1967; Florescu, 1996; Şofletea and Curtu, 2007; Fernández-López et al., 2013). The interval between mast years ranges from 2 to 5 years (Evans, 1988; Kerr and Evans, 1993; Fernández-López et al., 2013) and 6–10 years (Solymos, 1993). Climate change is increasing the frequency of mast years (Pesendorfer et al., 2020).

Acorn yield per tree averages 20–40 kg in typical years and 70–90 kg during mast years. Yields range from 500 to 700 kg/ha in average years to as much as 3200 kg/ha in high-yield years (Gordon and Faulkner, 1992). Acorns are harvested manually from the ground between early October and early November (Gordon, 1992a; Gordon and Faulkner, 1992). To minimize postharvest cleaning, intact healthy acorns are individually collected. Mobile vacuum seed collectors have also been used effectively for cleaner collections (Gordon, 1992a). When bulk harvesting, acorns need to be cleaned of debris (leaves, twigs, cups), then soaked in water to remove floating, non-viable seeds. Viable acorns, which sink, are air-dried in the shade for 5–10 days with periodic mixing (Iancu, 1999).

Before storage, *Q. petraea* acorns should undergo phytosanitary thermotherapy against *Ciboria batschiana* (black rot). This treatment involves immersing acorns in water at 41°C for 2–3 h followed by surface drying until acorns reach a moisture content of 45%. The acorns are then treated with dry iprodione and stored (Fernández-López et al., 2013). Although costly, this treatment maintains germination rates >80% until sowing in April (Gordon, 1992c).

Acorn storage must ensure acorn respiration while preventing moisture loss or overheating. To achieve this, acorns should be stored in ventilated places at temperature close to 0–2°C (Vlase, 1982; Gosling and Aldhous, 1994), or as low as –3°C (Joyce et al., 1998). Like other oak species, *Q. petraea* acorns are recalcitrant, showing a maximum moisture content of 50% (Gordon, 1992b). During storage, moisture levels should be maintained between 40% and 48% (Vlase, 1982; Gordon, 1992b; Joyce et al., 1998). If these conditions are fulfilled, *Q. petraea* acorns can be stored for up to 2 years (Gordon, 1992c). If not, optimal germination is reduced to a maximum of 6 months (Haralamb, 1967; Florescu, 1996). Short-term storage from fall to spring can be done:

(a) In ventilated wide mesh sacks. This is the most effective method and involves storing acorns in wide mesh (loose-weave) sacks, positioned individually with ventilation on all sides to prevent piling (Gordon, 1992a; 1992c).

(b) Outdoor, in sand or trenches.

(c) In containers that preserve moisture but allow gas-exchange. Acorns are mixed with an inert substrate, typically slightly humid or dry peat. Gas exchange is facilitated by installing a perforated plastic tube placed vertically in the middle and along the container. The container must be sealed with plastic, leaving the aeration tube protruding from the lid to facilitate inside aeration.

(d) In polyethylene bags with 40–60 μm thick walls and without any substrate.

(e) On forest platforms covered with sand and litter.

(f) In cellars with wooden floors where acorns are spread in 15–25 cm thick layers, mixed with sand, and regularly mixed with a shovel—initially daily, then every 2–3 days—for the first 6–8 weeks or until the surface appears dry (Gosling and Aldhous, 1994). This process ensures uniform drying (Vlase, 1982; Gordon, 1992b; Iancu, 1999; Fernández-López et al., 2013).

The average characteristics of seed lots are summarized in Table 11.55 Acorns do not require any pregermination treatments.

Vegetative propagation

At present, *Q. petraea* is mainly propagated by seed. However, as indicated before, operational-scale acorn storage for more than 1 year is challenging. Therefore, vegetative propagation can be a reliable alternative during years of poor acorn production (Chalupa, 1993, 2005).

Table 11.55 Characteristics of seed lots of *Quercus petraea*.

Purity (%)	No. seeds/kg	Germination capacity (%)
85–100 (90)	250–475 (350)	60–90 (75)

Mean values in brackets.
Negulescu and Săvulescu (1957, 1965); Stănescu (1979); Vlase (1982); Gordon (1992b); Mason (1994); Florescu (1996); Stănescu et al., (1997); Iancu (1999); Şofletea and Curtu (2007).

Vegetative propagation of Q. *petraea* can be performed through stem cuttings or in vitro methods (Chalupa, 1993). For cuttings, juvenile leafy cuttings collected between May and July from young, hedged stock plants (6 years old), or plants from seedlings or up to 30-year-old trees (Chalupa, 1993). Although older genotypes may be preferred due to proven phenotypic traits, old stock plants show low rooting potential (Struve et al., 2010).

Leafy cuttings (10–20 cm long) can be soaked in a 200 mg/L indole-3-butyric acid (IBA) solution for 20–24 h or treated with a talc-based rooting powder (1% IBA + 10% benomyl). They are then inserted into a rooting medium of peat and perlite (1:1 or 1:1.5, v/v). Rooting must be performed under controlled conditions, either in growth cabinets with fog systems or in greenhouses with intermittent fog (Chalupa, 1993). The production cycle for plants from cuttings is approximately 1 year. Rooted cuttings are lifted in June of the following year, after shoot formation, and transplanted to the nursery. High rooting rates and well-developed root systems are observed in cuttings from young trees (one to three years old), while cuttings from older trees (9–30 years old) show poor rooting. Rooting success is enhanced under continuous light compared with natural day length conditions (Chalupa, 1993). Despite its advantages, the main limitation of vegetative propagation by cuttings is the cost. Plants derived from cuttings are 1.5–3 times more expensive than conventional seedlings. Therefore, the use of cuttings is economically justified only when the propagated material shows significant genetic superiority over unimproved seedlings (Mason and Jinks, 1994).

Q. *petraea* can also been propagated vegetatively by tissue culture, using axillary shoot multiplication and somatic embryogenesis. For the first method, explants can be obtained from shoots of 3-6-month-old seedlings, 6-year-old hedged trees, or stump sprouts from trees no more than 40 years old (Chalupa, 1993). Leaves should be removed and stems cut into shoot-tip and nodal segments (10–20 mm long). These segments must be transferred to a low-salt nutrient medium, such as broadleaved tree medium or woody plant medium (WPM) (Chalupa, 1984), supplemented with a low concentration of cytokinin (0.2–0.6 mg/L 6-benzylaminopurine, BAP). Elongation occurs within 4–5 weeks, after which plants can be transferred outdoors and grown under

partial shade for 2–3 months. Following hardening, plants can be moved to the field in early summer (Chalupa, 1993).

Somatic embryogenesis is an efficient method for propagating a large number of fast-growing, disease-resistant plants (Chalupa, 1993). It also enables genetic transformation and long-term conservation through cryopreservation (Fernández-López et al., 2013). The process involves the following steps: (1) Induction of embryonic tissue, zygotic embryos, or leaf and stem segments are cultured on modified Schenk-Hildebrandt and Murashige-Skoog media or WPM supplemented with cytokinin; (2) Maintenance of embryonic tissue; regular subculturing is performed to sustain the embryogenic cultures; (3) Maturation of somatic embryos; embryos develop in optimized conditions; (3) Germination and plantlet development; somatic embryos are germinated and the resulting plantlets are transferred to containers for hardening; (4) Field transfer; hardened plants are transplanted to the nursery (Chalupa, 1993, 2005). Around 12%–18% of embryogenic cultures result in germinating somatic embryos. However, the main limitation of *Q. petraea* regeneration through somatic embryogenesis is the low conversion rate of somatic embryos into viable plants.

In summary, successful clonal propagation of oaks requires juvenile tissue as the initial explant, as mature plus-trees of *Q. petraea* are challenging to propagate. Tissue-cultured plants exhibit similar vigor to conventionally propagated plants, and the field growth of micropropagated juvenile-origin oak trees is comparable to that of control seedlings (Chalupa, 1993).

Seedling cultivation

Container production of *Q. petraea* plants is carried out in different European countries, but currently most production in Europe is bareroot stock.

Container plants

Container volume is commonly 220–250 cm^3 but can reach 400 cm^3 and higher volumes for 1-3 (4)-year-old seedlings. Containers 15–18 cm deep allow vigorous taproot growth and provide good outplanting performance for large seeded, strong root species such as *Q. petraea*. Growing media can be a mixture of peat and vermiculite (80:20, v:v) or peat, vermiculite, and coconut fiber (40:40:20) (Fernández-López et al., 2013). These authors also recommend that growing density should not exceed 250 plants/m^2 due to the large size of the leaves of this oak. For the same reason containers must be watered regularly, especially during hot summer days.

Germination under outdoor conditions starts in mid-April. Seedlings aged 1 + 0 ready for planting reach 20–40 cm in height and a root collar diameter (RCD) of 3–5 mm (Orešković et al., 2006; Jokanović et al., 2024). For 3 + 0 container seedlings, height ranges from 50 to 80 cm with a minimum RCD of 7 mm, while 4 + 0 plants exceed 80 cm and have an RCD of 9 mm (CDAF, 2014).

Bareroot plants

Acorns can be sown in the fall if the soil has enough moisture, allowing germination to continue until the first frost. Autumn sowing is preferred due to its higher germination rate (73%–93%) compared with spring sowing (71%–84%) (Rubțov, 1958). Before sowing, acorns should be soaked in a 1% formalin solution for 5–10 min to protect against pathogens and then treated with a nitrogen-based fertilizer. Sowing is made in beds with rills at a depth of 7–8 cm. During winter, germinating acorns must be protected with an 8–10 cm layer of straw, leaves, or reeds (Haralamb, 1967; Vlase, 1982; Iancu, 1999).

Spring sowing is performed in March–April after soaking the acorns in water for 48 h to restore moisture content (Joyce et al., 1998). Sowing is done in beds 60 cm wide and 15 cm high suitable for mechanized cultivation, with acorns placed in rills 4–6 cm deep. In case of drought, the culture is covered with straw or leaves, which must be removed once germination occurs, usually after 25–30 days (Haralamb, 1967; Abrudan, 2006, 2010) but retained between rills in dry areas. Sowing density in the rills is 30–37 acorns/m, which requires approximately 4000 kg of acorns/ha. The optimum cultivation density is 20–30 plants/m (Iancu, 1999) or 350 plants/m^2 (Rubțov, 1958). The production cycle of *Q. petraea* bareroot seedlings in forest nurseries ranges from 2 years (1 + 1 stock type with a minimum RCD of 6 mm and a height of at least 20 cm) (Iancu, 1999; Abrudan, 2006) to 4 years (1 + 3 stock type with a minimum RCD of 5–9 mm and height of 35–80 cm) (Boudru, 1992; Rozporządzenie, 2004; CDAF, 2014). In general, transplants should have a minimum height of 20 cm and a minimum RCD of 5–5.5 mm (Aldhous, 1994a) (Fig. 11.40).

Following heavy rains, the soil crust formed in *Q. petraea* cultures should be broken promptly using mechanical cultivators. Hoeing at the beginning of the growing season to a depth of 5–8 cm is also recommended. Weed control in nurseries is typically



Figure 11.40 Production of bareroot *Quercus petraea* seedlings in the south of Romania. *(forestmania.ro.)*

achieved through herbicides using authorized residual substances (Willoughby et al., 2009) applied the day after sowing.

In acid soil nurseries, liming with $CaCO_3$ (equivalent to 1.4 t/ha CaO) stimulates fine root and shoot growth, enhancing the shoot-to-root ratio and foliar nutrition by improving soil fertility and reducing Al toxicity and acid stress (Bakker et al., 1999).

Irrigation should use water at a temperature similar to the soil temperature, followed by shallow cultivation. Typical irrigation schedules include 400 m^3/ha 5–7 days before sowing, 100–150 m^3/ha during germination, and 350 m^3/ha every 12 days (Iancu, 1999). Supplemental irrigation (30–90 m^3/ha) reduces the severity of oak powdery mildew (*Erysiphe/Microsphaera alphitoides*), which affects young leaves and non-lignified tissues, potentially killing seedlings (Strouts et al., 1994; Kasprzyk et al., 2022). Controlling irrigation rates can be part of an integrated protection strategy against this pathogen (Kasprzyk et al., 2022). Overcrowded *Q. petraea* crops should be thinned manually after rainfall or irrigation, 1–1.5 months after seedling emergence.

Mycorrhization can be promoted by spreading a thin layer of forest soil from an oak forest to increase initial height growth (Iancu, 1999). *Q. petraea* seedlings are susceptible to oak mildew, which can be controlled through sulfur-based treatments, removal and burning of leaves in the fall. In the nursery, the roots of seedlings are also affected by common cockchafer (*Melolontha melolontha*), European mole cricket (*Gryllotalpa gryllotalpa*), European mole (*Talpa europaea*) that covers the plants with their mounds, rodents such as mice (*Apodemus* spp.) that eat the acorns, or the European hare (*Lepus europaeus*) that gnaw seedling stems.

To promote a fibrous root system, the taproot, which can reach up to 1 m after the first growing season (Haralamb, 1967; Iancu 1999), even 2 m in youth (Joyce et al., 1998), should be undercut 20 cm below the root collar in the following spring (May), when seedlings have developed two leaves (Haralamb, 1967; Iancu, 1999; Abrudan, 2010). Damaged or overly long roots (exceeding 20–25 cm) should be pruned (Abrudan, 2006).

Oak plants can be over-wintered outdoors in trenches 30–40 cm deep, with bundles placed leaning against the wall facing the prevailing wind or the south. The roots and 4–5 cm of the stem are covered with an 8–10 cm layer of compacted soil to prevent air penetration. Seedlings should be watered as needed. Transportation to planting sites should be conducted under cool, moist conditions, with plants covered to prevent plant dehydration. Upon arrival, seedlings should be immediately stored in a ditch, watered, and covered with soil (Iancu, 1999; Abrudan, 2006, 2010).

Field establishment

Q. petraea typically grows on hills, where site preparation is carried out in 1-m-wide strips along contour lines and alternating with undisturbed strips. Soil preparation within the strips involves plowing or manually digging 40 × 40 × 30 cm holes. On rocky

slopes, soil preparation is limited to small bench terraces measuring 2 × 2 m to 2 × 3 m. On steep, shallow-soil slopes, preparation involves either narrow terraces (<1.2 m wide) or wide terraces (>1.2 m wide), arranged along contour lines. The spacing between terraces ranges 2–3 m (Iancu, 1999).

Q. petraea can be established by planting or direct seeding. Plantings are typically arranged in rows with an initial stocking density ranging from 2500 to 15,000 plants/ha, but most commonly between 4000 and 6000 plants/ha (Iancu, 1999; Anonymous, 2022). An alternative to high-density row planting is cluster planting. This method uses clusters of 20–30 seedlings arranged in either nests (nest planting), with approximately 0.2 m between trees, or groups (group planting), with an initial spacing of 1 m between trees (Saha, 2024). In group planting, each group typically contains around 20 seedlings, surrounded by 8–16 shade-tolerant trainer trees, such as *Fagus sylvatica*, *Carpinus betulus*, and *Tilia cordata*. These trainer trees promote self-pruning and help to maintain a desired branch-free bole length of at least 25% of tree height by inhibiting epicormic branches (Skiadaresis et al., 2016; Saha, 2024). The distance between group centers ranges from 10 to 13 m, resulting in 60–100 groups/ha and an initial planting density of 1800 to 4200 seedlings/ha (Skiadaresis et al., 2016). *Q. petraea* trees planted in groups demonstrate similar quality and growth to those planted in traditional rows, while also offering cost savings during establishment and promoting higher tree diversity (Skiadaresis et al., 2016; Mölder et al., 2017; Andrzejczyk et al., 2023; Saha, 2024).

Another strategy for artificial regeneration of *Q. petraea*, particularly when natural regeneration fails, is transplanting naturally regenerated seedlings established under the parent stand. After one growing season, the survival rate of 4- and 5-year-old transplants ranged from 34.3% (in open canopies with direct solar radiation in summer) to 67.1% (under shelter and in areas with favorable soil moisture) (Kanjevac et al., 2023).

Direct seeding is used locally and on a much smaller scale than planting. Two methods of seeding are employed: broadcast seeding, which is not recommended because of the subsequent problems with competing vegetation; and seed-drill seeding. This latter method is more suitable and the most common one. Drills are opened 1.7–2.0 m apart, 5 cm deep; the acorns are sown in the drills at a density of 15–20 seeds/m (7–5 cm apart), covered with mineral soil and pressed in (Joyce et al., 1998). Autumn sowing (October–November) is preferred as there is no need for any winter seed storage (Joyce et al., 1998; Łukaszewicz and Gil, 2007). The amount of acorns used for seeding depends on the seeding method and ranges from 200 kg/ha in Poland (Lasy Państwowe, 2023) to 1000 kg/ha in Germany (Bartsch et al., 2020). However, as emphasized by Löf et al. (2019), issues such as poor seed quality, acorn predation by rodents, competing vegetation, and browsing damage can cumulatively contribute to reduced seedling establishment that leads to the unreliability of direct seeding (the least costly approach for artificially regenerating oaks) as a large-scale practice of establishing

oaks. Success can be increased if acorns can be protected from small predators using tree shelters or several types of seed shelters (see Löf et al., 2019).

Successful plantations require tending until complete crown closure (five to seven years after planting), including two to three summer interventions involving mechanical soil cultivation between rows and manual mowing or mulching within rows (0.5 m around plants) during the first 2–3 years after establishment (Haralamb, 1967; Iancu, 1999; Abrudan, 2006). Weeding is also necessary to control competing vegetation, including weeds, undergrowth, and stump sprouts (Iancu, 1999; Anonymous, 2022).

Fertilizers and irrigation are usually not required for *Q. petraea* plantings (Haralamb, 1967; Iancu, 1999; Anonymous, 2022). However, seedlings are vulnerable to damage from wildlife. Browsing by *Cervus elaphus* (red deer), and *Dama dama* (fallow deer) affects leaves and twigs, while *Oryctolagus cuniculus* (rabbits) damages bark. In addition, acorns are consumed by *Sus scrofa* (wild boar), *Sciurus vulgaris* (Eurasian red squirrel), *Oryctolagus cuniculus*, and *Glis glis* (fat dormouse) (Bary-Lenger and Nebout, 1993; Margaletić, 2003; Gačić et al., 2006). Individual protection methods include using nets, repellents, and rodenticides (Margaletić, 2003). Tree shelters with a height of 1.2 m have proven to be an effective compromise, promoting seedling survival and increasing height growth (Mayhead and Boothman, 1997). In several European countries, such as Sweden and Germany, large regeneration areas are commonly fenced to protect against browsing (Leonardsson et al., 2015; Mölder et al., 2019). While tree shelters and fencing effectively protect seedlings and regeneration areas, they significantly increase forestation costs.

Chapter References

Abrudan, I.V., 2006. Afforestations. Editura Universităţii Transilvania, Braşov, Romania (In Romanian).

Abrudan, I., 2010. Afforestations. Universitatea Transilvania, Braşov, Romania (In Romanian).

Aldhous, J.R., 1994. Nursery policy and planning. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Forestry Commission Bulletin, vol 111. HMSO, London, pp. 1–12.

Andrzejczyk, T., Liziniewicz, M., Bolibok, L., 2023. Growth and quality of 16-year-old sessile oak (*Quercus petraea* (Matt.) Liebl.) planted in traditional and alternative row planting patterns. Forestry 1–16. https://doi.org/10.1093/forestry/cpad059.

Anonymous, 2022. Good practice guide for species compositions, schemes and technologies for forest regeneration and afforestation of degraded lands. Monit. Ofic. Roman. Partea I 5–382, 1000 bis, 14 octombrie 2022.

Bakker, M.R., Kerisit, R., Verbist, K., Nys, C., 1999. Effect of liming on rhizosphere chemistry and growth of fine roots and of shoots of sessile oak (*Quercus petraea*). Plant Soil 217, 243–255.

Bartsch, N., Lüpke, B. von, Röhrig, E., 2020. Silviculture on Ecological Basis, eighth ed. UTB, Ulmer, Stuttgart, Germany (in German).

Bary-Lenger, A., Nebout, J.-P., 1993. Pedunculate and sessile oaks in France and in Belgium. In: Ecology-Economy-History-Silviculture. Editions du Perron, Alleur-Liège, France (in French).

Boudru, M., 1992. Forest and Silviculture: Afforestation and Reforestation. Les presses agronomiques de Gembloux, A.S.B.L. Gembloux, France (in French).

CDAF, 2014. Agroforestry in Wallonie. B. Good Practices. B.1. Choice and Reception of Seedlings. Centre du Developpement Agroforestier de Chimay, Chimay, Belgium (in French).

Chalupa, V., 1984. *In vitro* propagation of oak (*Quercus robur* L.) and linden (*Tilia cordata* Mill.). Biol. Plant. 26, 374–377.
Chalupa, V., 1993. Vegetative propagation of oak (*Quercus robur* and *Q petraea*) by cutting and tissue culture. Ann. Des. Sci. For. 50 (Suppl. 1), 295s–307s.
Chalupa, V., 2005. Protocol of somatic embryogenesis: pedunculate oak (*Quercus robur* L.) and sessile oak (*Quercus petraea* (Matt.) Liebl.). In: Mohan Jain, S., Gupta, P.K. (Eds.), Protocol for Somatic Embryogenesis in Woody Plants. Forestry Sciences, vol 77. Springer, Dordrecht, pp. 369–378.
Evans, J., 1988. Natural regeneration of broadleaves. In: Forestry Commission Bulletin 78, HMSO, London, UK.
Fernández-López, M.J., Barrio-Anta, M., Álvarez-Álvarez, P., López-Varela, B., Gómez-Fernández, J.A., 2013. *Quercus petraea* (Matt.) Liebl. And *Quercus robur* L. In: Pemán, J., Navarro-Cerrillo, R., Nicolás, J. L., Prada, M.A., Serrada, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Tomo II. Organismo Autónomo Parques Nacionales. Ministerio de Agricultura, Alimentación y Medio Ambiente. Serie Forestal, Madrid, Spain, pp. 251–277 (in Spanish).
Fletcher, A.M., 1992. Flower, fruit and seed development and morphology. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees. Forestry Commission Bulletin, vol 83. HMSO, London, pp. 59–70.
Florescu, G., 1996. Afforestations. Forest Seeds. Universitatea Transilvania, Braşov, Romania (in Romanian).
Gačić, D., Krstić, M., Laketić, M., 2006. Effect of big game on sessile oak forests in the national park "Đerdap". Šumarstvo (Belgrade) 1–2, 21–33 (in Serbian).
Gordon, A.G., 1992a. Cone and seed collection and handling before processing. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees, Forestry Commission Bulletin, vol. 83. HMSO, London, UK, pp. 80–85.
Gordon, A.G., 1992b. The processing of cones and seeds. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees, Forestry Commission Bulletin, vol 83. HMSO, London, UK, pp. 86–97.
Gordon, A.G., 1992c. Seed storage. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees. Forestry Commission Bulletin, vol 83. HMSO, London, UK, pp. 98–102.
Gordon, A.G., Faulkner, R., 1992. Identification and assessment of cone and seed crops. In: Gordon, A.G. (Ed.), Seed Manual for Forest Trees, Forestry Commission Bulletin, vol 83. HMSO, London, UK, pp. 71–79.
Gosling, P.G., Aldhous, J.R., 1994. Seed. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Forestry Commission Bulletin, vol 111. HMSO, London, UK, pp. 66–83.
Haralamb, A., 1967. Culture of Forest Species, third ed. Editura Agro-Silvică de Stat, Bucureşti, Romania (in Romanian).
Iancu, I., 1999. Manual of Forest Ranger. Theory and Practice. Tipografia Pegasus, Bucureşti, Romania (in Romanian).
Jokanović, D., Nikolić Jokanović, V., Živanović, K., Ilić, M., Antanasijević, N., Jovanović, F., 2024. Variability of morpho-anatomical traits of one year-old *Quercus* sp. container seedlings. Reforesta 18, 22–33.
Joyce, P.M., Huss, J., McCarthy, R., Pfeifer, A., Hendrick, E., 1998. Growing Broadleaves. Silvicultural Guidelines for Ash, Sycamore, Wild Cherry, Beech and Oak in Ireland. COFORD, Dublin. Ireland.
Kanjevac, B., Krstić, M., Babić, V., 2023. Artificial regeneration of sessile oak forests by planting natural young stock. Šumarstvo (Belgrade) 1–2, 55–67 (in Serbian).
Kasprzyk, W., Baranowska, M., Korzeniewicz, R., Behnke-Borowczyk, J., Kowalkowski, W., 2022. Effect of irrigation dose on powdery mildew incidence and root biomass of sessile oak (*Quercus petraea* (Matt.) Liebl.). Plants 11, 1248. https://doi.org/10.3390/plants11091248.
Kerr, G., Evans, J., 1993. Growing Broadleaves for Timber. Forestry Commission Handbook 9. HMSO, London, UK.
Lasy Państwowe, 2023. Silvicultural principles. In: DGLP, Warszawa, Poland (In Polish).
Leonardsson, J., Löf, M., Götmark, F., 2015. Exclosures can favour natural regeneration of oak after conservation-oriented thinning in mixed forests in Sweden: a 10-year study. For. Ecol. Manag. 354, 1–9. https://doi.org/10.1016/j.foreco.2015.07.004.
Löf, M., Castro, J., Engman, M., Leverkus, A.B., Madsen, P., Reque, J.A., Villalobos, A., Gardiner, E.S., 2019. Tamm Review: direct seeding to restore oak (*Quercus* spp.) forests and woodlands. For. Ecol. Manag. 448, 474–489. https://doi.org/10.1016/j.foreco.2019.06.032.

Łukaszewicz, J., Gil, W., 2007. History and present of afforestation and reforestation by seeding. Sylwan 3, 11–22 (in Polish).
Margaletić, J., 2003. Rodent damage and protection. In: Matić, S. (Ed.), Common Beech in Croatia, Academy of Forestry Sciences, Zagreb, Croatia, pp. 574–584.
Mason, W.L., 1994. Production of bare-root seedlings and transplants. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Forestry Commission Bulletin, vol 111. HMSO, London, UK, pp. 84–103.
Mason, W.L., Jinks, R.L., 1994. Vegetative propagation. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Forestry Commission Bulletin, vol 111. HMSO, London, UK, pp. 135–147.
Mayhead, G.J., Boothman, I.R., 1997. The effect of treeshelter height on the early growth of sessile oak (*Quercus petraea* (Matt.) Liebl.). Forestry 70 (2), 151–155.
Mölder, A., Nagel, R.V., Meyer, P., Schmidt, M., Rumpf, H., Spellmann, H., 2017. Historical retrospective of 19th century oak regeneration in the Spessart Mountains (Germany) – the importance of former silvicultural techniques for today's oak management. Forstarchiv 88 (3), 67–78 (in German).
Mölder, A., Sennhenn-Reulen, H., Fischer, C., Rumpf, H., Schönfelder, E., Stockmann, J., Nagel, R.V., 2019. Success factors for high-quality oak forest. Quercus robur, Q. petraea) regeneration. For. Ecosys. 6, 49. https://doi.org/10.1186/s40663-019-0206-y.
Negulescu, E., Săvulescu, Al, 1957. Dendrology. In: Editura Agro-Silvică de Stat, Bucureşti, Romania (in Romanian).
Negulescu, E.G., Săvulescu, Al, 1965. Dendrology, second ed. Editura Agro-Silvică, Bucureşti, Romania (in Romanian).
Orešković, Z., Dokuš, A., Harapin, M., Jakovljević, T., Maradin, R., 2006. Growing of seedlings of common oak (*Quercus robur* L.) and sessile-flowered oak (*Quercus petraea* (Matt.) Liebl) in different types of containers. Rad./Zavoda Znan. Umjetnički Rad. Požegi 9, 75–86 (in Croatian).
Pesendorfer, M.B., Bogdziewicz, M., Szymkowiak, J., Borowski, Z., Kantorowicz, W., Espelta, J.M., et al., 2020. Investigating the relationship between climate, stand age, and temporal trends in masting behavior of European forest trees. Glob. Change Biol. 26, 1654–1667. https://doi.org/10.1111/gcb.14945.
Rozporządzenie, 2004. Regulation of the Minister of Environment dated 18.02.2004 on detailed requirements to be met by forest reproductive material. J. Laws 31 (in Polish).
Rubţov, Şt, 1958. Cultivation of Woody Species in Nurseries (Forestry And Decorative).
Stănescu, V., 1979. Dendrology. In: Editura Didactică Şi Pedagogică, Bucureşti, Romania (in Romanian).
Stănescu, V., Şofletea, N., Popescu, O., 1997. Woody Forest Flora of Romania. Editura Ceres, Bucureşti, Romania (in Romanian).
Strouts, R.G., Gregory, S.C., Heritage, S.G., 1994. Protection against climatic damage, fungal diseases, insects and animal pests. In: Aldhous, J.R., Mason, W.L. (Eds.), Forest Nursery Practice. Forestry Commission Bulletin, vol 111. HMSO, London, UK, pp. 181–197.
Saha, S., 2024. Low-density tree planting: scope of oak cluster planting to create mixed forests. In: Martens, C., Clark, S., Schweitzer, C. (Eds.), The International Oak Symposium: Science-Based Management for Dynamic Oak Forests. General Technical Report SRS-278. USDA Forest Service, Southern Research Station, Asheville, NC, p. 12.
Skiadaresis, G., Saha, S., Bauhus, J., 2016. Oak group planting produces a higher number of future crop trees, with better spatial distribution than row planting. Forests 7, 289. https://doi.org/10.3390/f7110289.
Solymos, R., 1993. Improvement and silviculture of oaks in Hungary. Ann. Des. Sci. For. 50 (6), 607–614.
Şofletea, N., Curtu, L., 2007. Dendrology. Editura Universităţii "Transilvania. Braşov, Romania (in Romanian).
Struve, D., Ferrini, F., Bellasio, C., Fini, A., 2010. Propagation of *Quercus cerris, Q. petraea*, and *Q. pubescens* seedlings by stem cuttings. Hortscience 45 (11), 1729–1733.
Willoughby, I., Balandier, P., Bentsen, N.S., McCarthy, N., Claridge, J. (Eds.), 2009. Forest Vegetation Management in Europe: Current Practice and Future Requirements. COST Office. Brussels, Belgium.
Vlase, I., 1982. Conservation of Forest Seeds. Editura Ceres, Bucureşti, Romania (in Romanian).

Quercus pubescens Willd.

Claudia Cocozza[1] **and Ivona Kerkez Janković**[2]
[1]Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy; [2]University of Belgrade, Faculty of Forestry, Belgrade, Serbia

Propagation

Seed propagation

Acorns of *Quercus pubescens* (downy or pubescent oak) are typically collected in early October (Fig. 11.41). For harvesting methods applicable to this oak species, readers can refer to the guidelines provided for other oaks, such as *Q. ilex*, in this Compendium. As recalcitrant seeds, *Q. pubescens* acorns are highly sensitive to desiccation, which adversely affects their viability. To maintain viability from collection to sowing, the moisture content of *Q. pubescens* acorns should not drop below 40%, with an optimal range of 42%–48%.

For short-to-medium-term storage, it is important to prevent acorns from drying out while allowing respiration and avoiding fermentation. Storage in airtight containers must be avoided, and temperatures should be maintained between −4°C and 3°C. During stratification acorns are mixed with a humid growing medium and placed between layers of a porous substrate or mixed in a 1:1 volume ratio. Suitable substrates include peat, sand, sawdust, vermiculite, or coconut fiber, which absorb excess moisture and prevent germination while separating acorns to reduce fungal spread (Villar-Salvador et al., 2013). Acorns and substrate are placed in 30–60 L containers equipped with a perforated



Figure 11.41 Left: Acorn batch of *Quercus pubescens* before cleaning. Right: Early emerged seedling of *Quercus pubescens*. *(Ivona Kerkez Janković.)*

10 cm wide plastic tube vertically positioned to facilitate aeration. The container is then sealed with a porous yet durable material, such as plastic sheeting, paper, or perforated cardboard, leaving the aeration tube protruding. An alternative and simpler method involves storing clean, dry-surface acorns in polyethylene bags (40–70 μm wall thickness) with a zip-lock or heat seal. Bags are arranged in perforated, stackable plastic boxes to ensure air circulation. Polyethylene plastic prevents moisture loss while permitting some oxygen and carbon dioxide exchange to maintain respiration (Bonner, 2008; Rink and Williams, 1984). However, thicker plastic can result in acorn death.

Stored acorns must be checked periodically. If taproots grow too long, storage should be interrupted. Pregerminated acorns with root lengths of 0.5–5 cm are optimal for sowing, though roots exceeding this length may be trimmed to 3 cm before planting. In other oaks, this practice does not have any negative effect on nursery growth or outplanting survival, although the seedlings will lose their primary taproot and develop two to five thinner roots (Bonner, 1982; Hankin and Watson, 2020). To promote proper root and shoot formation, acorns should be sown horizontally in nurseries or in the field slightly tilted with the apex facing downward.

In some European seed processing facilities, acorns undergo thermotherapy, where they are soaked in water at 41°C for 2–3 h to eliminate the fungus *Ciboria batschiana*. After treatment, the acorns must be surface-dried to restore moisture content to approximately 45% and treated with fungicides such as methyl thiophanate, benomyl, or iprodione before storage.

Prior to sowing in seedbeds or containers, it is advisable to soak acorns in water to restore full moisture and remove non-viable seeds, especially in acorns stored for several weeks. Germination and emergence rates in seedbeds are highly variable, ranging from 5 to 67% depending on the mother tree (Šijačić-Nikolić et al., 2021) and storage conditions (Fig. 11.41). Average characteristics of *Q. pubescens* acorns are detailed in Table 11.56.

Vegetative propagation

It is possible to propagate *Q. pubescens* using semihardwood stem cuttings. One-year-old seedlings are harvested at the beginning of the summer with one or more shoots at least 5 cm long with six or more nodes (Struve et al., 2010). The length of cuttings varies

Table 11.56 Characteristics of seed lots of *Q. pubescens*.

Purity (%)	No. seeds/kg	Germination capacity (%)
98–100 (97)	190–829 (414)	24–90 (57)

Alía et al. (2009); Gradi (1989); Herrero Sierra and Nicolás Peragón (2013); Piotto and Falleri (2001); Šijačić-Nikolić et al. (2021).

between 3 and 10 cm and should have three to seven nodes with each seedling yielding one to five cuttings, depending on its size. Struve et al. (2010) recommend removing the basal leaves and cut the remaining leaves in half. The basal end of the cuttings should then be dipped for 5 s in a 0.1 M ascorbic acid solution and then in a 500 ppm solution of the potassium salt of indole butyric acid. Cuttings can be rooted in expanded foam trays with multiple cells. The trays are placed in a clear plastic propagation tent within a greenhouse. The ambient minimum and maximum temperatures inside the propagation tent should around 23 ± 2°C at night and 32 ± 2°C during the day with a 48% reduction in light (Struve et al., 2010). Rooting is evaluated in December and overwinter survival in March. A cutting is considered rooted if it has grown at least one root 1 cm long (Struve et al., 2010).

Seedling cultivation

There is relatively limited information on the nursery cultivation of *Quercus pubescens*. This species can be cultivated as container or bareroot plants, and most of the information presented here is based on our own experience.

Container plants

Container cultivation should ideally start in a greenhouse in regions with cold winters and frequent spring frosts. A growing medium composed of equal parts of black peat and natural soil (1:1, v:v) can be used. Fertilization should be applied twice during the growing period using a liquid NPK foliar fertilizer with added microelements (N = 10%, P_2O_5 = 8%, K = 6%, Fe = 0.02%, Mn = 0.02%, B = 0.01%, Cu = 0.02%, Mo = 0.001%, Zn = 0.006%) at a 0.3% concentration (Šijačić-Nikolić et al., 2021; Devetaković et al., 2023). Root deformation in both 1 + 0 and 2 + 0 seedlings, including root twisting and the development of multiple taproots, has been observed in Bosnaplast 18 containers (hexagonal, hard plastic containers with dimensions of 32 × 21.5 × 18 cm, v = 220 cm^3, and 33 holes per tray). Regular irrigation and manual weeding are recommended. One-year-old container seedlings showed an average root collar diameter of 2.93 mm (range 1.61–4.62 mm) and an average height of 10 cm (range 6–16.1 cm) (Šijačić-Nikolić et al., 2021; Devetaković et al., 2023).

Given the ecological and morphological similarities between *Q. pubescens* and *Q. faginea*, readers are encouraged to consult additional information in this Compendium regarding container cultivation characteristics of *Q. faginea*, particularly for plantations on dry sites.

Bareroot plants

Nursery cultivation of bareroot plants can be conducted in seedbeds or Dunemann beds (Fig. 11.42), starting in late October for autumn sowing or late February for spring



Figure 11.42 One year old bareroot seedlings of Quercus pubescens in a Duneman bed. *(Ivona Kerkez Janković.)*

sowing. Before sowing, it is advisable to treat seeds with systemic fungicides containing copper oxychloride (283.8 g/kg) and metalaxyl-M (38.8 g/kg) as well as rodenticides (4-hydroxycoumarin derivatives) in powder form, applied directly to the seeds.

Seedbed preparation should be conducted immediately before sowing and includes mowing, chopping, disking of cover crops (if present), deep ripping (if necessary), disking, plowing, and bedding. Sowing depths are up to 5 cm for spring sowing and up to 7 cm for autumn sowing. The recommended row spacing is 20 cm, with acorn spacing of 2 cm within the row. Acorns should be placed horizontally, as a vertical position negatively affects germination and seedling development, according to nursery growers' observations (Šijačić-Nikolić et al., 2021; Devetaković et al., 2023).

Stock types such as 1 + 0, 2 + 0, 1 + 1, 1 + 2, and 3 + 0 can be used for planting. However, transplanting, even within the nursery, can result in significant seedling mortality. Shading is unnecessary, even during the germination phase. Weeding can be performed manually or with selective herbicides. For broad-leaved weeds, a translocating herbicide from the benzoic acid derivatives group (e.g., dicamba-DMA, 577.9 g/L) is effective, while for annual and perennial narrow-leaved weeds, herbicides from the cyclohexenone chemical group (e.g., clethodim) are recommended.

Moderate drip irrigation systems have proven effective. Fertilization can be done with granular NPK fertilizer (N = 14%, P = 14%, K = 14%) at 300 kg/ha, applied

once or twice annually, or with liquid foliar fertilizers containing microelements (as previously described). The effects of combining granular and foliar fertilizers on plant growth have not been studied.

Average seedling height and root collar diameter vary by age (Šijačić-Nikolić et al., 2021; Devetaković et al., 2023) in the following way (Table 11.57):

Rough handling of seedlings can lead to severe root damage and significantly impact outplanting performance. Although no specific information is available on seedling storage, based on experience, lifting should occur up to 3 days before planting. Seedlings should be bundled and their roots covered with polyethylene bags.

Field establishment

Quercus pubescens exhibits a broad altitudinal range, particularly in southern European countries. While it is more commonly found on hillsides between 200 and 800 m, it can grow from coastal plains up to elevations of 1200–1300 m. Although indifferent to pH, *Q. pubescens* prefers lime-rich, well-drained soils in the northern parts of its range but is also found on acidic soils in warmer regions such as Sicily and Crete. It behaves as a heliophilous and thermophilous species, well-adapted to moderate summer drought and low winter temperatures, though it avoids continental areas with frequent severe frosts and prolonged droughts. In the Balkan Peninsula's under a more continental climate, seedlings exhibit an evergreen habit during their juvenile phase (one to 2 years old), retaining green leaves throughout mild winter conditions on sunny sites (Bobinac et al., 2024).

As a poor resprouter, *Q. pubescens* is susceptible to being outcompeted by more resilient species under frequent anthropogenic disturbances such as cutting, wildfires, and overgrazing. However, it demonstrates a capacity for rapid colonization through seed dispersal once disturbances are removed. Forests dominated by *Q. pubescens* are common across southern Europe and are confined to warm microclimatic conditions in central Europe. Vegetation surveys of these communities have been conducted in Germany, Bulgaria, Czech Republic, Italy, Austria, Slovakia, Hungary, Greece, Montenegro, and Serbia (Pasta et al., 2016). In the Balkan Peninsula's continental climate, natural regeneration has been observed to be very poor (Šijačić-Nikolić et al., 2021).

Table 11.57 Morphological characteristics of different stock type in bareroot *Quercus pubescens*.

Age/stock type	Height (cm)	Root collar diameter (mm)
1 + 0	20	3.3
2 + 0	51	6.6
2 + 1	60	9.3
3 + 0	99	9.6

Q. pubescens has been used to enrich *Pinus halepensis* plantations with the success of direct seeding being influenced by light and water availability. Light in the understory of dense pine plantations is as low as 13% of the outside irradiance, which is insufficient for optimal growth of Q. pubescens juveniles (Prévosto et al., 2011a, 2011b). Effective site preparation, using mechanical and chemical methods, reduces competition from herbaceous and woody species (Lowery and Gjerstad, 1991). Soil improvement practices such as soil ripping enhance soil physical and chemical properties, benefiting seedling growth (Lowery and Gjerstad, 1991; Savill et al., 1997; Filothei and Ganatsas, 2023).

In marginal populations, *Q. pubescens* plays a crucial role in preventing erosion on degraded habitats. Effective site preparation is therefore essential for restoring degraded oak forest ecosystems, contributing to global goals (Filothei and Ganatsas, 2023). Container-grown 1 + 0 seedlings exhibit better survival and height growth compared with bareroot seedlings, though with slightly lower diameter growth (Devetaković et al., 2023). Large 2 + 0 bareroot seedlings achieve survival rates of up to 93%, but survival drops to below 10% in 2 + 1 and 3 + 0 seedlings, likely due to combined factors such as prolonged dry periods during the growing season and transplantation shock. Common planting spacing are 1.5–2 × 1.5–2 m.

Direct seeding is also viable, provided that acorns are protected from rodents using tree or seed shelters (Castro et al., 2015; Oliet et al., 2015). For autumn sowing in cold regions, mulching is recommended to protect the acorns.

Chapter References

Alía Miranda, R., García del Barrio, J.M., Iglesias Sauce, S., Mancha Núñez, J.A., de Miguel y del Ángel, J., Nicolás Peragón, J.L., Pérez Martín, F., Sánchez de Ron, D., 2009. Regiones de procedencia de especies forestales españolas. Organismo Autónomo Parques Nacionales, Madrid, Spain.

Bobinac, M., Šušić, N., Šijačić-Nikolić, M., Janković, I.K., Veljović-Jovanović, S., 2024. Photosynthetic insights into winter-green leaves in *Quercus pubescens* Willd. seedlings. Arch. Biol. Sci. 76, 223–232.

Bonner, F.T., 1982. The effect of damaged radicles of presprouted red oak acorns on seedling production. Tree Planters' Notes (U.S. Dep. Agric. For. Serv.) 33 (4), 13–15.

Bonner, F.T., 2008. Storage of seeds. In: Woody Plant Seed Manual. USDA Forest Service, pp. 86–95.

Castro, J., Leverkus, A.B., Fuster, F., 2015. A new device to foster oak forest restoration via seed sowing. N. For. 46, 919–929. https://doi.org/10.1007/S11056-015-9478-4.

Devetaković, J., Đilas, M., Kerkez Janković, I., 2023. Stocktype impact on survival and growth of one-year-old *Quercus pubescens* seedlings on the edge of Panonnian basin. Šumarski list 147, 547–554.

Filothei, M., Ganatsas, P., 2023. Effect of land preparation methods on restoration success of degraded oak forest ecosystems. Dendrobiology 89, 56–64. https://doi.org/10.12657/denbio.089.006.

Gradi, A., 1989. Vivaistica forestale. In: Edagricole. Bologna, Italy.

Hankin, S., Watson, G., 2020. Oak taproot growth disruption differentially impacts root architecture during nursery production. Forests 11 (8), 798. https://doi.org/10.3390/F11080798.

Herrero Sierra, N., Nicolás Peragón, J.L., 2013. Datos característicos de lotes de semillas. CNRGF El Serranillo, Dirección General de Desarrollo Rural y Política Forestal, Ministerio de Agricultura, Alimentación y Medio Ambiente. In: Pemán García, J., Navarro Cerrillo, R., Nicolás Peragón, J.L., Prada Saéz, A., Serrada Hierro, R. (Eds.), Producción y manejo de semillas y plantas forestales, Organismo Autónomo de Parques Nacionales, Ministerio de Agricultura, Alimentación y Medio Ambiente. Madrid, Spain, vol II, pp. 734–739.

Lowery, R.F., Gjerstad, D.H., 1991. Chemical and mechanical site preparation. In: Sutherland, J.R. (Ed.), Forest Regeneration Manual, pp. 251–261. https://doi.org/10.1007/978-94-011-3800-0_13.

Oliet, J.A., Vázquez de Castro, A., Puértolas, J., 2015. Establishing *Quercus ilex* under Mediterranean dry conditions: sowing recalcitrant acorns versus planting seedlings at different depths and tube shelter light transmissions. N. For. 46, 869–883. https://doi.org/10.1007/S11056-015-9495-3.

Pasta, S., de Rigo, D., Caudullo, G., 2016. *Quercus pubescens* in Europe: distribution, habitat, usage and threats. In: San-Miguel-Ayanz, J., de Rigo, D., Caudullo, G., Houston Durrant, T., Mauri, A. (Eds.), European Atlas of Forest Tree Species. Publ. Off. EU, Luxembourg. E019e5c.

Piotto, B., Falleri, E., 2001. La conservazione dei semi. In: Piotto, B., di Noi, A. (Eds.), Propagazione per seme di alberi e arbusti della flora mediterranea. ANPA, Rome, Italy.

Prévosto, B., Monnier, Y., Ripert, C., Fernandez, C., 2011a. Can we use shelterwoods in Mediterranean pine forests to promote oak seedling development? For. Ecol. Manag. 262, 1426–1433.

Prévosto, B., Monnier, Y., Ripert, C., Fernandez, C., 2011b. Diversification of *Pinus halepensis* forests by sowing *Quercus ilex* and *Quercus pubescens* acorns: testing the effects of different vegetation and soil treatments. Eur. J. For. Res. 130, 67–76.

Rink, G., Williams, R.D., 1984. Storage technique affects white oak acorn viability. Tree Plant. Notes 35 (1), 3–5.

Savill, P., Evans, J., Auclair, D., Falck, J., 1997. Site preparation. In: Savill, P. (Ed.), Plantation Silviculture in Europe. Oxford Academic, pp. 59–76. https://doi.org/10.1093/oso/9780198549093.003.0005.

Šijačić-Nikolić, M., Vilotić, D., Ivetić, V., Aleksić, J., Milovanović, J., Nonić, M., Devetaković, J., Jokanović, D., Perović, M., Popović, V., Lučić, A., Kerkez-Janković, I., Jankov, V., 2021. Identification and Monitoring of the Gene Pool of Rare, Vulnerable and Endangered Species of Plants of SP "Košutnjak Forest" - Study. University of Belgrade, Faculty of Forestry, Secretariat for Environmental Protection of the City of Belgrade, Belgrade (In Serbian: Идентификаци а и Мониторинг генофонда ретких, рањивих и угроЖених врста биљака СП „ШуМа Кошутњак" – студи а).

Struve, D., Ferrini, F., Bellasio, C., Fini, A., 2010. Propagation of *Quercus cerris, Q. petraea*, and *Q. pubescens* seedlings by stem cuttings. Hortscience 45, 1729–1733.

Villar-Salvador, P., Oliet Palá, J.A., Heredia-Guerrero, N., Uscola, M., Goicoetxea, P., 2013. *Quercus faginea* Lam. y *Quercus humilis* Mill. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales. Organismo Autónomo Parques Nacionales, Serie Forestal, pp. 206–225.

Quercus pyrenaica Willd.

Enrique Andivia[1], Juan A. Oliet[2], Francisco Javier Lucas-Villar[2], Pablo González-Goicoetxea[3] and Rafael Serrada-Hierro[4]

[1]Department of Biodiversity, Ecology and Evolution, Faculty of Biological Sciences, Universidad Complutense de Madrid, Madrid, Spain; [2]Escuela Tecnica Superior de Ingeniería de Montes, Forestal y del Medio Natural, Universidad Politecnica de Madrid, Madrid, Spain; [3]Department of Forestry, NEIKER-BRA, Vitoria-Gasteiz, Spain; [4]Sociedad Española de Ciencias Forestales, Madrid, Spain

Propagation

Seed propagation

Acorn collection usually takes place between October and November. As for other *Quercus* species, recommended harvesting time is when the acorns turn a dark brown color, without traces of green areas. Collection is done manually from the ground after shaking trees. To ensure high germination rates, avoid collecting acorns that have fallen spontaneously to the ground. The processing and storage of acorns follow similar recommendations to those for other *Quercus* species. Firstly, acorns should be immersed in hot water (45°C) for about 2 h to eliminate any potential presence of beetle and moth larvae. Acorns that float should also be discarded. Another effective method for disinfecting acorns is to immerse them for 30 min in a hypochlorite solution (1%) and then rinse them thoroughly with water. Secondly, acorns should be dried until they reach 40%–45% moisture content. Finally, the acorns should be stored in 50 μm thick polyethylene bags inside open containers that allow air circulation, kept in a refrigerated chamber at 1–2°C. Hydration by soaking acorns in water for 48 h before planting is highly recommended. Following these recommendations has resulted in high germination rates (Table 11.58).

Vegetative propagation

Q. pyrenaica has a great capacity for resprouting. However, propagation through cuttings is difficult, although possible with root cuttings with stems. Grafting has moderate success and is usually carried out under controlled conditions in the greenhouse (Vivero et al., 2000). Despite the fact that somatic embryogenesis and other embryogenesis methods have been successfully applied to the propagation of oak species (Correidora

Table 11.58 Characteristics of seed lots of *Quercus pyrenaica*.

Purity (%)	No. seeds/kg	Germination capacity (%)
90–100 (99)	125–330 (185)	70–100 (81)

Mean values in brackets.
References and technical documents included in Lucas-Villar et al. (2013).

et al., 2014; Gomez-Garay et al., 2014), there is no information about specific methods for the vegetative propagation of *Q. pyrenaica*.

Seedling cultivation

Container plants

Q. pyrenaica, similar to other Mediterranean oaks, is manly produced in containers. Sowing usually takes place in November under greenhouse conditions at around 20°C. Sowing is also possible in spring, but late sowing usually results in lower seedling quality (Lucas-Villar et al., 2013). Recommendations for sowing are similar to those for other *Quercus* species. The acorn is placed horizontally or with the apex slightly pointing down and the acorn covered with approximately 1 cm of substrate. Acorns are often pregerminated before sowing, with the optimal radicle length between 0.5 and 5 cm. The use of shading nets (around 20% of shade) is recommended.

The cultivation of *Q. pyrenaica* seedlings is usually done in containers with volume of 300–400 cm^3. The container should have an anti-spiraling system and a minimum depth of 18 cm for one-year-old seedlings and around 35 cm for two-year-old seedlings. Cultivation density should not exceed 300 seedlings/m^2. The recommended substrate is peat, mixed with inorganic components such as vermiculite or perlite (3:1 by volume).

As for other oaks, acorn nutritional reserves support seedling growth during the first months of cultivation, after which fertilization should be provided to prevent nutrient limitations (Villar-Salvador et al., 2010). Fertilization trails with *Q. pyrenaica* are scarce but slow-release fertilizers can be used at rates of 2.5–3 kg/m^3 of substrate, with an NPK 16–8–12 fertilizer type (Calderón, 2004). Seedlings are usually stored outdoors in the nursery. Recommendations for producing high-quality *Q. pyrenaica* seedlings (1 + 0 container stock) point to seedlings of height and diameter larger than 20 and 2 cm, respectively (Lucas-Villar et al., 2013).

Field establishment

Water stress, seed predation, and herbivory are the main limiting factors for oak regeneration (Pemán et al., 2017). Planting has been widely used and recommended as the main establishment method for this species (Montoya, 1995). For planting, one-year-old seedlings (1 + 0) are recommended since 2 + 0 seedlings exhibit higher foliar area. Larger seedlings can lead to increased water stress, especially when planted under a canopy where shade reduces the investment in root biomass (Rodríguez-Calcerrada et al., 2007). Planting should be performed during the wet season in autumn on sites with mild winters (Navarro-Cerrillo et al., 2021). On sites with cold winters, planting is usually done in February.

Planting *Q. pyrenaica* is recommended on deep non-limestone soils that have high water availability and on sites with favorable microclimates. It is advisable to carry out

intensive soil preparation through linear subsoiling as deeply as possible, following the contour lines on slopes of less than 35%. For slopes up to 60%, digging holes along the lines of maximum slope is recommended. In addition, controlling competing vegetation before outplanting is also recommended. However, nurse shrubs can improve seedling survival by ameliorating water stress and protecting against herbivores (Castro et al., 2006; Perea and Gil, 2014; Torroba-Balmori et al., 2015). Recommended planting density should not exceed 2000 seedlings/ha with a spacing of 2 × 2.5 m (Álvarez et al., 2000). Planting densities of approximately 400 seedlings/ha, using an excavator to dig holes of approximately 60 × 60 × 60 cm, have shown survival rates higher than 60% in the mountains in southern Spain (Leverkus et al., 2015).

The use of tube tree shelters is highly recommended since mortality due to herbivory can exceed 90% (Gómez et al., 2003). Tube shelters, especially perforated types, provide physical protection to seedlings, and improve seedling growth (Oliet et al., 2003). Under normal conditions, establishment or maintenance irrigation is unnecessary, although it is important to control vegetation to reduce competition for water during the first years after planting. Nevertheless, irrigation at planting is highly recommended on harsh sites. Fertilization after planting is unnecessary. Inoculation of *Scleroderma citrinum* Pers., a mycorrhizal fungus, through irrigation with spore suspension in outplanted seedlings during the first year has been shown to increase seedling growth (Duñabeitia et al., 2004).

Chapter References

Álvarez, P., Barrio, M., Díaz, R.A., Higueras, J., Riesco, G., Rigueiro, A., , … Villarino, J.J., 2000. Manual de selvicultura de frondosas caducifolias. Serie Manuales Técnicos. Proyecto Columella, Lugo.

Calderón, C., 2005. Influencia de la época de siembra en los parámetros morfológicos de la calidad de planta en vivero de Quercus petraea (Matts.) Liebl. y *Quercus pyrenaica* Willd. In: Actas del IV Congreso Forestal Español. Sociedad Española de Ciencias Forestales, Spain.

Castro, J., Zamora, R., Hódar, J.A., 2006. Restoring *Quercus pyrenaica* forests using pioneer shrubs as nurse plants. App. Veg. Sci. 9 (1), 137–142. https://doi.org/10.1111/j.1654-109X.2006.tb00663.x.

Corredoira, E., Toribio, M., Vieitez, A.M., 2014. Clonal propagation via somatic embryogenesis in *Quercus* spp. In: Ramawat, K.G., Mérillon, J.M., Ahuja, M.R. (Eds.), Tree Biotechnology. Taylor and Francis, UK, pp. 264–302.

Duñabeitia, M., Rodríguez, N., Salcedo, I., Sarrionandia, E., 2004. Field mycorrhization and its influence on the establishment and development of the seedlings in a broadleaf plantation in the Basque Country. For. Ecol. Manage. 195 (1–2), 129–139. https://doi.org/10.1016/j.foreco.2004.02.038.

Gómez, J.M., Garcıa, D., Zamora, R., 2003. Impact of vertebrate acorn-and seedling-predators on a Mediterranean *Quercus pyrenaica* forest. For. Ecol. Manage. 180 (1–3), 125–134. https://doi.org/10.1016/S0378-1127(02)00608-4.

Gomez-Garay, A., Manzanera, J.A., Pintos-Lopez, B., 2014. Embryogenesis in oak species. A review. Forest Syst. 23 (2), 191–198. https://doi.org/10.5424/fs/2014232-0582.

Leverkus, A.B., Castro, J., Delgado-Capel, M.J., Molinas-González, C., Pulgar, M., Marañón-Jiménez, S., , … Querejeta, J.I., 2015. Restoring for the present or restoring for the future: enhanced performance of two sympatric oaks (*Quercus ilex* and *Quercus pyrenaica*) above the current forest limit. Restor. Ecol. 23 (6), 936–946. https://doi.org/10.1111/rec.12259.

Lucas-Villar, F.J., Oliet Palá, J., González Goicoetxea, P., Serrada Hierro, R., pyrenaica, Q., En Pemán, J., Navarro Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R., 2013. Producción y manejo de semillas

y plantas forestales (tomo II). Organismo autónomo Parques Nacionales. Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, España, pp. 278–291.

Montoya, J.M., 1995. Técnicas de reforestación con encinas, alcornoques y otras especies de *Quercus mediterráneos*. En: Forestación en tierras agrícolas. In: Orozco-Bayo, E., Monreal-Montoya, J. (Eds.), Universidad de Castilla-La Mancha, pp. 199–213.

Navarro Cerrillo, R., Serrada Hierro, R., Gómez Mampaso, V., Ocaña Bueno, L., Pemán García, J., Pemán García, J., Navarro-Cerrillo, R.M., Aránzazu Prada Sáez, M., Serrada Hierro, R., 2021. Siembras y plantaciones. En: Bases técnicas y ecológicas del proyecto de repoblación forestal. In: Ministerio para la Transición Ecológica y el Reto Demográfico. Madrid, pp. 722–780. Coords.

Oliet, J.A., Navarro-Cerrillo, R.M., Contreras, O., 2003. Evaluación de la aplicación de tubos y mejoradores en repoblaciones forestales. In: Junta de Andalucía. Consejería de Medio Ambiente.

Pemán, J., Chirino, E., Espelta, J.M., Jacobs, D.F., Martín-Gómez, P., Navarro-Cerrillo, R., Oliet, J.A., Vilagrosa, A., Villar-Salvador, P., Gil-Pelegrín, E., 2017. Physiological keys for natural and artificial regeneration of oaks. In: Gil-Pelegrín, E., Peguero-Pina, J., Sancho-Knapik, D. (Eds.), Oaks Physiological Ecology. Exploring the Functional Diversity of Genus *Quercus* L. Springer International Publishing, pp. 453–511.

Perea, R., Gil, L., 2014. Shrubs facilitating seedling performance in ungulate-dominated systems: biotic versus abiotic mechanisms of plant facilitation. Eur. J. For. Res. 133, 525–534. https://doi.org/10.1007/s10342-014-0782-x.

Rodríguez-Calcerrada, J., Pardos, J.A., Gil, L., Aranda, I., 2007. Summer field performance of Quercus petraea (Matt.) Liebl and Quercus pyrenaica Willd seedlings, planted in three sites with contrasting canopy cover. New For 33, 67–80. https://doi.org/10.1007/S11056-006-9014-7/METRICS.

Torroba-Balmori, P., Zaldívar, P., Alday, J.G., Fernández-Santos, B., Martínez-Ruiz, C., 2015. Recovering *Quercus* species on reclaimed coal wastes using native shrubs as restoration nurse plants. Ecol. Eng. 77, 146–153. https://doi.org/10.1016/j.ecoleng.2015.01.024.

Villar-Salvador, P., Heredia, N., Millard, P., 2010. Remobilization of acorn nitrogen for seedling growth in holm oak (*Quercus ilex*), cultivated with contrasting nutrient availability. Tree Physiol. 30 (2), 257–263. https://doi.org/10.1093/treephys/tpp115.

Vivero, J.L., Prados, J., Hernández-Bermejo, J.E., 2000. Libro Rojo de la Flora Silvestre amenazada de Andalucía. Tomo II: Especies Vulnerables. Quercus pyrenaica Willd.

Quercus robur L.

Barbara Mariotti[1], Valeriu-Norocel Nicolescu[2] and Magnus Löf[3]
[1]Department of Agriculture, Food, Environment and Forestry (DAGRI), University of Florence, Firenze, Italy; [2]Faculty of Silviculture and Forest Engineering, Transilvania University of Braşov, Braşov, Romania; [3]Swedish University of Agricultural Sciences, Southern Swedish Forest Research Centre, Lomma, Sweden

Propagation

Seed propagation

Acorns of *Quercus robur* L. (pedunculate oak) are harvested manually from late September to early November, either directly from the ground or from nets lying on the ground (Löf et al., 2019). The average characteristics of seed lots are summarized in Table 11.59. Acorns should be collected as soon as possible after they drop to reduce the risk of fungal infection. Since germination starts immediately, acorns should be stored as soon as possible if they are not sown after collection. A simple cutting test is often used to obtain a quick visual assessment of acorn soundness during collection (Löf et al., 2019).

Acorns are recalcitrant seeds; therefore, they will not survive drying or freezing during storage and cannot be stored at operational scale for long periods, usually <1 year. They are stored in nurseries at around 0 ± 2°C with high air humidity. Historically, they were stored over the winter in jute sacks submerged in running water, in streams or rivers. Thermotherapy, that is, steaming or submerging in warm water at 41°C for 2.5 h, or fungicides are often employed to decrease infestation by fungi and maintain acorn quality during the storage (Knudsen et al., 2004; Kalemba et al., 2021). The hotter the treatment the shorter the time needed (Kalemba et al., 2021). Immersing acorns in water or "float testing" is commonly used to remove desiccated and insect-damaged acorns from acorn batches. This also increases acorn moisture prior to storage; if implemented prior to seeding, it may improve germination. In general, these pretreatments (thermotherapy, fungicides, and float testing) increases germination to between 60% and 90%. Acorns are either seeded in seedbeds or in plant containers a few cm below the soil surface (Suszka et al., 1994). It is important to protect seed sites from rodents and birds, and keep soil or container growing medium moist. *Q. robur* requires higher

Table 11.59 Characteristics of seed lots of *Quercus robur L.*

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
85–91 (88)	90–100 (97)	118–350 (233)	57–87 (76)

Mean values in brackets.
Banach et al. (2023); Bonner and Karrfalt (2008); Božič et al. (2021); Fernández López et al. (2013); Gradi (1980); Moraldi (2023); Piotto and Noi (2001); Zocca (1999).

soil temperatures than many other tree species for optimal root growth (Knudsen et al., 2004).

Vegetative propagation

Vegetative propagation is mainly used for research, conservation of valuable genotypes, preservation of population genetic biodiversity, reproduction of valuable plants, or ornamental purposes. Cutting, grafting, in vitro micropropagation, or somatic embryogenesis are techniques that have all been tested and used with variable and inconsistently encouraging results (Worrell, 2016). Although vegetative propagation could be useful to overcome a shortage of acorns linked to their recalcitrant nature and the strong masting behavior in some populations, the level of success relative to the production cost likely prohibits commercial scale use in forest nurseries.

Stem cutting propagation protocols usually require softwood and semihardwood cuttings collected from juvenile plants. Higher success is obtained with material taken from 1- to 3-year-old seedlings or, in case of older plants, from hedged plants (Chalupa, 1993; Worrell, 2016). Rooting percentages range between 35% and 80% (Worrell, 2016). Such outcomes were obtained in research trials, however, and achieving the higher success rates (>60%) needed in commercial settings might be unobtainable.

Successful cutting propagation needs rooting under consistently high humidity, usually provided by a fog-producing system. Leaf cuttings are usually collected and inserted in the rooting medium in late spring and early summer (May–July). Before insertion, cuttings are soaked in a hormonal solution with indole-3-butyric acid (IBA) or treated with a talc-based rooting powder containing IBA. The rooting medium can be a mixture of peat and sand (1:1) including combinations of growth hormones and fungicides. A low level of fertilization (slow release fertilizer Osmocote 1.5–2.5 g/L) and a pH < 5.5 are suggested (Spethmann, 2020; Spethmann and Harms, 1993). A new shoot growth of 30–50 cm can be reached by autumn (Chalupa, 1993), and minimal overwinter losses and survival rates between 78% and 94% after transplanting in the nursery have been reported (Worrell, 2016). There are few available data about root quality or field performance of plants obtained by cuttings. Nevertheless, long-term performance of plants from cuttings appears to be at least as good as from seedlings (Worrell, 2016).

Several micropropagation methods have been developed but are used solely for research purposes. A large variety of protocols and techniques recently were reviewed (Meyad et al., 2023). Most protocols involve in vitro multiplication of shoots from axillary buds using embryos from acorns (=zygotic embryos) and seedlings as explants (Meyad et al., 2023), and, less frequently, tissues from mature trees. They provide different success rates, however, depending on the genotype of the individuals. Generally, available techniques are not cost-effective for operational use.

Seedling cultivation

Container plants

Seedling production in containers generally starts between late February and April, depending on climatic conditions. These conditions also influence whether to start production in a greenhouse to protect acorns and emerging seedlings from low temperatures and late frosts (Fig. 11.43). Once the seedlings are established, the containers can be moved outdoors until the end of the growing season. In southern Europe, shading may be necessary during the hottest hours of sunny summer days to protect the seedlings from excessive radiation (shade percentage is between 30% and 50%, depending on latitude and climate).

Acorns can be sown without any previous treatment. They should be placed horizontally or slightly inclined at a depth of 2–3 cm to ensure correct radicle and seedling development from the beginning. As in other *Quercus* species, the size of the acorn can positively affect the intensity of the first flush of shoot growth (García-Cebrián et al., 2003; Bonito et al., 2011; Merchegui et al., 2021). Stored acorns can be removed from cold storage some days (max 7 days) before sowing to select pregerminated acorns with a short radicle; the radicle should not exceed 2–3 cm long to avoid damage during manipulation. If the radicle is longer, its apex can be pruned before sowing to stimulate root proliferation (Pardos et al., 2001; Mucha et al., 2018).

Plants usually emerge 1 month after seeding, but emergence rates depend on weather conditions. Cultivation can be done in containers with a volume of at least 300–400 mL and a depth of 15–18 cm. However, to provide enough rooting volume, especially for the large and rapidly developing taproot, we recommend containers of at least 500 mL, since large containers usually increase the field performance of seedlings (Mariotti et al., 2015a; 2015b). Due to the high germination rate of acorn batches, one acorn per container or cell is enough. If two acorns are sown, only one must be left after emergence. Cultivation density depends on container type and size, and values around 250 seedling m^2 or lower are recommended (Fernández López et al., 2013). Lower cultivation density results in higher growth of *Q. robur* seedling (Fernández López et al., 2013).

Seedlings are frequently cultivated in fertilized blonde peat or mixtures of coir with peat as organic bases, with additions of vermiculite or perlite up to 20% of the total volume. As *Q. robur* is sensitive to drought, short and frequent watering should be applied during the growth phase, ensuring that moisture reaches the bottom of the container. This is important to prevent the roots from growing upwards and then downwards, which can cause a characteristic root deformation of improperly irrigated oak species. During the hardening phase, adjusting the irrigation regime is effective to arrest growth (Fernández López et al., 2013). We recommend surface irrigation but each irrigation event should be at a high watering rate. As a general guideline, when the weight loss of the container exceeds approximately 50% compared with the weight of the pot or

tray with a fully saturated substrate, it indicates the need for irrigation (Fernández López et al., 2013).

For forest restoration or reforestation purposes, one growing season in the nursery, or at most two, is usually enough to produce marketable seedlings of the recommended size (Table 11.60). In southern European countries or areas with favorable environmental conditions, seedlings are outplanted in late fall to facilitate establishment by the higher soil water content during fall and late winter. In cold winter areas seedlings are transferred to cold storage and then outplanted in early spring (Löf et al., 2004, 2006). In moderately cold areas, seedlings can be stored outdoors. High-quality seedlings should have a low number of branches and a well-developed stem with clear apical dominance. Spiraling or other root defects caused by inappropriate container structure should be absent, and the plug must be firm with the root system having a large amount of fine roots.

Bareroot plants

For cultivating bareroot plants, the soil should be prepared with deep plowing to a depth of approximately 40 cm across the entire surface, followed by cross tilling to a depth of around 15–20 cm (Fernández López et al., 2013). This process is useful for applying necessary amendments and fertilizers (both organic and mineral) and the possibility of using fertigation during the cultivation cycle (Fernández López et al., 2013).

Similar to other species with recalcitrant seeds, seeding before early frosts is generally recommended (October–November, depending on local climate conditions). Alternatively, seeding can be delayed to spring, usually when night temperatures do not drop below freezing and late frosts are unlikely. This typically occurs in March or April,

Table 11.60 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Quercus robur*.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	6–80	>45	22–80	>45
Root collar diameter (cm)	0.15–1.04	>0.7	0.6–0.9	>0.7
Shoot dry weight (g)	0.12–11.7	>5.0	2.3–18.8	>8.0
Root dry weight (g)	0.37–11.4	>5.5	3.2–23.2	>9.0
Plant dry weight (g)	0.6–23	>10.5	5.5–42.0	>17.0
Shoot/root dry weight ratio	0.4–1.0	>0.65	0.4–1.3	>0.7
Shoot [N] (mg/g)		>7	6.7–9.7	>7
Root [N] (mg/g)		>6	4.9–9.0	>6

Andersen (2010); Fernández López et al. (2013); Kormanek et al. (2023); Mariotti et al. (2015b); Mijatović et al. (2022); O'Reilly et al. (2008); Schmal et al. (2010).



Figure 11.43 *Quercus robur* seedlings during the first weeks in the nursery. *(Barbara Mariotti.)*

depending on the local climate. To protect the acorns from predators, fencing or netting is highly recommended. Acorns should be sown 3–5 cm below the soil surfaces, in furrows 3–4 cm deep and spaced about 20 cm apart (Fernández López et al., 2013). If sowing is in the fall, deeper sowing is recommended to prevent the exposure of the acorns to air due to heavy rains or alternating freeze/thaw cycles. In northern Europe, we recommend mulching seed beds with a thick layer (8–10 cm) of straw, leaves, or reeds. In Mediterranean countries, seeds are covered with a sterile layer of peat or bark to prevent soil dehydration and weed growth. A roller can be used to compact the soil around the acorns to minimize air spaces that could hinder acorn germination (Fernández López et al., 2013).

Sowing density depends on the method used, whether mechanized sowing in strips or scatter sowing, the latter being less common today. Depending on local practice, 5 to 10 acorns are sown per linear m. Lower cultivation density increases oak seedling nursery growth and improves outplanting performance (Andersen, 2010). Recommended cultivation densities vary between 25 and 50 seedlings/m^2. During cultivation, weed control should be performed regularly, either manually or with selective herbicides targeting narrow-leaved and broad-leaved weeds separately. For seedlings with a woody stem, herbicides based on Clethodim and Pendimethalin can be suitable options (see Kerkez et al., 2025).

Root pruning can improve root system structure and fibrosity and reduce the shoot/root ratio by slowing shoot growth. Roots of *Q. robur* seedlings can be pruned up to 5-6 times, as they develop a strong root system (Fernández López et al., 2013). After lifting, small plants can be transplanted to another bed and cultivated for a second year. The cultivation cycle typically lasts 1 or 2 years, depending on when the seedlings reach a minimum root collar diameter of 0.7 cm and a height of 45–50 cm. During cultivation, the irrigation regime will depend on local weather conditions, soil properties, cultivation density, and meteorological conditions, ranging from two-three days to 7 days a week.

Q. robur seedlings can tolerate early storage without significant negative effects on survival, but premature lifting can reduce outplanting growth (Brønnum, 2005). High quality seedlings should have minimal branching and show a well-developed stem with clear apical dominance. During lifting operations, it is crucial to preserve the integrity of the taproot as much as possible, and seedlings must have a fibrous root system (Starkey, 2019).

Field establishment

The main environmental limitations for seedling establishment are water and light competition from neighboring vegetation, and browsing from hares and ungulates (Davies, 1985; Dey et al., 2008). There are several methods to control competing vegetation. Herbicides are the most cost-efficient but are not always allowed due to environmental concerns, especially for reforestation (Ammer et al., 2011; Fernández López et al., 2013). Mechanical site preparation, prescribed burning, and mulching have all been used to improve oak seedling survival and growth (Löf et al., 2019; Petersson et al., 2020). Young oaks are considered relatively drought-tolerant, but only moderately shade tolerant (Jensen and Löf, 2017). Thus, if overtopped by neighboring vegetation, mortality of planted seedlings may be high.

In the southern edge of the range of the species in Europe, the presence of some vegetation can be beneficial during the initial growth phase, once the young oaks are taller than the competition, by providing protection from wind, sun, and wildlife, while increasing humidity and density (Fernández López et al., 2013). These benefits can result in straighter stems, thinner branches, and greater height growth. Some associated species, such as *Rubus ulmifolius* and *Pteridium aquilinum*, can inhibit regeneration but also provide some protection against browsing. The presence of other tree species or some tall shrubs such as *Cytisus* spp., which do not compete strongly for nutrients or water, can also be beneficial. In contrast, *Ulex* spp. or *Erica* spp. compete strongly with young seedlings due to their extensive root systems, so they must be controlled. It is sufficient to clear an area around the oak two-thirds of its height.

Seedlings grow deep taproots in suitable soil conditions, which increases their resistance to soil drought (Löf, 2000; Löf and Welander, 2004). Therefore, they should be planted in deep soils with intense site preparation such as mounding or repeated herbicide treatment to clear the site from competing vegetation (Löf et al., 2006). Under acceptable growing conditions, *Q. robur* seedlings can tolerate moderate browsing and other disturbances due to their capacity for resprouting (Löf, 2000). In addition to competing with the oak seedlings for resources, neighboring vegetation can provide habitat for voles and other rodents, which may clip seedlings below or above ground during winter and especially under snow cover. During years with high vole populations, this may cause significant mortality in planted seedlings. Normally, one or

maximum 2-year seedlings are used according to growing condition and latitude, both bareroot and container seedlings. Seedlings can be planted both during autumn and in spring.

Expensive fences are often needed to protect plantations from browsing animals. While fencing effectively reduces browsing damage it can be impractical and costly, especially if relatively low numbers of seedlings must be established. However, group plantings (clusters) with low oak densities within groups may reduce costs to some extent (Saha et al., 2017). Tree shelters are an alternative tool to protect seedlings from browsing that may also provide a favorable microclimate (Redick and Jacobs, 2020). Tree shelters can also promote apical dominance and lower branch development. However, shelters can reduce plant biomass and favor allocation to the shoot, unbalancing the shoot/root ratio (Mariotti et al., 2015c). Thus, in harsher conditions, their use should be weighed accordingly.

The application of fertilizer is beneficial, as it enhances resilience and contributes to establishment and growth of the plant. In soils deficient in P and K but rich in N, the recommended fertilizer should contain P and K two to three times higher than N (Fernández López et al., 2013). The use of slow-release fertilizers, available as encapsulated granules or tablets is an option. Fertilization should be applied manually to each tree in the planting hole at depth, which reduces the overall amount of fertilizer used and minimizes weed growth (Fernández López et al., 2013).

Q. robur has weak apical dominance, so starting with high planting densities (Fig. 11.44) can promote the development of straight-stemmed trees. In France and Germany, high densities are achieved through natural regeneration of mature stands, often starting with hundreds of thousands of oaks and shade species/ha. For plantations intended to produce quality timber, initial overall densities should be high. In central Europe, typical densities range from 4000 to 10,000 trees/ha, with one-third to one-half being accessory shade species that help the oaks grow straight (Fernández López



Figure 11.44 1 + 0 *Quercus robur* seedlings with and without shelter, 2 years after outplanting in the field. *(Barbara Mariotti.)*

et al., 2013). In France and Belgium, recommended densities for reforestation are less than 2000 trees/ha, with spacing of 1.5 × 1.5 m or 2.5 × 1.5–2 m, as larger spacing complicates future tree selection (Fernández López et al., 2013). High densities with long rotations increase establishment costs and a balance must be found between silvicultural needs and economic considerations. For northern Spain suggested densities are around 1800–2550 trees/ha (Fernández López et al., 2013). Mixing with other species is recommended to increase biodiversity and stability, while providing early income from harvesting fast-growing secondary species.

Chapter References

Ammer, C., Balandier, P., Scott-Bentsen, N., Coll, L., Löf, M., 2011. Forest vegetation management under debate – an introduction. Eur. J. For. Res. 130, 1–5. https://doi.org/10.1007/s10342-010-0452-6.

Andersen, L., 2010. Spacing in the nursery seedbed and subsequent field performance of *Quercus robur* L. and *Fagus sylvatica* L. Eur. J. Hortic. Sci. 75.

Banach, J., Kormanek, M., Malek, S., Durlo, G., Skrzyszewska, K., 2023. Effect of the changing seedlings density of *Quercus robur* L. grown in nursery containers on their morphological traits and planting suitability. Sylwan 167.

Bonito, A., Varone, L., Gratani, L., 2011. Relationship between acorn size and seedling morphological and physiological traits of *Quercus ilex* L. from different climates. Photosynthetica 49, 75–86. https://doi.org/10.1007/s11099-011-0014-2.

Bonner, F.T., Karrfalt, R.P. (Eds.), 2008. The Woody Plant Seed Manual. Agricultural Handbook 727. U. S. Department of Agriculture, Forest Service, Washington, DC.

Božič, G., Westergren, M., Železnik, P., Konrad, H., Nagy, L., Lanšćak, M., Novčić, Z., Rukavina, S., Schueler, S., Stojnić, S., 2021. Planting Material Use and Conservation Guidelines - Interreg Refocus. https://dtp.interreg-danube.eu/uploads/media/approved_project_output/0001/46/76b57ed77c-be776929842edba65584e002889384.pdf. (Accessed 5 October 2024).

Brønnum, P., 2005. Assessment of seedling storability of *Quercus robur* and *Pinus sylvestris*. Scand. J. For. Res. 20. https://doi.org/10.1080/02827580410019526.

Chalupa, V., 1993. Vegetative propagation of oak (*Quercus robur* and *Q. petraea*) by cutting and tissue culture. Ann. des Sci. For. 50. https://doi.org/10.1051/forest:19930730.

Davies, R.J., 1985. The importance of weed control and the use of tree shelters for establishing broadleaved trees on grass-dominated sites in England. Forestry 58. https://doi.org/10.1093/forestry/58.2.167.

Dey, D.C., Jacobs, D., McNabb, K., Miller, G., Baldwin, V., Foster, G., 2008. Artificial regeneration of major oak (*Quercus*) species in the eastern United States - a review of the literature. For. Sci. https://doi.org/10.1093/forestscience/54.1.77.

Fernández López, M.J., Barrio Anta, M., Álvarez Álvarez, P., López Varela, B., Gómez Fernández, J.A., 2013. *Quercus petraea* (Matt.) Liebl. And *Quercus robur* L. In: Alía, R., Nuria, M., Monfort, A., Leal, D.A., Sauce, S.I. (Eds.), Producción y Manejo de Semillas y Plantas Forestales. Organismo Autónomo Parques Nacionales Ministerio De Medio Ambiente, pp. 251–278.

García-Cebrián, F., Esteso-Martínez, J., Gil-Pelegrín, E., 2003. Influence of cotyledon removal on early seedling growth in *Quercus robur* L. Ann. For. Sci. 60 (1), 69–73.

Gradi, A., 1980. Vivaistica forestale. In: Edagricole, Bologna (Italy).

Jensen, A.M., Löf, M., 2017. Effects of interspecific competition from surrounding vegetation on mortality, growth and stem development in young oaks (*Quercus robur*). For. Ecol. Manage. 392, 176–183. https://doi.org/10.1016/j.foreco.2017.03.009.

Kerkez, I., Oliet, J.A., Lösing, H., 2025. *Fagus sylvatica*. In: Stanturf, J., Villar-Salvador, P., Mariotti, B., Ivetić, V., Madsen, P., Montagnoli, A., Andivia, E., Bebre, I., Dimitrova, A., Klisz, M. (Eds.), Guidelines for Climate Adaptive Forest, Restoration and Reforestation Projects. Elsevier.

Knudsen, I.M.B., Thomsen, K.A., Jensen, B., Poulsen, K.M., 2004. Effects of hot water treatment, biocontrol agents, disinfectants and a fungicide on storability of English oak acorns and control of the pathogen, *Ciboria batschiana*. For. Pathol. 34. https://doi.org/10.1046/j.1439-0329.2003.00348.x.

Kalemba, E.M., Wawrzyniak, M.K., Suszka, J., Chmielarz, P., 2021. Thermotherapy and storage temperature manipulations limit the production of reactive oxygen species in stored pedunculate oak acorns. Forests 12, 1338. https://doi.org/10.3390/f12101338.

Kormanek, M., Małek, S., Banach, J., Durło, G., 2023. Effect of changing substrate density and water application method on substrate physical properties and container-grown seedling growth. Forests 14. https://doi.org/10.3390/f14071490.

Löf, M., 2000. Establishment and growth in seedlings of *Fagus sylvatica* and *Quercus robur*: influence of interference from herbaceous vegetation. Can. J. For. Res. 30, 855–864. https://doi.org/10.1139/cjfr-30-6-855.

Löf, M., Castro, J., Engman, M., Leverkus, A.B., Madsen, P., Reque, J.A., Villalobos, A., Gardiner, E.S., 2019. Tamm Review: direct seeding to restore oak (*Quercus* spp.) forests and woodlands. For. Ecol. Manage. https://doi.org/10.1016/j.foreco.2019.06.032.

Löf, M., Dey, D.C., Navarro, R.M., Jacobs, D.F., 2019. Mechanical site preparation for forest restoration. New For. 43, 825–848. https://doi.org/10.1007/s11056-012-9332-x.

Löf, M., Rydberg, D., Bolte, A., 2006. Mounding site preparation for forest restoration: survival and short term growth response in *Quercus robur* L. seedlings. For. Ecol. Manage. 232. https://doi.org/10.1016/j.foreco.2006.05.003.

Löf, M., Thomsen, A., Madsen, P., 2004. Sowing and transplanting of broadleaves (*Fagus sylvatica* L., *Quercus robur* L., *Prunus avium* L. and *Crataegus monogyna* Jacq.) for afforestation of farmland. For. Ecol. Manage. 188. https://doi.org/10.1016/j.foreco.2003.07.013.

Löf, M., Welander, N.T., 2004. Influence of herbaceous competitors on early growth in direct seeded *Fagus sylvatica* L. and *Quercus robur* L. Ann. For. Sci. 61, 781–788. https://doi.org/10.1051/forest:2004075.

Mariotti, B., Maltoni, A., Chiarabaglio, P.M., Giorcelli, A., Jacobs, D.F., Tognetti, R., Tani, A., 2015a. Can the use of large, alternative nursery containers aid in field establishment of *Juglans regia* and *Quercus robur seedlings*? N. For. 46, 773–794. https://doi.org/10.1007/s11056-015-9505-5.

Mariotti, B., Maltoni, A., Jacobs, D.F., Tani, A., 2015b. Container effects on growth and biomass allocation in *Quercus robur* and *Juglans regia* seedlings. Scand. J. For. Res. 30, 401415. https://doi.org/10.1080/02827581.2015.1023352.

Mariotti, B., Maltoni, A., Jacobs, D.F., Tani, A., 2015c. Tree shelters affect shoot and root system growth and structure in *Quercus robur* during regeneration establishment. Eur. J. For. Res. 134, 641–652.

Merchegui, T., Pardos, M., Jacobs, D.F., 2021. Effect of acorn size on survival and growth of Quercus suber L. seedlings under water stress. Eur. J. For. Res. 140, 175–186.

Meyad, C., Henkrar, F., Bouhaddou, N., Khabar, L., 2023. Micropropagation of *Quercus* spp., complications and solutions—an overview. Vitr. Cell. Dev. Biol. - Plant. https://doi.org/10.1007/s11627-023-10354-4.

Mijatović, L., Pavlović, S., Jović, L., Devetaković, J., Vilotić, D., 2022. Potential of new planting container in *Quercus robur* seedlings production–first report. Reforestat. Challeng. 14, 1–8. https://doi.org/10.21750/R.

Moraldi, M., 2023. Compendio di Vivaistica Forestale. ARSIAL Regione Lazio, Roma (Italy).

Mucha, J., Jagodziński, A.M., Bułaj, B., Łakomy, P., Talaśka, A.M., Oleksyn, J., Zadworny, M., 2018. Functional response of *Quercus robur* L. to taproot pruning: a 5-year case study. Ann. For. Sci. 75, 1–12. https://doi.org/10.1007/s13595-018-0708-8.

O'Reilly, C., De Atrip, N., Doody, C., O'Leary, D., Doody, P., Thompson, B., 2008. Increasing the yield and quality of broadleaf planting stock through higher N fertilisation in the nursery. Ir. Times.

Pardos, M., Pardos, J.A., Montero, G., 2001. Growth responses of chemically root-pruned cork oak seedlings in the nursery. J. Environ. Hortic. 19 (2), 69–72. https://doi.org/10.24266/0738-2898-19.2.69.

Petersson, L.K., Dey, D.C., Felton, A.M., Gardiner, E.S., Löf, M., 2020. Influence of canopy openness, ungulate exclosure, and low-intensity fire for improved oak regeneration in temperate Europe. Ecol. Evol. 10, 2626–2637. https://doi.org/10.1002/ece3.6092.

Piotto, B., Noi, D., 2001. Propagazione per seme di alberi e arbusti della flora mediterranea, ed. ANPA Agenzia Nazionale per la Protezione dell'Ambiente Dipartimento Prevenzione e Risanamento Ambientali.

Redick, C.H., Jacobs, D.F., 2020. Mitigation of deer herbivory in temperate hardwood forest regeneration: a meta-analysis of research literature. Forests 11, 1220. https://doi.org/10.3390/f11111220.

Saha, S., Kuehne, C., Bauhus, J., 2017. Lessons learned from oak cluster planting trials in central Europe. Can. J. For. Res. https://doi.org/10.1139/cjfr-2016-0265.

Schmal, J.L., Jacobs, D.F., Reilly, C.O., 2010. Nitrogen Budgeting and Plant Quality of Exponentially Fertilized Bareroot *Quercus robur* Seedlings in Ireland, pp. 1–13.

Spethmann, W., 2020. Long cuttings: a novel method to propagate woody plants. In: Caula, A., Beyl, R.N. T. (Eds.), Plant Propagation Concepts and Laboratory Exercises. https://doi.org/10.1201/b17340-23.

Spethmann, W., Harms, P., 1993. Influence of fertilized substrate on rooting and growth of oak cuttings. Ann. des Sci. For. 50. https://doi.org/10.1051/forest:19930731.

Starkey, T., 2019. Hardwood seedling growth and development and the impact of pruning and environmental stresses. In: McNabb, K., Pike, C.C. (Eds.), A Nursery Guide for the Production of Bareroot Hardwood Seedlings, vol 733. U.S. Department of Agriculture, Forest Service Agriculture Handbook, pp. 89–100.

Suszka, B., Muller, C., Masimbert, M., 1994. Seeds of forest broadleaves from harvest to sowing. Institut National de la Recherche Agronomique, second ed. https://doi.org/10.1093/forestscience/43.4.602

Worrell, R., 2016. Vegetative Propagation of Oak – what Are the Best Options?.

Zocca, A., 1999. La propagazione di alberi e arbusti, Edagricole. ed. Bologna (Italy).

Quercus suber L.

Enrique Andivia[1], Rafael M. Navarro Cerrillo[2], Luna Morcillo[3], Alberto Vilagrosa[3] and Vasco Paiva[4]
[1]Department of Biodiversity, Ecology and Evolution, Faculty of Biological Sciences, Universidad Complutense de Madrid, Madrid, Spain; [2]Department of Forestry, Faculty of Agriculture and Forestry, University of Córdoba, DendrodatLab-ERSAF, Laboratory of Dendrochronology, Silviculture and Climate Change, Córdoba, Spain; [3]Mediterranean Center for Environmental Studies (CEAM Foundation), Joint Research Unit, University of Alicante-CEAM, University of Alicante, Alicante, Spain; [4]Universidade de Trás-os-Montes e Alto Douro (UTAD), Coimbra, Portugal

Propagation

Seed propagation

Acorn collection usually takes place between the last weeks of November and December, although *Quercus suber* (cork oak) presents a broad fruiting period from October to January (Navarro-Cerrillo et al., 2013). The acorns of this species show annual and biennial maturation, with biennial maturation being more common in areas with shorter vegetative periods (Elena et al., 1993). As a result, three types of acorns have been described (Montero and Cañellas, 1999): early acorns, from late September, accounting for between 5% and 10% of the final acorn production; mid-season acorns, from November, with high germination potential and representing 65% of the production; and late acorns, between January and February, with low germination potential and comprising 25% of the production.

The most suitable harvesting time is when the acorns turn a dark brown color, without traces of green areas. This procedure ensures high germination rates by discarding immature embryos and minimizing acorn predation. Collection is done manually from the ground after shaking trees. The collection of spontaneously fallen acorns directly from the ground is not recommended. The processing and storage of acorns follow similar recommendations to those for other *Quercus* species. We recommend immersing the acorns in hot water (45°C) for about 2 hours to eliminate any potential presence of beetle and moth larvae. In addition, any acorns that float should be discarded. We recommend sowing acorns after immersion, but if acorns need to be stored then they should be dried until they reach 40% moisture content. This can be achieved by drying them in chambers with a relative humidity of 30% and a temperature of 30°C for 24–36 h (Merouani et al., 2001). Then, the acorns should be stored in 50 μm thick polyethylene bags inside open containers that allow air circulation in a refrigerated chamber at a temperature of 1–2°C.

The acorns of this species exhibit high variability in size and shape, factors that influence seedling development. Therefore, it is recommended to select acorns weighing over 5 g to ensure adequate nutrient reserves and seedling quality (Navarro-Cerrillo et al., 2013). This species does not require pregermination treatment, although hydration before planting is highly recommended by soaking them in water for 48 h. Overall, germination trials with this species following these recommendations have shown very positive results, with an average germination rate of around 80% (Table 11.61).

Table 11.61 Characteristics of seed lots of *Quercus suber*.

Purity (%)	No. seeds/kg	Germination capacity (%)
82–100 (99)	70–347 (194)	50–100 (78)

Mean values in brackets.
References and technical documents included in Navarro-Cerrillo et al. (2013); Moreno Moraldi et al. (2024)

Vegetative propagation

The recalcitrant nature of *Q. suber* acorns, both for seed conservation and morphogenic capacity, has hindered the vegetative propagation of this species (Meyad et al., 2023). Asexual reproduction of *Q. suber*, however, has been developed from cuttings/grafting, micropropagation, and somatic embryogenesis (Bueno et al., 1992; Hernández et al., 2003). The latter presents many advantages over organogenesis from other materials such as grafting, zygotic embryos, or leaves from seedlings and adult trees. Somatic embryogenesis is an alternative for the mass propagation of *Q. suber* seedlings with desired characteristics. The applications of this propagation method to *Q. suber* are numerous and have been widely recognized (Corredoira et al., 2014), but it requires specific cultivation protocols in the nursery to obtain elite genotypes. Advantages are for seedling cultivation and for planting disease-tolerant/resistant materials (e.g., *Phytophthora cinnamomi*). Recent work has overcome some difficulties of the propagation method, as well as the possible generalization of grafts. Thus, Peñuelas Rubira and Benito Matías (2018) defends the "domestication" of *Q. ilex* and *Q. suber* to increase the establishment rate of plus plants. However, *Q. suber* cultivation through seeds will continue to be the main propagation method to meet the demand for productive plantations and reforestation (Chirino et al., 2008a).

Seedling cultivation

Container plants

Q. suber seedlings are only produced in containers. Sowing can be carried out immediately after acorn collection (fall and winter). Since this species exhibits rapid height growth, early sowing in nurseries located in warm climates (with an average annual temperature above 16 or 17°C) is not recommended, especially when using large acorns. In such cases, sowing in February with acorns that have been stored according to previously mentioned protocols is recommended (Navarro-Cerrillo et al., 2013). The sowing is done with the apex downwards, at an angle of about 45 degrees, and covering the acorn with approximately 1 cm of growth media (Chouial and Benamirouche, 2016). In many nurseries, two acorns are sown per cell, with one of the two seedlings being selected as

soon as possible. Germination takes between 4 and 6 weeks and depends on nursery conditions. In areas with high risk of frosts, initial use of greenhouses or heating systems is advisable. The recommended cultivation period is about 35 weeks. Due to its tendency for height growth, the use of shading nets is not recommended, except in the early germination stages. In these cases, nets should be white.

The cultivation of *Q. suber* seedling requires the use of containers with volumes of 300 cm^3 or 400 cm^3 (Chirino et al., 2008a), although in Portugal the recommended size of containers for this species is 200 cm^3. Nevertheless, we recommend containers with volume of 400 cm^3 because of the rapid height growth this species exhibits. Large containers are also associated with lower planting densities and seedlings with more balanced shoot to root ratios (Benamirouche et al., 2020). This target seedling (1 + 0) has proven to optimize the success of planting for forest restoration (Pardos et al., 2002).

For the cultivation of high functional quality seedlings, older stock types can be considered when environmental conditions are especially favorable. This type of seedling requires using larger container volumes (1000 to 3500 cm^3) to avoid problems with root malformation and achieve adequate root architecture. Using long containers (e.g., 30 cm long and 590 cm^3 volume) compared with standard containers (e.g., 18 cm long) favors the development of a long tap root, which is important for this species to maintain the capacity to resprout and to colonize deep soil horizons (Chirino et al., 2008b). Indeed, these seedlings were able to develop deep roots in a short time in the field and maintained a high capacity for water transport to the leaves throughout the root system.

Recommended substrate should have high porosity to promote adequate root system development such as blonde peat, humus peat, coconut coir, or pine bark as the main organic component. Given the variety of components available, it is possible to formulate growing media with the desired characteristics according to container, irrigation, fertilization, and cultivation schedule. *Q. suber* seedlings require the use of growing media with high porosity to promote root growth and fibrosity. The most common mixtures for plants (1 + 0) typically used for forest restoration have been peat: perlite (3:1 vol.), or peat: coconut coir: perlite (6:3:1 vol.), although there is a trend to progressively replace peat with coir (Mariotti et al., 2023). Some other growing media showing good results include sand mixed with peat and coir (1:1:1 vol.). Innovative materials have been also assessed in growing media for this species. Chirino et al. (2011) studied the impact of water retention in various polymers and clay types combined with the substrate. The main findings indicated that small amounts of these polymers increased water retention capacity and seedling survival after outplanting. In the case of long-term cultivation (2 + 0, 1 + 1), growing media should be formulated using coconut coir, *Sphagnum* peat, black peat, perlite with a controlled release fertilizer (>3 g/L) (pH 5.5 to 6.2, electrical conductivity 120 to 159 mS/m, particle size 0/20 mm, apparent density

300–350 kg/m^3). The pH should be slightly acidic regardless of the selected substrate since alkaline substrates may lead to Fe and P limitations.

Similar to other oaks, acorn nutritional reserves support seedling growth during the first months of cultivation, after which fertilization should be provided to prevent nutrient limitations (del Campo, 2002). *Q. suber* seedlings are especially sensitive to N deficiency (Navarro Cerrillo et al., 2003), yet nutritional status has shown contrasting effects on seedling survival (del Campo, 2002; Trubat et al., 2010). Accordingly, and considering studies on nutrient remobilization from acorns in Mediterranean oaks (Villar-Salvador et al., 2010), we recommend providing fertilization during the second shoot flush of growth using NPK fertilizers at around 12-24-08 (%) during the growing phase and 8–6–20 (%) during hardening (Navarro-Cerrillo et al., 1998). Slow release fertilizers should be applied carefully to avoid undesirable seedling growth in the nursery, affecting plant quality (Hernández et al., 2009). They can be used at rates of 1.8–2 kg/m^3 of substrate, with NPK 15–9–11 fertilizer type. Phosphoric fertilization has been recommended for the prevention of damage by *Phytophthora cinnamomi* both in nurseries and during field establishment (Navarro-Cerrillo et al., 2004). Fertilizer enriched with phosphorus and potassium during the last phases of the nursery period did not improve morphology or functional seedling characteristics (Chirino et al., 2004). A traditional practice in Portugal is to submerge the plug of seedlings before planting in a solution with 10–12% of N and 50–52% of P_2O_5 to stimulate root growth.

Irrigation can be used, along with low planting density, to control excessive aboveground growth. However, drought hardening should be avoided since some studies showed a negative effect on seedling establishment (del Campo, 2002). However, limitations in the watering regime during the nursery period produced more balanced seedlings in terms of plant biomass and size, with better results in the field (Chirino et al., 2004, Chirino et al., 2008a). In addition, some studies recommend the addition of mycorrhiza in the nursery to improve outplanting results, especially in degraded areas (Maghnia et al., 2019 and refs. therein). Seedlings are stored outdoors in the nursery. Observed and recommended morphological and physiological characteristics of *Q. suber* container stock are shown in Table 11.62. In this species, seedlings with signs of damage, with multiple stems, with root deformation, or very high shoot:root should be avoided (Navarro-Cerrillo et al., 2013).

Field establishment

Water stress, seed predation, and herbivory are the main limiting factors for cork oak regeneration across the species range (Arosa et al., 2015). In the last decade, some studies have pointed to the vulnerability of *Q. suber* seedlings to pests, especially to *Phytophthora cinnamomi*, which strongly limits the survival of seedlings (Domínguez-Begines et al.,

Table 11.62 Observed morphological and physiological attributes and recommended values of high-quality 1 + 0 container seedlings of *Quercus suber*.

Attributes	Observed values	Recommended values
Shoot height (cm)	13–36	25–60
Root collar diameter (cm)	0.28–0.58	>0.3
Shoot dry weight (g)	0.8–3.8	>3
Root dry weight (g)	2.5–7.5	>7
Plant dry weight (g)	4–11	>10
Shoot/root dry weight ratio	0.2–0.5	>0.8
Shoot [N] (mg/g)		>12
Shoot [P] (mg/g)		>1
Shoot [K] (mg/g)		>4

Chirino et al. (2004, 2008a); Hernández et al. (2009); Trubat et al. (2010); Navarro-Cerrillo et al. (2013); Morcillo et al. (2020).

2021). Planting has been widely used and recommended as main establishment method for *Q. suber*. Seeding also has been widely used, yet differences in survival between establishment methods are contingent upon environmental context, which precludes specific guidelines (González–Rodríguez et al., 2011). However, acorn predation after seeding is very high, which greatly limits the success of reforestation programs using this method. For seeding, the recommendation is to place between two and three acorns per hole. Both, planting and seeding, are usually performed manually. As for other Mediterranean oaks, plantation should be performed during the wet season in autumn, especially on sites with mild winters (Andivia et al., 2011).

The outplanting survival of *Q. suber* can be increased using high-intensity soil preparation techniques (Cortina et al., 2009), although recommended techniques are contingent upon soil conditions (Löf et al., 2012). In abandoned croplands, the recommended soil preparation techniques are subsoiling and deep plowing. In forest soils, the recommended techniques are mechanized holes. Whatever the technique, it is recommended to reach planting hole depths between 0.35 and 0.9 m (Cortina et al., 2009). Existing vegetation should be removed using clearing or tillage (Fig. 11.45). In addition, controlling competing vegetation the first years after outplanting can also improve seedling performance. However, seedlings are highly vulnerable to excessive radiation thus nurse shrubs, such as *Retama* spp., can improve seedling survival, especially under more arid conditions or high herbivore pressure (Simonson et al., 2014). In mixed shrublands, with seeder species (as *Cistus* spp.) or resprouter species (as *Erica* spp.), the proximity of seeders can negatively affect seedling survival while resprouters can favor plant establishment (Pérez-Devesa et al., 2008).

Planting density for this species varies widely (Montero and Cañellas, 1999). High densities have been recommended for productive stands or restoration plantations (>1000 seedlings/ha). However, most authors recommend planting densities lower than 600 trees/ha (approximately 4 × 4 m spacing) (Montero and Cañellas, 1999;

Navarro-Cerrillo et al., 2013) (Fig. 11.45). For the restoration of *dehesa* and *montados* agroforestry systems, a density close to the final one (around 100 to 200 trees/ha) is recommended, or an adequate tree management plan that ensures the sustainability of the structure and function of these ecosystems (Simões et al., 2016).

There is no consensus regarding the use of tree shelters (Fig. 11.45). Some authors highly recommend it in *Q. suber* afforestation projects, as they not only provide physical protection to the seedling against predation, radiation, or desiccation, but also improve rooting and height growth rates (Mercheguit and Pardos, 2018; El Alami et al., 2023). Some others, nevertheless, found some disadvantages for establishment because seedlings tend to be slender and etiolated under light-limiting conditions in the shelters (Quilhó et al., 2003). To prevent damage from livestock herbivory the extension of brush piles around planted seedlings, especially using spiny species, may be a valid alternative to deter cattle and deer, as observed in other oak species (Weitkamp et al., 2001).

Irrigation at planting is highly recommended, especially on harsh sites. Irrigation during the first summer season, with three to four abundant irrigations, has greatly improved the performance of *Q. suber* seedlings (Pérez-Ramos and Marañón, 2009). Some authors found more effective watering when applied copiously on each seedling immediately after planting and, when possible, two or three more times during the first 2 years (Cortina et al., 2009). Mulching with cereal straw or other suitable materials has been shown to reduce direct evaporation and decrease transpiration by hampering the establishment of competing herbaceous vegetation. This technique may improve seedling survival and growth (Lucas-Borja et al., 2019). Fertilization supply in the field is not a common practice in *Q. suber* plantations, but it can be recommended for poor soils.



Figure 11.45 Planted *Quercus suber* seedlings with individual tree shelters. *(Pedro Villar-Salvador.)*

Chapter References

Andivia, E., Fernández, M., Vázquez-Piqué, J., 2011. Autumn fertilization of *Quercus ilex* ssp. *ballota* (Desf.) Samp. nursery seedlings: effects on morpho-physiology and field performance. Ann. For. Sci. 68, 543–553. https://doi.org/10.1007/s13595-011-0048-4.

Arosa, M.L., Ceia, R.S., Costa, S.R., Freitas, H., 2015. Factors affecting cork oak (*Quercus suber*) regeneration: acorn sowing success and seedling survival under field conditions. Plant Ecolog. Divers 8 (4), 519–528. https://doi.org/10.1080/17550874.2015.1051154.

Benamirouche, S., Chouial, M., Guechi, W., 2020. Radicle length and container size effects on root deformities in the Mediterranean oak *Quercus suber* L. Bois Forests des Tropiques 343, 17–26. https://doi.org/10.19182/bft2020.343.a31669.

Bueno, M.,A., Astorga, R., Manzanera, J.A., 1992. Plant regeneration through somatic embryogenesis in *Quercus suber* L. Physiol Plant 85, 30–34. https://doi.org/10.1111/j.1399-3054.1992.tb05259.x.

Chirino, E., Vilagrosa, A., Rubio, E., 2004. Efectos de la reducción del riego y la fertilización en las características morfológicas de *Quercus suber*. Cuadernos de la Sociedad Española de Ciencias Forestales 17, 51–56.

Chirino, E., Vilagrosa, A., Vallejo, V.R., 2008a. Técnicas de vivero orientadas a mejorar la calidad de brinzales de *Quercus suber* para restauración de alcornocales mediterráneos degradados. Cuadernos de la Sociedad Española de Ciencias Forestales 28, 163–169.

Chirino, E., Vilagrosa, A., Vallejo, V.R., 2011. Using hydrogel and clay to improve the water status of seedlings for dryland restoration. Plant Soil 344, 99–110. https://doi.org/10.1007/s11104-011-0730-1.

Chirino, E., Vilagrosa, A., Hernández, E.I., Matos, A., Vallejo, V.R., 2008b. Effects of a deep container on morpho-functional characteristics and root colonization in *Quercus suber* L. seedlings for reforestation in Mediterranean climate. For Ecol. Manage 256 (4), 779–785. https://doi.org/10.1016/j.foreco.2008.05.035.

Chouial, M., Benamirouche, S., 2016. Effects of sowing methods on growth and root deformations of containerized cork oak (*Quercus suber* L.) seedlings in nursery. Ecol. Mediterr. 42 (1), 21–28.

Corredoira, E., Toribio, M., Vieitez, A.M., 2014. Clonal propagation via somatic embryogenesis in *Quercus* spp. In: Ramawat, K.G., Mérillon, J.M., Ahuja, M.R. (Eds.), Tree Biotechnology. Taylor and Francis, UK, pp. 264–302.

Cortina, J., Pérez-Devesa, M., Vilagrosa, A., Abourouh, M., Messaoudène, M., Berrahmouni, N., , … Khaldi, A., 2009. Field techniques to improve cork oak establishment. In: Aronson, J., Pereira, J.S., Pereira, J.S., Pausas, J.G., Pausas, J.G. (Eds.), Cork Oak Woodlands: Ecology, Adaptive Management, and Restoration of an Ancient Mediterranean Ecosystem. Island Press, Washington, DC, pp. 141–149.

Del Campo, A., 2002. Régimen de cultivo, desarrollo en vivero, calidad de planta y respuesta al establecimiento en cuatro especies de frondosas mediterráneas. Tesis Doctoral. Universidad de Córdoba.

Domínguez-Begines, J., Ávila, J.M., García, L.V., Gómez-Aparicio, L., 2021. Disentangling the role of oomycete soil pathogens as drivers of plant–soil feedbacks. Ecology 102 (8), e03430. https://doi.org/10.1002/ecy.3430.

El Alami, S.L., El Aboudi, A., El Antry, S., El Mnouar, E.A., Dallahi, Y., Rabhi, A., 2023. Effect of tree shelters and regeneration method on survival and growth of cork oak plantations in the Maamora forest, Morocco. J. Ecol. Eng. 24, 360–374. https://doi.org/10.12911/22998993/165784.

Elena, J.A., Río, J.M., García, J.L., Santamaría, I.G., 1993. Ecological aspects of the floral phenology of the cork-oak (*Quercus suber* L.): why do annual and biennial biotypes appear? Ann. For. Sci. 50, 114–121. https://doi.org/10.1051/forest:19930710.

González-Rodríguez, V., Navarro-Cerrillo, R.M., Villar, R., 2011. Artificial regeneration with *Quercus ilex* L. and *Quercus suber* L. by direct seeding and planting in southern Spain. Ann. For. Sci. 68, 637–646. https://doi.org/10.1007/s13595-011-0057-3.

Hernández, I., Celestino, C., Toribio, M., 2003. Vegetative propagation of *Quercus suber* L. by somatic embryogenesis. I: factors affecting the induction in leaves from mature cork oak trees. Plant Cell Rep. 21, 759–764. https://doi.org/10.1007/s00299-003-0605-x.

Hernández, E.I., Vilagrosa, A., Luis, V.C., Llorca, M., Chirino, E., Vallejo, V.R., 2009. Root hydraulic conductance, gas exchange and leaf water potential in seedlings of *Pistacia lentiscus* L. and *Quercus suber*

L. grown under different fertilization and light regimes. Environ. Exp. Bot. 67, 269–276. https://doi.org/10.1016/j.envexpbot.2009.07.004.

Löf, M., Dey, D.C., Navarro Cerrillo, R.M., Jacobs, D.F., 2012. Mechanical site preparation for forest restoration. New For. 43, 825–848. https://doi.org/10.1007/s11056-012-9332-x.

Lucas-Borja, M.E., González-Romero, J., Plaza-Álvarez, P.A., Sagra, J., Gómez, M.E., Moya, D., Cerdà, A., de Las Heras, J., 2019. The impact of straw mulching and salvage logging on post-fire runoff and soil erosion generation under Mediterranean climate conditions. Sci. Total Environ. 654, 441–451. https://doi.org/10.1016/j.scitotenv.2018.11.161.

Maghnia, F.Z., Abbas, Y., Mahé, F., Prin, Y., El Ghachtouli, N., Duponnois, R., Sanguin, H., 2019. The rhizosphere microbiome: a key component of sustainable cork oak forests in trouble. For Ecol. Manage 434, 29–39. https://doi.org/10.1016/j.foreco.2018.12.002.

Mariotti, B., Oliet, J.A., Andivia, E., Tsakaldimi, M., Villar-Salvador, P., Ivetić, V., , … Cocozza, C., 2023. A global review on innovative, sustainable, and effective materials composing growing media for forest seedling production. Curr For Rep 9 (6), 413–428. https://doi.org/10.1007/s40725-023-00204-2.

Merouani, H., Branco, C., Almeida, M.H., Pereira, J.S., 2001. Effects of acorn storage duration and parental tree on emergence and physiological status of Cork oak (*Quercus suber* L.) seedlings. Ann. For. Sci. 58, 543–554. https://doi.org/10.1051/forest:2001144.

Mechergui, T., Pardos, M., 2018. Effects of moderate water stress and shading on survival, growth and resource allocation of two Cork oak (Quercus suber L.) provenances. Revue d'Ecologie 73, 180–190.

Meyad, C., Henkrar, F., Bouhaddou, N., Khabar, L., 2023. Micropropagation of *Quercus* spp., complications and solutions—an overview. Vitro Cell Dev. Biol. Plant 59 (4), 446–460. https://doi.org/10.1007/s11627-023-10354-4.

Montero, G., Cañellas, I., 1999. El alcornoque (*Quercus suber* L.). In: Manual de reforestación y cultivo. Mundi-Prensa, Madrid.

Morcillo, L., Moutahir, H., Cortina, J., Vilagrosa, A., 2020. The role of population and half-sib family on driving suitable functional traits for *Quercus suber* L. forests restoration. Forests 11 (6), 680. https://doi.org/10.3390/f11060680.

Moreno Moraldi, 2024. Vivaistica forestale - Conoscerla per praticarla con successo. Compagnia delle Foreste, Arezzo, p. 221.

Navarro Cerrillo, R.M., del Campo, A., Alejano, R., Álvarez, L., 1998. Caracterización de calidad final de planta de encina, alcornoque, algarrobo y acebuche en cinco viveros en Andalucía. Informaciones Técnicas 53/98. In: Consejería de Agricultura y Pesca. Junta de Andalucía.

Navarro Cerrillo, R.M., Arribas García, M.A., Gallegos Férula, V., Alcantara Vara, E., 2003. Deficiencias minerales en plantas de una savia de dos especies de frondosas mediterráneas (*Quercus suber* L. *y Ceratonia siliqua*). Investigación Agraria. Sistemas y Recursos Forestales 12 (1), 61–73.

Navarro Cerrillo, R.M., Gallo, L., Sánchez, E., Trapero, A., Fernández, P., 2004. Efecto de distintas fertilizaciones de fósforo en la resistencia de brinzales de encina y alcornoque a *Phytophthora cinnamomi* Rands. Investigación Agraria. Sistemas y Recursos Forestales 13 (3), 550–558.

Navarro Cerrillo, R.M., Sánchez Lancha, A., Arroyo Sauces, M., Plaza, L., Mallofret, E., Lara, M., 2013. In: *Quercus suber* En Pemán, J., Navarro Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales (tomo II). Organismo autónomo Parques Nacionales. Ministerio de Agricultura, Alimentación y Medio Ambiente, Madrid, España, pp. 305–341.

Pardos, M., Cañellas, I., Montero, G., 2002. Seguimiento en campo de un ensayo sobre calidad de planta de alcornoque (*Quercus suber* L.). Cuadernos de la Sociedad Española de Ciencias Forestales 14, 83–88.

Pérez-Devesa, M., Cortina, J., Vilagrosa, A., Vallejo, R., 2008. Shrubland management to promote *Quercus suber* L. establishment. For Ecol. Manage 255 (3–4), 374–382. https://doi.org/10.1016/j.foreco.2007.09.074.

Pérez-Ramos, I., Marañón, T., 2009. Effects of waterlogging on seed germination of three Mediterranean oak species: ecological implications. Acta Oecol. 35 (3), 422–428. https://doi.org/10.1016/j.actao.2009.01.007.

Peñuelas Rubira, J., Benito Matías, L.F., 2018. Son posibles las plantaciones de Quercus mediterŁaneos para la produccion Ł intensiva de bellota y son una oportunidad para el desarrollo rural? Actas del VII Congreso Forestal Español. Plasencia, Spain.

Quilhó, T., Lopes, F., Pereira, H., 2003. The effect of tree shelter on the stem anatomy of cork oak (*Quercus suber*) plants. IAWA J. 24 (4), 385–395. https://doi.org/10.1163/22941932-90000343.

Simões, M.P., Belo, A.F., Fernandes, M., Madeira, M., 2016. Regeneration patterns of *Quercus suber* according to montado management systems. Agrofor. Syst. 90, 107–115. https://doi.org/10.1007/s10457-015-9818-6.

Simonson, W.D., Allen, H.D., Coomes, D.A., 2014. Overstorey and topographic effects on understories: evidence for linkage from cork oak (*Quercus suber*) forests in southern Spain. For. Ecol. Manage 328, 35–44. https://doi.org/10.1016/j.foreco.2014.05.009.

Trubat, R.J., Cortina, A., Vilagrosa, A., 2010. Nursery fertilization affects seedling traits but not field performance in *Quercus suber* L. J. Arid Environ. 74 (4), 491–497. https://doi.org/10.1016/j.jaridenv.2009.10.007.

Villar-Salvador, P., Heredia, N., Millard, P., 2010. Remobilization of acorn nitrogen for seedling growth in holm oak (*Quercus ilex*), cultivated with contrasting nutrient availability. Tree Physiol. 30 (2), 257–263. https://doi.org/10.1093/treephys/tpp115.

Weitkamp, W.H., Tietje, W.D., Vreeland, J.K., 2001. Brush piles and mesh cages protect blue oak seedlings from animals. Calif. Agric. 55 (2). Retrieved from: https://escholarship.org/uc/item/1kr870wc.

Salix L.

María Aránzazu Prada Sáez[1], Jesús Rueda Fernández[2], Fernando Magdaleno Mas[3] and Roberto Martínez Romero[4]

[1]Generalitat Valenciana, VAERSA, Quart de Poblet, Spain; [2]Valladolid, Spain; [3]Ministerio para la Transición Ecológica y el Reto Demográfico, Madrid, Spain; [4]Scottish Environment Protection Agency, Stirling, United Kingdom

Many species of *Salix* (willows) are naturally associated with riverbanks or waterlogged soils. In contrast, others can be found in cool forests, crevices formed in walls, or rocky substrates of high mountain or tundra environments. This file focuses on riparian species, which are particularly relevant to environmental restoration activities.

Propagation

Seed propagation

Salix species bloom between late winter and mid-spring, depending on the species, elevation, and latitude. The fruits develop quickly, within one and 2 months after flowering, so the capsules open between March and June, in the same spring of flowering. To collect seeds, it is essential to wait until the capsules are ripe and begin to open, showing the tufts of long hairs attached to the seeds that facilitate their natural dispersal. Harvesting before dehiscence can seriously compromise seed viability. However, the optimal harvest time must be carefully monitored, as seed dispersal by wind occurs rapidly within a few days.

Harvesting is done manually from the ground by cutting the fruiting catkins or the twigs that carry them with scissors. Seeds that have already come out of the fruit can be collected from the ground if they have fallen recently, provided they have not been moistened by dew or rain or fallen on damp soil. Thus, collected seeds should show tufts fluffy just as they came out of the fruit avoiding those with tufts that are clumping or bunched.

The collected material must be placed in paper bags and kept cool until sowing. If the catkins are not entirely open, they can be left to dry inside the bag at room temperature for about 2 days, after which the capsules will open fully. During the drying process, it is advisable to move the bags frequently so that the drying is homogeneous, and the seeds detach from the capsules.

Removing the seed tufts for storage or sowing is unnecessary. However, tufts can be removed by sieving on a fine mesh screen using compressed air or suction to facilitate seed handling (Dreesen, 2003; Dawes, 2003).

It is important that the cleaning process should not be prolonged, as *Salix* seeds rapidly lose their viability if kept at room temperature for too long (Young and Young, 1992). The seeds have minimal reserve tissues due to their small size (Table 11.63). Karrenberg and Suter (2003) indicated that, under natural conditions, their viability is reduced by 50% after only one to 2 weeks. Similarly, Maroder et al. (2000) observed

Table 11.63 Characteristics of seed lots of European riparian *Salix*.

Purity (%)	No. seeds/kg	Germination capacity (%)
60–70 (65)	1,801,200–50,000,000 (9,836,030)	90–100

Mean values in brackets.
Catalán (1991); Young and Young (2012); Zasada et al. (2008); SER, INSR, RBGK, SID (2024).

a total loss of viability in batches preserved at 25°C for 2 weeks. Therefore, it is recommended to sow as soon as possible after seed collection.

For short-term storage (up to 1 month), we recommend placing the seeds in airtight containers at 2–4°C (Bewyley and Black, 1982). For longer-term preservation, seed moisture can be reduced and seeds stored between −70 and −18°C. Although longevity will increase due to the lower storage temperature, a loss of viability is to be expected within months (Maroder et al., 2000). The seeds may also be cryopreserved in liquid nitrogen without losing viability (Maroder et al., 2000; Wood et al., 2003).

No pretreatment is needed to promote the germination of *Salix* seeds since they have high germination rates (close to 100%), provided they are handled properly. However, late frosts before harvesting can drastically reduce their viability (Young and Clements, 2003). Seeds germinate readily at temperatures typical of springtime. If germination is carried out in a controlled environment, we recommend varying the temperature between 15–20°C and 25°C. The standard conditions for characterizing the germination of *Salix* seed lots involve an alternating temperature cycle of 20–30°C and alternating 16-h light and 8-h dark periods for at least 14 days (ISTA, 2011).

Germination is epigeal, with an initial appearance of tiny hairs on the hypocotyl that attach the seedling to the substrate, followed by radicle development (Zasada et al., 2008). Germination is rapid, occurring in less than 24 h. Table 11.63 summarizes the characteristics of seed lots of European species of the genus. It should be noted that many seeds can be obtained from a single female specimen, so collecting several specimens is recommended to promote genetic diversity.

Salix plants are not usually produced from seeds due to the ease of vegetative propagation. The production of plants from seeds has several disadvantages, including complex handling due to the small size and a less flexible collection period.

Vegetative propagation

Most species of *Salix* can be propagated easily using parts of stems or branches. In fact, vegetative propagation using lignified hardwood cuttings is a common practice in nurseries that produce *Salix* plants for different purposes (ornamental, revegetation of riverbanks, mine restoration, phytoremediation, biomass production, or basketry materials).

Lignified stems or segments are also used in various bioengineering techniques applied to riparian restoration.

The ease of vegetative propagation of many of the species in this genus is due to their ability to develop adventitious roots from stems or branches when placed in a hydrated medium. The application of in vitro techniques only makes sense for species or cultivars that present difficulties for conventional propagation by less expensive methods (Edson et al., 1995; Amo-Marco and Lledo, 1996; Liesebach and Naujoks, 2004; Parzymies et al., 2023).

The optimum size of the plant parts depends on the planting technique. Hardwood cuttings for production in nurseries, either bareroot or container, can vary in length from 5–10 cm mini-cuttings (Dumroese et al., 2003; Landis et al., 2003) to 20–30 cm cuttings. The typical diameter of cuttings for nursery production varies between 0.4 and 2 cm. The cuttings' diameter will depend on the age of the harvested branches and the species. Specifically, shrub *Salix* species tend to produce thinner branches than tree species.

For direct planting of living segments on banks or bioengineering structures, the materials used are typically very different from those recommended for nurseries. For example, for direct planting of cuttings or stakes, segments 30–60 cm long are commonly used but they can reach up to 100 cm in length and have a diameter of 8 cm. Living bundles, for example used in streambank protection, require complete branches of 150–200 cm and diameters of up to 4 cm.

The season, the position within the plant, and age influence the carbohydrate and hormone levels of the branches, which can ultimately affect the rooting ability of the cuttings. Genetic variation in rooting ability among Salicaceae clones has also been reported (McIvor *et al.*, 2014). Generally, *Salix* species root very quickly, allowing stems older than 1 year to be collected. Hoag (2007) indicated that smooth bark material up to 7 years old can be used, although it is also possible to use even older branches or stems. The material should be harvested from vigorous branches, however, that are as young and as close to the base of the plant as possible. Harvesting can be done throughout the winter, during the vegetative dormancy period, although collecting the material at the end of the winter is preferable to reduce storage time.

Harvesting is done by cutting the stems of the plants with pruning shears. If stems for grafting is the goal, branches are cut and lateral branches removed. Removing the secondary branches allows the plant to maintain a proportionate relationship between the shoot and roots. It also facilitates transporting and handling the grafting materials. Hardwood cuttings are obtained by cutting rods into segments of the desired size, keeping their polarity. For this purpose, usually a crosscut is made at the upper end and a bevel cut made at the base. In any case, the cuts must be clean and made with clean tools to prevent contamination by pathogens.

Some growers establish stool beds to ensure an easy and continuous supply of high quality propagation material in quantities needed for large plantings or multiple sites. This approach not only reduces production costs but also enables controlled mixing of clones of hardwood cuttings, promoting genetic diversity and ensuring a balanced ratio of both sexes in the batches produced. It is essential, however, to maintain precise identification of the strains and to avoid excessive propagation from the same genotypes. Periodic renewal of stool beds with new genotypes collected from the field is recommended to maintain genetic vigor.

The collection of plant material in stool beds is carried out as indicated for field collections. Excess branches are removed from the stocks at the end of winter before they sprout again, after obtaining the necessary vegetative material. The yield in the production of reproductive material is very high for *Salix*. Thus, in species such as *Salix atrocinerea*, *S. eleagnos* or *S. purpurea*, it is possible to obtain 3 to 6 stems per stool in the first year and double these Figures in the second year. The stool bed can be maintained for several years, but it is recommended to be renewed every 4–5 years to avoid reducing the quality of the stems obtained from old stools.

The stool beds are also attractive for producing plants from seed, as they are sites where phenological monitoring can be carried out, and seeds of all female clones can be collected easily and cheaply, regardless of phenotypic variation at the time of their dehiscence.

Salix materials for habitat restoration or revegetation are usually obtained from plant parts collected from wild populations, often close to the area to be restored. The strategy of producing materials close to the area of action promotes biodiversity as it does not significantly alter evolutionary processes occurring in local populations, given the relative ease of interbreeding common with *Salix* species. If large quantities are needed of plants from a population close to the site where the intervention is planned, the plants to be used as stools can be partially coppiced. This rejuvenates the plants to obtain more vigorous stems and should be done in late winter of the previous year.

In the case of mass collection of plant parts in natural populations, care should be taken to avoid damaging the source plants. Extract branches from the inner parts of the crowns, leaving more than two-thirds of the branches (Hoag, 2007). When collecting material in the field, it is advisable to aim for a balanced representation of both sexes and diverse donor plants from different parts of the population. Avoid exposing the collected material to the sun and shelter the collection from wind during transportation to prevent it from drying out. Once collected, the material can be stored for 2–4 months in the dark at 2–4°C and 85% relative humidity to avoid dehydration.

The materials should be soaked in water before use for 24–72 h to ensure proper hydration; this enhances rooting and stimulates vigorous growth. According to Schaff et al. (2002), keeping *S. nigra* cuttings in running water for ten days prior to planting (a process

that attempts to mimic the action of the river) increases root biomass and rooting rates under different substrate moisture conditions.

The rooting rate is generally very high, with values between 80% and 100% if the material is collected just before the onset of spring. Lower yields should be expected if the material has been stored. As is usual in many plant species, the genotype of the mother plant and its vigor are key factors for rooting ability.

Seedling cultivation

Container plants

Common practice is to place several seeds per container or cell because seeds are too small to be handled individually. They can be sown directly in containers volumes of 200–300 cm^3. For a two-year-old crop, Dreesen (2003) suggested starting the crop in containers with small cells and increasing the container size as the plant develops. For the first phase, trays with 20 cm^3 cells, typical in horticulture, can be used. The seeds should be uncovered after sowing. These can be kept in the greenhouse during the first weeks without requiring a large space, with adequate irrigation. Keep the substrate constantly moist while preventing waterlogging. In the first week, it is recommended to keep the plants in a greenhouse. Subsequently, when the small seedlings can maintain a consistent root ball, they are transplanted into 150–200 cm^3 containers, where they are grown until late summer of the first year (or spring of the second year if growth is very slow) when they are transplanted into final 3 L containers.

Because germination is not staggered, it is not recommended to sow seeds in trays and transplant seedlings to containers, unless the number of available seeds is limited. Transplanting is time-consuming; because seedlings are delicate at early stages, their roots can break or deform when introduced into the container.

Given the high germination rates, it is normal for several seedlings to germinate per container. Once the survival of the plants in their initial phase is assured, the most vigorous plant should be left in each container, removing any additional ones. If growth starts in small cells, this task can be carried out at the time of transplanting to larger containers.

At present, the most widespread practice for cultivating *Salix* plants in nurseries is the rooting of cuttings in containers. The type of container can vary greatly, depending on the size of the cutting or micro-cutting and the desired plant height. For plants of 30–60 cm, use containers from 0.3 to 1.2 L; use 3 L containers for plants 60–100 cm, and up to 1–1.50 m in height or even more, depending on the species (Fig. 11.46). The cuttings should be hydrated by immersing them in clean water for 24–48 h before inserting them in the containers. In shrub species, the cuttings are inserted into the substrate so that 3 to 4 well-formed buds stick out. For tree species, it is better to leave only one or two well-formed buds exposed.



Figure 11.46 One-year-old seedlings of *Salix purpurea* cultivated in 330, 650, and 1500 mL containers. *(María Aránzazu Prada.)*

Rooting hormones are generally unnecessary because cuttings contain sufficient concentrations of growth promoters. Nevertheless, trials have shown that applying indole-3-butyric acid can promote the formation of longer or more numerous roots (Quiñones Martorello et al., 2019; Ficko and Naeth, 2022; Yoon et al., 2021). This auxin is commonly used to stimulate root production in cuttings of different species (Salaš et al., 2013; Davies et al., 2017). Any commercial product suitable for woody species with indole-3-butyric acid as the active ingredient can be used. The manufacturer's guidelines regarding the most appropriate product concentration and exposure time should be followed.

The substrate used must ensure water retention but, at the same time, provide sufficient aeration and drainage so that the plants do not suffer from prolonged waterlogging. A mixture of peat and perlite in a 1:1 proportion can be used for the rooting phase. For cultivation in a single container, a medium-structured substrate consisting of a mixture of peat and coconut fiber, to which perlite is usually added, is used. In any case, weed and pathogen-free substrates should be used, and commercial mixtures are recommended. There are commercially available mixtures of these components enriched with fertilizers and corrected pH that can be used to cultivate this species.

Although not essential, fertilizers can be applied as soon as leaves appear. Dumroese et al. (2003) consider it suitable to incorporate liquid fertilizer with variable concentrations of NPK and other nutrients, adjusted to the requirements of the plants in the different stages of development. Dreesen (2003) recommended the following protocols: (i) for growing cuttings, incorporate a slow-release fertilizer of 15-9-11 NPK with a life of 5–6 months at doses of 3.5 kg/m^3 of substrate for smaller containers 15 g on the substrate in 3L containers; (ii) for growing from seed, initially apply a solution of 200 mg/L of an NPK 20-10-20 complex fertilizer once or twice a week. At the time of each transplanting apply a slow-release fertilizer with a life of 5–6 weeks according to the same protocol given above.

Weeds should be controlled, regardless of the type of production, particularly at the beginning of cultivation. Check frequently for pests and diseases, applying appropriate chemicals if necessary.

Bareroot plants

Cultivating bareroot plants from cuttings allows for mass production at a significantly lower cost than cultivation in containers and with a much lower environmental impact, as no containers or substrates are used. It should be noted, however, that the period for outplanting bareroot plants in the field is shorter than for container plants. Compared with direct planting of field-collected materials, planting bareroot seedlings has the advantage of possibly higher yields of local plants while avoiding reducing or depleting natural populations.

The most widespread practice is to use cuttings, although similar to producing plants in containers, bareroot plants can be obtained in seed beds. The following information focuses on cultivating cuttings. The soil for cultivating bareroot cuttings should be intensively tilled, for example by subsoiling. It is advisable to carry out tillage a few months before planting, during the autumn or at the beginning of the previous winter, allowing the soil to rest until the end of the winter. An organic fertilizer can be added and integrated into the soil during tillage. It is advisable to lightly till the soil again just before planting the cuttings to homogenize it much as possible.

Once the soil has been prepared, insert the cuttings into the soil in rows with at least one bud emerges above the soil surface. As *Salix* internodes are short, it is normal for 3 to 4 buds to appear. The cuttings can be pretreated with a rooting product. Competition between plants decreases the lateral development of the aerial part, facilitating handling during cultivation, transportation, and planting. The distance between rows will be defined by the width of the available mechanical equipment. The distance between plants in the same row can be 20–30 cm, but no more (Fig. 11.47).

Irrigation should be planned in advance, including blanket, sprinkler, or drip irrigation. Drip irrigation is recommended (Fig. 11.47), at least in the Mediterranean region,



Figure 11.47 One-month-old bareroot *Salix* plants from cuttings. Note the drip irrigation line. *(María Aránzazu Prada.)*

as it saves water. If drip irrigation is used, the irrigation lines should be installed before planting the cuttings. The distance between irrigation nozzles will depend on the distance between cuttings in the same row (or vice versa). Drip irrigation requires a weekly check, as nozzles often become clogged.

The cuttings should be watered immediately after being planted to settle the soil and ensure that the cuttings come into contact with the soil and there are no air pockets. During the vegetative period, plants should be watered regularly at varying rates and frequency depending on weather conditions and plant demand. In any case, *Salix* species require permanent moisture in the soil profile without prolonged waterlogging.

Salix plants are not very demanding of nutrients. If the soil is impoverished, however, such as by continuous cultivation, or if plants do not grow vigorously, a mineral or organic fertilizer should be added. A soil analysis is advisable to detect nutrient deficiencies or conductivity problems. As a reference, consider the requirements of another plant of the Salicaceae family; *Populus* ×*canadensis* clone 'I-214' on a 2-year production cycle requires 340 kg/ha of nitrogen, 100 kg/ha of phosphoric anhydride (P_2O_5), 300 kg/ha of potassium oxide (K_2O), and 490 kg/ha of calcium oxide (CaO) (Rueda et al., 2020).

Due to their prolific budding, bareroot plants develop a well-branched aerial system. Pruning is not necessary during cultivation if the plants are intended for revegetation of riverbanks. Plants can be intensively pruned, however, before removing them to facilitate transportation or to remove the less vigorous branches from the stock to reduce the leaf area, especially for 2-year-old plants. Competing vegetation should be controlled. Weeds can be removed between rows by harrowing with machinery, while those around plants in the same row can be removed by hand with a hoe. This also reduces soil compaction that is caused mainly by irrigation water.

Table 11.64 Morphological attributes of 1-year-old seedlings of *Salix* spp. (observed values).

Attributes	Container seedlings	Bareroot seedlings (cm)
Shoot height (cm)[a]	30–180 cm (from seeds) 50–180 cm (from cuttings)	Up to 200

[a]Shoot height varies depending on the species growth habit and/or container size.
Dreesen (2003); Morgenson (1991); unpublished data from the authors.

It is recommended that bareroot *Salix* plants leave the nursery after 1 year, although 2-year-old plants are also used. The rapid growth of *Salix* makes it inadvisable to grow bareroot material any longer, as large plants are more expensive to manage and they have lower survival in the field. As with other crops, when planning to produce bareroot *Salix* plants over multiple years, it is advisable to divide the land into sectors and allow each sector to lie fallow for 1 year on a rotational basis.

The plants are extracted with a tractor-attached blade tool designed explicitly for lifting bareroot woody crops. As the machine pulls up the plants, other operators must lift them and place them on the transport vehicles. Plants should be lifted on the same day they are transported to the final planting site for immediate planting. Plants should be covered with a tarpaulin during transport to prevent drying out. If planting cannot be done on arrival at the site, the plants should be kept in a cool place with the roots covered with moist soil or sand. Table 11.64 summarizes the size 1-year-old seedlings of *Salix* species.

Field establishment

Plants of the genus *Salix* are used for multiple purposes in mild climate areas. Beyond the traditional use of certain species in basketry, due to their rapid growth and ability to assimilate heavy metals, they are used to produce roundwood, biomass, or for phytoremediation of contaminated soils (Zalesny et al., 2016). Techniques for the establishment of energy crops at high densities using selected cultivars (Kuzovkina, 2008) are collected in numerous fact sheets available online (e.g., Barry Caslin et al., 2015; PSU, 2014; Kwapong, 2023). For the production of sawn wood, veneer, or the chipping industry, arboreal cultivars of the *Salix* genus selected for growth, stem straightness, and technologically favorable wood characteristics, among other traits, are used (Cerrillo et al., 2015). In this case, planting and silvicultural techniques similar to those applied to species of the genus *Populus* are employed (Stanturf and van Oosten, 2014). The ability of *Salix* species to thrive on temporarily waterlogged land allows this type of cultivation on sites unsuitable for poplars.

The use of *Salix* in different bioengineering techniques is described in Zeh (2007) and Adam et al. (2008). The most widespread use in Europe is for riverbank restoration or revegetation for erosion control and stabilization and to increase the biodiversity of

these habitats. The following recommendations focus on the field establishment of *Salix* plants, both bareroot and container stock types, as part of riparian restorations.

Riparian restoration usually requires clearing and site preparation, which can be done mechanically. In many riverbeds in the Mediterranean region, it is necessary to previously eliminate reed beds of *Arundo donax*, which occupies the riverbanks with very aggressive, invasive growth. These reed beds are removed with machinery and the land is reprofiled. A geotextile is installed and maintained for a couple of years to prevent the reed from growing back from remining rhizomes and to reduce erosion until vegetation has reestablished. Subsequently, the *Salix* is planted.

The most suitable planting time is between autumn and spring, outside the vegetative period, and when the soil is moist. For bareroot or container plants, holes of approximately 40 × 40 × 40 cm are dug, usually by hand. The proximity of *Salix* plantations to the watercourse usually makes heavy machinery inadvisable since it can destabilize the banks, damage the bed and remnant vegetation, among other effects. Lighter machinery is preferable that has a long arm that allows the holes to be made some distance away from the riverbank. It is advisable to use excavators with small volume buckets to dig holes in the sites closest to the river flow to facilitate planting without excessive earth movements that could threaten bank stability.

Salix species are usually planted in the areas of most significant contact between riparian vegetation and water. Shrub species are used in areas where the riparian forest is closer to the watercourse, while arboreal species are planted further away from the bank. In addition, depressions in the ground away from the bank are more suitable for shrub than arboreal species of the genus *Salix*.

The high capacity of *Salix* to reproduce vegetatively provides opportunities to economize efforts and to naturalize the distribution of the species along the river section. Plants can be distributed irregularly to give a more natural appearance from the beginning. For example, groups of 15–50 plants can be made, leaving unplanted spaces between groups that will be colonized later as the plantings develop. Magdaleno Mas and Goikoetxea Barrios (2021) designed low-density copses of 25–50 plants each, within a 4 × 4 m reference frame, irregularly distributed at an average of 5 copses/ha, to favor natural regeneration. This layout provided an open but more or less continuous land occupation, with densities of around 125 plants/ha. Different planting densities are advisable, depending on the distance to the watercourse and the water level since plantings closer to the waterway will be affected by early floods. A planting density of 800–1200 plants/ha is recommended in the area closest to the watercourse, with a reduction to 500–800 plants/ha farther away. At more distant locations, specimens of arboreal *Salix* species are planted, along with other riparian species, at densities of 300–500 plants/ha.

The closer to the watercourse, the greater the risk of the plant being affected by an early flood. The acceptable mortality in Spain ranges from <13% for plantings with

more than 1000 plants/ha to <7% for lower density plantations (adapted from Serrada, 2000). The assessment of the percentage of dead plants should be carried out in the month of July after planting. Dead plants should be replaced the year after planting if needed based on the condition of the population.

These copses include groups of several species of the genus, selected according to the composition of the natural formations of the area to be recovered. Within these copses, some other species of the usual riparian plant species can be included according to the specific conditions of the section of the river being revegetated. The condition and evolution of the banks must also be considered. In river sections where erosion occurs (e.g., outer bank of meanders), it is advisable to cover the ground with vegetation as soon as possible without leaving clearings between copses. Where sedimentation occurs (e.g., inner bank of meanders), different planting designs can be chosen when the soil has been deposited. In the case of recent coarse sediments, it will be necessary to wait for floodwaters to deposit fine elements.

An efficient approach is to mark beforehand the intended locations of the copses and the spots where the material will be planted. Any of the usual field markers can be used, the simplest being biodegradable dyes for all types of surfaces. Protective tubes are not recommended; in the case of the presence of livestock, it is preferable to fence the planting area. Individual protection with anti-herbivore netting could be used for larger specimens planted far from the river course. Perennial ground cover may quickly compete with the planted *Salix*. If necessary to avoid failure of the planting, hoe an area of 1 m^2 around each plant. Mechanized weeding may be used in specific circumstances where the new planting is shaded by a shrub layer. Selective herbicides are strongly discouraged due to their adverse environmental consequences.

Irrigation needs to be applied at the time of planting. The suitability of subsequent maintenance irrigation will depend on the proximity of the planting to the riverbed, the water flow during the summer season, and annual climate. If watering is necessary, bareroot plants will require more water than those grown in containers. In rivers with a natural hydroperiod, it may be convenient to water the plants at least three times in each of the two summer periods after planting to shorten the duration of the summer drought. However, this will be unnecessary in regulated rivers without a low water flow period.

Chapter References

Adam, P., Debais, N., Gerber, F., Lachat, B., 2008. Le génie végétal. Un manual technique au service de l'aménagement et de la restauration des milieux aquatiques. In: La Documentation Française, Paris.

Amo-Marco, J.B., Lledo, M.D., 1996. In vitro propagation of *Salix tarraconensis* Pau ex Font Quer, an endemic and threatened plant. In Vitro Cellul. Dev. Biol. Plant 32, 42–46. https://doi.org/10.1007/BF02823012.

Bewyley, J.D., Black, M., 1982. Physiology and biochemistry of seeds in relation to germination. In: Viability, Dormancy, and Environmental Conditions. Springer-Verlag, Berlin Heidelberg New York, vol 2.

Barry Caslin, B., Finnan, J., Johnston, C., McCracken, A., Walsh, L., 2015. Short rotation coppice willow. In: Best Practice Guidelines. Teagasc, Crop Research Centre, Oak Park, Carlow. AFBI, 2AFBI, Agri-Food and Bioscience Institute, Newforge Lane, Belfast.

Catalán, G., 1991. Semillas de árboles y arbustos forestales. In: Instituto Nacional para la Conservación de la Naturaleza. Ministerio de Agricultura, Pesca y Alimentación, Madrid.

Cerrillo, T., Álvarez, J.A., Álvarez, J.L., Battistella, A., et al., 2015. La forestación de salicáceas como aporte al desarrollo sustentable del Delta del Paraná. In: XXIX Jornadas Forestales de Entre Ríos, 24-25 de septiembre de 2015, Concordia. INTA Concordia e Asociación de Ingenieros Agrónomos del nordeste de Entre Ríos, Concordia.

Davies, F.T., Geneve, R.L., Wilson, S.B., 2017. Hartmann and Kester's Plant Propagation Principles and Practices, ninth ed. Pearson Education Inc, New York.

Dawes, D., 2003. Using a shop vacuum to clean Salicaceae seeds. Nativ. Plants J. 4, 140. https://doi.org/10.3368/npj.4.2.140.

Dreesen, D., 2003. Propagation protocol for container willows in the Southwestern US using seeds. Nativ. Plants J. 4, 118–124. https://doi.org/10.3368/npj.4.2.118.

Dumroese, R.K., Wenny, D.L., Morrison, S.J., 2003. Propagation protocol for containers Willows and Poplars using mini-cuttings. Nativ. Plants J. 4, 137–139. https://doi.org/10.3368/npj.4.2.137.

Edson, J.L., Leege-Brusven, A.D., Wenny, D.L., 1995. Improved vegetative propagation of Scouler willow. Tree Planters' Notes (U.S. Dep. Agric. For. Serv.) 46, 58–63.

Ficko, S.A., Naeth, A., 2022. Influence of treatment on rooting of arctic *Salix* species cuttings for revegetation. Arct. Antarct. Alpine Res. 54 (1), 62–77. https://doi.org/10.1080/15230430.2022.2042946.

Hoag, J.C., 2007. How to Plant Willows and Cottonwoods for Riparian Restoration. US Department of Agriculture Natural Resources Conservation Service, Boise, Idaho. Technical Note Plant Material no. 23.

ISTA (International Seed Testing Association), 2011. International Rules for Seed Testing. ISTA, Bassersdorf, Switzerland.

Karrenberg, S., Suter, M., 2003. Phenotypic trade-offs in the sexual reproduction of Salicaceae from flood plains. Am. J. Bot. 90, 749–754. https://doi.org/10.3732/ajb.90.5.749.

Kuzovkina, J., 2008. The registration of *Salix* cultivars. Acta Hortic. 799, 141–141.

Kwapong, N.A., 2023. Willow SRC (*Salix* spp.). Biomass Connect. https://www.biomassconnect.org/technical-articles/willow-crop-information-factsheet.

Landis, T.D., Dreesen, D.R., Katen, R.K., 2003. Sex and the single *Salix*: consideration for riparian restoration. Nativ. Plants J. 4, 110–117. https://doi.org/10.3368/npj.4.2.110.

Liesebach, M., Naujoks, G., 2004. Approaches on vegetative propagation of difficult-to-root *Salix caprea*. Plant Cell Tissue Organ Cult. 79, 239–247. https://doi.org/10.1007/s11240-004-0665-9.

Magdaleno Mas, F., Goikoetxea Barrios, M., 2021. Restauración de bosques de ribera: el caso de los ríos Arga y Aragón (Navarra). In: Pemán García, J., Navarro Cerrillo, R.M., Prada Sáez, M.A., Serrada Hierro, S. (Eds.), Bases técnicas y ecológicas del proyecto de repoblación forestal. Tomo II. Ministerio para la Transición Ecológica y el Reto Demográfico Madrid.

Maroder, H.L., Prego, I.A., Facciuto, G.R., Maldonado, S.B., 2000. Storage behaviour of *Salix alba* and *Salix matsudana* seeds. Ann. Bot. 86 (5), 1017–1021. https://doi.org/10.1006/anbo.2000.1265.

McIvor, I.R., Sloan, S., Pigem, L.R., 2014. Genetic and environmental influences on root development in cuttings of selected Salix and Populus clones – a greenhouse experiment. Plant Soil 377, 25–42. https://doi.org/10.1007/s11104-013-1770-5.

Morgenson, G., 1991. Vegetative propagation of poplar and willow. In: The Intermountain Forest Nursery Association Annual Meeting, August 12-16, Park City, Utah.

Parzymies, M., Pogorzelec, M., Głębocka, K., Sliwinska, E., 2023. Micropropagation protocol and genetic stability of the *Salix myrtilloides* plants cultivated in vitro. Biology 12, 168. https://doi.org/10.3390/biology12020168.

PSU, 2014. Renewable and Alternative Energy Fact Sheet. The Pennsylvania State University, PennState Extension, State College, Pennsylvania.

Quiñones Martorello, A., Fernández, M.E., Monterubbianesi, M.G., Colabelli, M.N., Laclau, P., Gyenge, J.E., 2019. Effect of combined stress (salinity + hypoxia) and auxin rooting hormone addition

on morphology and growth traits in six *Salix* spp. clones. New For. 51, 61–80. https://doi.org/10.1007/s11056-019-09719-8.

Rueda, J., García Caballero, J.L., Martínez Sierra, F., Prada, M.A., 2020. Viveros de chopos en Castilla y León. Junta de Castilla y León, Consejería de Fomento y Medio Ambiente, Valladolid.

Salaš, P., Sasková, H., Mokričková, J., Litschmann, T., 2013. Evaluation of different types of rooting stimulators. Acta Univ. Agric. Silvic. Mendel Brun. 60, 217–228.

SER, INSR, RBGK, SID, 2023. Seed Information Database (SID). Available from: https://ser-sid.org/.

Schaff, S.D., Pezeshki, S.R., Shields Jr., F.R., 2002. Effects of pre-planting soaking on growth and survival on black willow cuttings. Restor. Ecol. 10, 267–274. https://doi.org/10.1046/j.1526-100X.2002.02035.x.

Serrada, R., 2000. Apuntes de repoblaciones forestales. 3ª edición. In: Escuela Universitaria de Ingeniería Técnica Forestal. Universidad Politécnica de Madrid, Madrid.

Stanturf, J.A., van Oosten, C., 2014. Operational poplar and willow culture. In: Isebrands, J., Richardson, J. (Eds.), Poplars and Willows: Trees for Society and the Environment. CABI, pp. 200–257.

Wood, C.B., Pritchard, H.W., Lindgaard, K., 2003. Seed cryopreservation and longevity of two *Salix* hybrids. Cryo-Letters 24, 17–26.

Yoon, A., Oh, H.E., Kim, S.Y., Park, Y.G., 2021. Plant growth regulators and rooting substrates affect growth and development of *Salix koriyanagi* cuttings. Rhizosphere 20, 100437. https://doi.org/10.1016/j.rhisph.2021.100437.

Young, J.A., Clements, C.D., 2003. Seed germination of willow species from a desert riparian ecosystem. J. Range Manage. 56, 496–500. https://doi.org/10.2307/4003842.

Young, J.A., Young, C.G., 1992. Seeds of Woody Plants in North America. Discorides Press, Portland, Oregon.

Zalesny, R.S., Stanturf, J.A., Gardiner, E.S., Bañuelos, G.S., Hallett, R.A., Hass, A., Stange, C.M., Perdue, J.H., Young, T.M., Coyle, D.R., Headlee, W.L., 2016. Environmental technologies of woody crop production systems. BioEnergy Res. 9, 492–506. https://doi.org/10.1007/s12155-016-9738-y.

Zasada, J.C., Douglas, D.A., Buechler, W.M., 2008. *Salix* spp. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual, Agriculture Handbook 727. US Department of Agriculture, Forest Service, Washington DC, pp. 1000–1009.

Zeh, H., 2007. Ingeniería Biológica. Manual Técnico. Federación Europea de Ingeniería del Paisaje - Verein für Ingenieurbiologie. In: Vdf Hochschurverlag AG an der ETH Zürich, Zürich.

Sorbus domestica L.

Petar Zhelev[1], Ina Aneva[2] and Nasko Iliev[1]
[1]University of Forestry, Sofia, Bulgaria; [2]Department of Plant and Fungal Diversity and Resources, Institute of Biodiversity and Ecosystem Research, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

Sorbus domestica (service tree) fruits typically ripen and are harvested in October–November. Before seed extraction, fruits can be stored for a short time in ventilated areas at temperatures below 10°C to prevent fermentation and mold formation. Seeds are extracted manually or mechanically squashing the fruits, followed by flotation, flushing to remove pulp and debris, and sifting (Nikolova et al., 2021; Moraldi, 2024). Larger fruits are preferred (Drvodelić et al., 2018) because they produce more seeds with higher germination rates, although processing large and small fruits is labor-intensive. Seeds can be sown right after extraction or undergo short-term storage in moist sand until next spring. For long-term storage, seed moisture content must be reduced to 6%–8%, and seeds stored at 0–5°C for up to 8 years (Nikolova et al., 2021).

Seeds show deep dormancy, which can be broken through cold, wet stratification. One option is storing seeds in wet sand at 0–3°C for 120 days (Nikolova et al., 2021), but stratification under fluctuating temperatures (−5°C to 5°C) releases dormancy more effectively than a constant 4°C presowing treatment (Paganová, 2007). The quantitative characteristics of Sorbus domestica seeds are shown in Table 11.65.

Vegetative propagation

Vegetative propagation by rooting stem cuttings is very difficult and therefore is not applied. Some studies have reported successful rooting of stem cuttings taken from seedlings or coppice shoots and rooted in a mixture of peat and sand 1:1, or peat and perlite 1:1 (Čížková, 1997; Benedíková, 2009). Grafting is used to increase flowering and fruiting. While micropropagation is feasible and effective (Arrilaga et al., 1991; Nikolaou et al., 2008) its economic viability is unclear.

Table 11.65 Characteristics of seed lots of *Sorbus domestica*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
0.5–2.3 (0.75)	60–89 (77)	25,000–38,000 (32,000)	50–98 (57)

Mean values in brackets.
Piotto and Di Noi (2001); Prknová (2015); Drvodelić et al. (2018); Nikolova et al. (2021); Moraldi (2024).

Table 11.66 Morphological attributes of 1 + 0 container and bareroot seedlings of *Sorbus domestica* as well as recommended values for quality plants.

	Container seedlings		Bareroot seedlings	
Attributes	**Observed values**	**Recommended values**	**Observed values**	**Recommended values**
Shoot height (cm)	30–100	>30	20–90	>30
Root collar diameter (cm)			0.3–0.9	>0.5

Milev et al. (2004); Špíšek and Benediková (2015); Davidović and Jemrić (2020).

Seedling cultivation

Container plants

Container plants are produced mostly for ornamental purposes but can be feasible also for forestry use (Davidović and Jemrić, 2020). The seeds can be sown directly in containers, minimum 300 mL in volume (von Schmeling, 2000; Davidovic and Jemrić, 2020), with wide spacing (at least 30 cm) among plants for optimal foliage development. Morphological characteristics of container S. domestica 1+0 seedlings are presented in Table 11.66.

Bareroot plants

Seeds are sown in nursery seedbeds during autumn, immediately after extraction, or in spring following stratification, preferably in rows at a depth of 2 cm, with a density of 40 seeds/m (Milev et al., 2004). The rows should be spaced 20 cm apart. On average, 30 to 35 seedlings can be cultivated per meter in the first year, with approximately 25 surviving into the second year. By the end of the first year, seedlings typically reach a height of 10–20 cm; however, growth can be enhanced through standard NPK fertilization, allowing most plants to attain the minimum required height of 30 cm for planting. Seedlings may be transplanted to another bed for an additional year of cultivation to achieve the desired size. Morphological characteristics of bareroot S. *domestica* 1+0 seedlings are presented in Table 11.66.

Field establishment

Sorbus domestica can grow in both full sun and semishade. It is relatively tolerant of variations in soil pH and can grow in slightly basic soils. The recommended planting density is 2–3 m × 1–1.5 m or larger to ensure adequate light access to all parts of the crown. This species is vulnerable to late frosts. Planting is carried out in spring or autumn, with the latter being preferable on drier sites. Soil preparation, whether mechanical or manual depending on the terrain, is advisable; however, if infeasible,

seedlings can be planted in holes made using mechanical augers. On drier sites, localized irrigation is recommended during prolonged dry summer periods, with 5 L per plant applied 3–5 times.

The age of seedlings used depends on their size – 1 + 0 seedlings may be planted if they meet the required height of >20 cm; otherwise, 2 + 0 seedlings should be used. Some damage from animal browsing may occur, necessitating protective measures such as fencing or tree guards in areas with high deer populations. In addition, the control and manual removal of competing herbaceous vegetation may be required during the first year of cultivation.

Chapter References

Arrilaga, I., Marzo, T., Segura, J., 1991. Micropropagation of juvenile and adult *Sorbus domestica* L. Plant Cell Tissue Organ Cult. 27, 341–348.

Benedíková, M., 2009. Methods for regeneration and breeding of *Sorbus domestica* L. Lesnický průvodce (Trnava) 3, 3–18 (in Czech with English summary).

Čížková, L., 1997. Testing of Vegetative Propagation of Broadleaved Species. Report of a Project Conservation of the Genepool of Selected Broadleaved Tres in Moravia. Forest Research Institute of Moravia (in Czech).

Davidović, D., Jemrić, T., 2020. Dynamics of vegetative growth of seedlings of Service tree (*Sorbus domestica* L.) during the first three years after sowing. Zbornik Akademije Poljoprivrednih Znanosti 2, 37–44 (in Croatian with English summary).

Drvodelić, D., Oršanić, M., Vuković, M., Jatoi, M.A., Jemrić, T., 2018. Correlation of fruit size with morpho-physiological properties and germination rate of the seeds of Service Tree (*Sorbus domestica* L.). SEEFOR 9 (1), 47–54. https://doi.org/10.15177/seefor.18-01.

Milev, M., Alexandrov, P., Petkova, K., Iliev, N., 2004. Sowing Materials of Broadleaved Species. Videnov & Son Publ. House, Sofia. Bulgaria (in Bulgarian).

Moraldi, M., 2024. Vivaistica forestale - Conoscerla per praticarla con successo. Compagnia delle Foreste, Arezzo, 221pp.

Nikolaou, P., Zagas, D., Scaltsoyiannes, V., Barbas, E., Xilogianni, V., Tsoulpha, P., Tsaktsira, M., Voulgaridou, E., Iliev, I., Triandafilou, K., Scaltsoyiannes, A., 2008. Advances in the micropropagation of Service tree (*Sorbus domestica* L.). Propag. Ornam. Plants 8, 154–157.

Nikolova, A., Tsvetanova, Z., Koyuva, L., 2021. Guidebook LIFEFORHAB. Forest Seed Control Station, Sofia (in Bulgarian). https://www.lifeforhab.eu/wp-content/uploads/2021/03/Guidebook-LIFEFORHAB-text.pdf.

Paganová, V., 2007. Generative reproduction of *Sorbus domestica* L. as a limiting factor of its wider utilization in conditions of Slovakia. Propag. Ornam. Plants 7 (4), 199–203.

Piotto, B., Di Noi, A. (Eds.), 2001. Propagazione per seme di alberi e arbusti della flora Mediterranea. Manuale ANPA, Roma, p. 212.

Prknová, H., 2015. Long-term storage of service tree (*Sorbus domestica* L.) seeds and induction of their germination. J. For. Sci. 61 (10), 417–421. https://doi.org/10.17221/57/2015-JFS.

Špíšek, Benedíková, M., 2015. Distribution and reproduction method of service tree. In: Paganová, V. (Ed.), Proceeding of Scientific Conference *Sorbus domestica* L. In Urban Context and in Landscape, pp. 22–25.

Von Schmeling, W.K.-B., 2000. Der Speierling. Druckhaus Göttingen, 184 pp.

Tamarix L.

María Aránzazu Prada Sáez[1], Jesús Rueda Fernández[2], Fernando Magdaleno Mas[3] and Roberto Martínez Romero[4]

[1]Generalitat Valenciana, VAERSA, Quart de Poblet, Spain; [2]Valladolid, Spain; [3]Ministerio para la Transición Ecológica y el Reto Demográfico, Madrid, Spain; [4]Scottish Environment Protection Agency, Stirling, United Kingdom

Propagation

Seed propagation

Species of *Tamarix* can be easily propagated from seed. The time of collection is usually variable, considering that many species may flower in winter/spring (between March and May) and in summer or autumn (Baum, 1978). The appearance of inflorescences may vary according to the climatic conditions at the site and the year. However, this characteristic is more stable in certain species (e.g., *T. boveana* flowers in spring).

Dehiscence takes place in spring, mostly in April, or autumn, depending on the flowering season, coinciding with the two rainy seasons typical of the Mediterranean climate. The fruiting catkins are collected when the capsules begin to open. Seeds disperse quickly thanks to the tuft of hairs they bear, which facilitates dispersal by the wind. Therefore, collection should not be delayed once the seed hairs emerge from the fruits. The infructescences are collected manually by cutting the twigs that carry them with pruning shears and subsequently deposited in trays or paper bags to allow the fruits to open completely. Careful harvesting is recommended, avoiding including pieces of twigs and leaves in the batch that would interfere with cleaning the small hairy seeds. Shepperd (2008) believes that removing the capsules is not worthwhile because of the labor involved in this task.

Collected seeds rapidly lose viability at room temperature (Shepperd, 2008) and dehydrated seeds can be stored in a chamber at 4°C for several years. indicate that, under these storage conditions, however, a 50% loss of viability can be expected (Reynolds and Alexander, 1974). On the contrary, Li et al. (2023) observed no loss of viability in batches preserved at low moisture content for 1 year at 4°C and at room temperature. The key to maintaining viability in the short-term, therefore, is to reduce their moisture content and keep seeds in airtight containers. Seeds with reduced moisture content (at equilibrium with 15% relative humidity) can be kept at −20°C without losing viability for at least 7 months (SER, INSR, RBGK, SID, 2023).

The potential seed production of one individual *Tamarix* tree is very high; a mature specimen can produce an estimated 0.5-1 million seeds per year (Tomanek and Zeigler, 1962). Thus, to produce batches of plants with some genetic diversity, it is recommended that seeds be collected from several plants.

Sowing should be done immediately after harvesting when the germination capacity is highest. Pregermination treatments are unnecessary as the seeds do not show dormancy. Germination takes place within 24 h (Di Tomaso, 1998). According to

Table 11.67 Characteristics of seed lots of European *Tamarix*.

No. seeds/kg	Germination capacity (%)
1,000,000–100,000,000 (4,431,000)	50–100 (75)

Mean values in brackets.
Piotto and Di Noi (2004); SER, INSR, RBGK, SID (2023).

Young et al. (2004), with sufficient moisture in the substrate, the optimum conditions for germination of *T. ramossisima* seeds are at temperatures between 15 and 25(30)°C, with a daily oscillation of ±5–10°C (or no oscillations, according to Li et al. (2021). For this species, Li et al. (2021) noted that seed batches collected at the end of the summer showed more significant variation in germination capacity and that optimal temperatures were higher than those required for batches collected in the spring (35–40°C). Table 11.67 summarizes the characteristics of seed lots of European *Tamarix* species.

Vegetative propagation

Tamarix species can be easily propagated using hardwood cuttings from branches, typically from cuttings harvested in winter. Cuttings harvested in the summer can also be used, but these need to be kept under controlled conditions during the rooting stage, with adequate watering to prevent them from drying out.

Young branches with a diameter of 0.5–1 cm are cut with pruning shears to obtain cuttings. The branches are cut into segments 10–30 cm long, depending on the type of cultivation to be carried out (container cultivation of mini-cuttings, hardwood cuttings, or bareroot cultivation). If cuttings are to be planted directly in the final location, longer stem segments (or complete stems) of three or more years, greater thickness (2.5–3 cm minimum diameter) can be used. The polarity of the branches should be maintained by making a beveled cut at the cutting base, indicating the end to be planted, which also increases the rooting surface.

Cuttings should be placed in a growing medium or outplanted at the final site immediately after cutting. Materials harvested early in winter can be stored at 2–4°C and 85% relative humidity to prevent dehydration until planting in early spring. If the storage time is brief, cuttings can be buried in moist sand or soil and shaded. If storage lasts only a few days, they are immersed in clean water.

Tamarix materials used in riparian restoration projects are usually collected from wild populations. Gathering the materials from specimens near the restoration site (with the corresponding permits) is advisable. This ensures the use of species and material native to the area, given the difficulty non-experts have with taxonomic identification of the genus. It also promotes the conservation of the genetic resources of local populations and avoids introducing uncontrolled modifications to their gene pool. Some professional

plant operators install stool beds for an accessible and continuous supply of quality materials in the quantities needed for large or multiple sites. In this case, to maintain genetic diversity and ease of taxonomic identification, it is advisable to establish stool beds with materials collected from local populations. This keeps the strains perfectly identified and avoids very long multiplication cycles that lead to overuse of only a few clones. For this purpose, renewing the strains will be necessary from time to time with new genotypes collected in the field. Under no circumstances should plants of unknown origin be used such as those marketed for ornamental purposes.

Seedling cultivation

Container plants

Tamarix species can be readily propagated from seed, although the small seeds are difficult to handle. It is recommended to start cultivation by sowing seeds in small containers of about 20 cm^3, with subsequent transplanting to larger containers. *Tamarix* seedlings grow slowly and require moist conditions to thrive, beginning with saturated substrates during the first 2–4 weeks of growth (Horton et al., 1960). Because of their slow growth, *Tamarix* plants obtained from seeds are usually planted after 2 years in the nursery since one-year-old plants are small and their roots cannot maintain root ball consistency (Viveros Fuenteamarga, Valladolid, Spain, personal communication).

Mass cultivation of plants of this genus is generally done from lignified cuttings obtained from branches harvested in the field during the winter. Depending on the container size, cuttings are usually 10–30 cm long. Once immersed in water for 24–78 h to hydrate them, cuttings are put directly into 300–1200 cm^3 cells to obtain plants 30–60 cm tall or in containers of up to 3 L if larger plants grown over longer period are desired (Fig. 11.48). A mixture of coconut fiber and peat with added perlite to increase porosity can be used as a substrate, as the plants need humidity and good drainage.

Although applying rooting hormones is not necessary, due to genotypic variation in rooting capacity it is recommended to use commercially available woody crop rooting stimulants and apply them according to the manufacturers' instructions. indicate that the Survival, rooting ability, and subsequent development of plants is appreciably improved if growth regulators are applied to cuttings in the following proportions: 100 mg/L gibberellin A3, 1000 mg/L indole-3-butyric acid, 250 mg/L naphthaleneacetic acid, and 500 mg/L of 6-benzylamine purine (Chen et al., 2023). *Tamarix* plants are undemanding regarding nutrients, so fertilizers are not necessary or small amounts of fertilizer could be supplied if cuttings are used for plant production.

Bareroot plants

Tamarix species can be produced as bareroot stock from seed, reaching 40–90 cm in height over long growing cycles (Navarro-Cerrillo and Gálvez, 2001). The usual practice, however, is the cultivation of lignified, 25–30 cm long cuttings inserted in



Figure 11.48 One-year-old *Tamarix* plants cultivated in a container nursery from cuttings. *(María Aránzazu Prada.)*

previously prepared soil. Soil preparation consists of intense and deep tillage by plowing to eliminate competing vegetation and break up and aerate the soil. In addition to soil preparation, it is advisable to apply an organic fertilizer. Just before planting the cuttings, a light tillage is recommended to achieve the finest possible structure and favor soil contact.

The irrigation system should be designed and installed before planting. Drip irrigation is recommended. If this system is used, the cuttings are placed in the soil in rows, following the drip tubes (Fig. 11.49). The distance between plants can be 20–30 cm. The distance between rows is determined by the width of the machinery used for maintenance work. The cuttings are inserted into previously moistened (but not waterlogged) soil, allowing a segment with at least one bud to emerge. Subsequently, the cuttings should be watered to eliminate air pockets and allow them to come into contact with the soil. During the vegetative period, plants should be watered regularly at varying rates and frequency depending on the weather conditions at the growing site. It is essential to control competing vegetation, which must be eliminated by hand around the plants and by machine harrowing between planting rows.



Figure 11.49 Cultivation of bareroot plants of *Tamarix* using cuttings and irrigated by a drip system. *(María Aránzazu Prada.)*

Bareroot *Tamarix* plants can leave the nursery after 1 or 2 years. They are extracted with a tractor-attached blade tool designed specifically for lifting bareroot woody crops. As the plants are uprooted, other operators must lift them and place them in the transport vehicle. Plants should be lifted on the same day they are transported to the final planting site and planted immediately afterward. Plants should be covered with a tarpaulin during transportation to prevent drying out. If planting cannot be done on arrival at the final site, the plants should be kept in a cool place with the roots covered with moist soil or sand. Table 11.68 summarizes the size 1-year-old seedlings of *Tamarix* species.

Field establishment

Tamarix species are used in forestation in riparian and perilacustral (rivers and lakes) environments in the Mediterranean climate. They are adapted to saline soils, so they are especially recommended for revegetation of humid depressions and slightly saline coastal plains where other woody species have difficulty growing. *Tamarix* have traditionally been included in extensive landscaping and environmental restoration of degraded areas since they tolerate nitrified and contaminated soils. Some studies indicate that *Tamarix*

Table 11.68 Morphological attributes of 1-year-old seedlings of *Tamarix* spp. (observed values).

Attributes	Container seedlings	Bareroot seedlings (cm)
Shoot height (cm)	7–15 cm (from seeds) 60–200 cm (from cuttings)[a]	Up to 200

[a]Shoot height varies depending on the container size.
Li et al. (2010); unpublished data from the authors.

can be used for phytoremediation (Millán et al., 2007; Manousaki et al., 2008; Hegedüs et al., 2009).

For riverbank revegetation, a detailed study of the terrain must be carried out to identify sites particularly favorable for each species from a hydrogeomorphological point of view. If direct planting is to be carried out, it is essential to choose the correct elevation and install the cuttings close to the water to ensure proper rooting and subsequent growth. Container plants, once rooted, can be planted within the riparian environment in the band farthest from the riverbed, together with other species that will not shade it.

Tamarix can be planted in small areas or copses or by distributing the plants irregularly in the area to be revegetated. The planting density can vary depending on the goal of the intervention (recovery of ecological processes, improvement of river dynamics, protection of aquatic ecosystems, and increasing plant biodiversity, among others). Due to their heliophilous (shade-intolerant) nature and relatively slow growth compared with other riparian species, it is advisable to choose low planting densities of around 500–750 plants/ha. These densities can be significantly increased if direct planting is carried out since mortality is usually higher. Mixed stands with a varied structure is recommended and including other riparian or perilacustral species that fulfill varied ecological functions.

The most advisable time to establish plants or cuttings at their final location is at the end of the winter dormancy and before the buds sprout (end of January to end of February). To plant container cuttings, excavating the planting spot should be done manually or with the bucket of a small machine to avoid compacting the soil. It is not advisable to go deep in very compacted soils due to lack of oxygen or to have more than two buds protruding from the aerial part. Sprouting in spring, in the year of planting, does not always indicate success since it may occur at the expense of the reserves stored in the cuttings.

Abundant irrigation should be applied after planting so that the plants and cuttings come into contact with the soil and no air pockets remain. Maintenance irrigation may be required during the first and second summers, particularly in dry and sandy soils. In the case of wet, loamy soils, maintenance irrigation may be unnecessary.

The adaptive strategy of *Tamarix* species, combined with their low palatability, generally makes protection against herbivores unnecessary. If protection is required, light-colored screens or tree shelters should be used, as the plants prefer direct sunlight, and shade can affect their morphological and reproductive characteristics. To promote branched growth, pruning is recommended once or more during the first few years. Although plants in large containers are commercially available. they require greater care in the field after planting.

Chapter References

Baum, B.R., 1978. The genus *Tamarix*. In: Israel Academy of Sciences and Humanities, Jerusalem.

Chen, H., Hong, L., Ren, A., Yu, K., Wang, K., He, S., Liu, C., Xing, J., 2023. Growth regulators on the shooting and rooting of *Tamarix chinensis* stem cuttings. Rhizosphere 25. https://doi.org/10.1016/j.rhisph.2023.100679.

Di Tomaso, J.M., 1998. Impact, biology, and ecology of saltcedar (*Tamarix* spp.) in the Southwestern United States. Weed Technol. 12, 326–336.

Hegedüs, R., Kosáros, T., Gál, D., Pekár, F., Bíroné Oncsik, M., Lakatos, G., 2009. Potential phytoremediation function of energy plants (*Tamarix tetranda* Pall. and *Salix viminalis* L.) in effluent treatment of an intensive fish farming system using geothermal water. Agric. Environ. 1, 31–37.

Horton, J.S., Mounts, F.C., Kraft, J.M., 1960. Seed germination and seedling establishment of phreatophyte species. In: Fort Collins, CO: USDA, Forest Service, Rocky Mountain Forest and Range Experiment Station. Station Paper No. 48.

Li, W., Khan, M.A., Zhang, X., Liu, X., 2010. Rooting and shoot growth of stem cuttings of saltcedar (*Tamarix chinensis* Lour) under salt stress. Pak. J. Bot. 42, 4133–4142.

Li, C., Wei, X., Lan, H., 2021. The environmental adaptation strategy of seed germination, and roles of the seed pappus on dispersal and hypocotyl hairs on seedling anchorage in *Tamarix ramosissima*. AoB Plants 13 (6), plab065. https://doi.org/10.1093/aobpla/plab065.

Manousaki, E., Kadukova, A.J., Papadantonalis, N., Kalogerakis, N., 2008. Phytoextraction and phytoexcretion of Cd by the leaves of *Tamarix smyrnensis* growing on contaminated non-saline and saline soils. Environ. Res. 106, 326–332. https://doi.org/10.1016/j.envres.2007.04.004.

Millán, R., Carpena, R.O., Schmid, T., Sierra, M.J., Moreno, E., Peñalosa, J., Gamarra, R., Esteban, E., 2007. Rehabilitación de suelos contaminados con mercurio: estrategias aplicables en el área de Almadén. Ecosistemas 16, 56–66.

Navarro-Cerrillo, R.M., Gálvez, C., 2001. Manual para la identificación y reproducción de semillas de especies vegetales autóctonas de Andalucía. Tomo II. Consejería de Medio Ambiente. Junta de Andalucía, Córdoba.

Piotto, B., Di Noi, A. (Eds.), 2001. Propagazione per seme di alberi e arbusti della flora mediterranea. ANPA, Roma.

Reynolds, H.G., Alexander, R.R., 1974. *Tamarix*. In: Seeds of Woody Plants in the United States. USDA, Forest Service, Washington, DC, pp. 420–421. Agriculture Handbook 450.

SER, INSR, RBGK, SID, 2023. Seed Information Database (SID). Available from: https://ser-sid.org/

Shepperd, W.D., 2008. *Tamarix chinensis* Lour. In: Bonner, F.T., Karrfalt, R.P. (Eds.), The Woody Plant Seed Manual. Agriculture Handbook, vol 727. USDA, Forest Service, Washington, DC.

Tomanek, G.W., Zeigler, R.L., 1962. Ecological Studies of Tamarix. Fort Hays Kansas State College, Hays, Kansas.

Young, J.A., Clements, C.D., Harmon, D., 2004. Germination of seeds of *Tamarix ramosissima*. J. Range Manage 57, 475–481. https://doi.org/10.2111/1551-5028(2004)057[0475:GOSOTR]2.0.CO;2.

Taxus baccata L.

Oleh Hnatiuk[1], Taras Parpan[1], Xavier García-Martí[2] and Volodymyr Mahuran[3]
[1]Ukrainian Research Institute of Mountain Forestry Named After P.S. Pasternak. Department of Ecology and Forest protection., Ivano-Frankivsk, Ukraine; [2]Escola de Capatassos Agrícoles de Catarroja, Diputació de València, Catarroja, València, Spain; [3]National Forestry University of Ukraine, Lviv, Ukraine

Propagation

Seed propagation

Taxus baccata (yew) is a dioecious plant that sometimes behaves as a monoecious species (Thomas and Polwart, 2003). In male specimens, microsporangia appear in autumn in the leaf axils of the newest shoots. Female trees display megastrobili arranged in small cones, frequently with pairs of megastrobili positioned oppositely. It takes about 1 month from pollination to fertilization. Fertilization occurs from late May to early June, with pollination starting when the average daily air temperature exceeds 5°C and continues for 8–12 days and the seed germ appears in late autumn.

The seeds ripen in 6 weeks and are loosely encased by a fleshy red aril (Iszkuło et al., 2011; Stützel and Röwekamp, 1999). Female trees fruit annually and become reproductive at 20–30 years old if growing in open spaces and 70–120 years if growing in the understory (Hulme, 1996). The optimal time for harvesting *T. baccata* seeds is from mid-September to early November. Seeds can be harvested manually from the tree or by gently shaking the branches and picking the fruits from the ground. Seed processing includes the following operations: removal of seed arils by gentle crushing, washing in water, selecting clean seeds, and finally drying at 20–25°C. For large seed batches, the use of an industrial mixer (Fig. 11.50) equipped with a whisk attachment, followed by decantation to separate unviable seed and impurities is highly effective (García Martí, 2007).

The number of pure seeds in a high seed production year varies from 1.5–2.0 kg in small, 30-40-year-old individuals (the size of a bush) to 10 kg in 100-120-year-old trees. The average seed length is 6.4 ± 1.8 mm, and the diameter is 4.3 ± 1.3 mm (Hnatiuk and Huz, 2021). The weight of 1000 seeds ranges from 45–92 g. The average characteristics of seed lots are summarized in Table 11.69.

T. baccata seeds can be preserved for at least 5–6 years by drying them at room temperature to a moisture content of about 10% and then storing at 1–3 °C. Germination can be hypogeal and epigeal. In seed lots, a fraction germinates in the first year, while the rest do it in the second or even in the third year (Casals et al., 2015). While seed viability is approximately 100% (Thomas and Polwart, 2003), germination rates under natural soil conditions range from 50%–70% (Melzack and Watts, 1982). In nature, germination can be stimulated when fruits have been digested by frugivore birds and mammals (Martínez et al., 2008). Various techniques and recommendations have been suggested to break the dormancy and enhance germination. The main limitations



Figure 11.50 Top left. *Taxus baccata* fruit processing for seed separation and cleaning. Top right: Root formation in 2-year-old cuttings. Bottom left: Six-year-old *T. baccata* plants cultivated from cuttings (4 + 2) in pots. Bottom right: 2-year-old container seedlings. *(Oleh Hnatiuk (both pictures of cuttings); Xavier García-Martí (rest of the pictures).)*

to germination are a hard seed cover, immaturity of embryos, and occurrence of endogenous inhibitors like abscisic acid-ABA (Le Page-Degivry, 1973).

Nykolaeva et al. (1985) suggests a two-stage stratification process: first stratification in sand (also possible in peat) at 16°C for 3 months and then a second stratification at 5°C for another 3 months. They also advise soaking the seeds in a 0.5% $KMnO_4$ solution for

Table 11.69 Characteristics of seed lots of *Taxus baccata*.

Fruit to seed yield (% in weight)	Purity (%)	No. seeds/kg	Germination capacity (%)
11–32[a] (21.5)	91–93 (92)	11,000–21,000 (16,000)	20–98 (59)

Mean values in brackets.
[a]The yield of pure *T. baccata* seeds collected from small trees (the size of a bush) ranges from 12%–19%, while those collected from trees ranges 11%–32%.
Herbut and Turys (2008); Pers-Kamczyc and Suszka (2022); Schopmeyer (1974) and unpublished results by Hnatiuk.

2 h before stratification. Suszka (1986) proposed a three-phase pretreatment as follows. In the first phase, seeds are stratified for 6–6.5 months at alternating temperatures of 15°C for 24 h and 20°C for the next 24 h. In the second phase the seeds are maintained at a constant temperature of 3°C for 4–4.5 months. In the last phase, seeds are exposed daily to alternating temperatures of 3°C for 16 h and 20°C for 8 h. The methodology developed by Boiko (2014) entails mixing the seeds with wet river sand and then drying the clean seeds for 2–4 days in a ventilated room. Afterward, they can be stored for 4–5 months in a refrigerator at 4–6°C. In the spring of the following year, the seeds can be sown in containers filled with a mixture of deciduous forest soil, peat, and sand in a 2:1:1 (v:v:v) proportion.

Lisoviy et al. (2016) found that after 18 months of stratification that included 6 months of storage at 10–12°C in a cool, dry place and 12 months of soil burial at a depth of 10–15 cm outdoors, up to 75% of seeds germinated over 3 years. Specifically, 7%–10% germinated in the first year, 40%–50% in the second year, and an additional 10%–15% in the third year. By the end of the first growing season, seedlings reached a height of 3–5 cm. By the end of the second season, the average height of the seedlings was about 8.0 cm, ranging from 3.9 to 12.5 cm. In the third year, the average height of the seedlings was 10.2 cm, with a range from 4.4 to 13.7 cm.

Vegetative propagation

T. baccata can be propagated using winter (woody) and summer (green) cuttings. Vegetative propagation by cuttings is carried out in spring (May) and autumn (September). Typical *T. baccata* cuttings have a length of 15–20 cm with some lateral shoots. The needles should be removed from the lower part of the cutting, and the cuttings kept in water for 1 day. The following mixtures can be used as rooting media: peat + sand both at 2:1 or 1:1; peat + black soil (1:1) (Lisoviy et al., 2016); forest soil + peat + farmyard manure (1:1:1) (Das and Jha, 2018a); soil + vermicompost + farmyard manure (2:1:1) (Chauhan, 2018). The most used plant growth regulators for the rooting of cuttings (Fig. 11.50) include 0.5%–1.0% IBA (indole-3-butyric acid), 0.5%-1.0% NAA (α-naphthalene acetic acid), and 1.2% K-IBA (indole-3-butyric acid potassium salt (Bayraktar

et al., 2018; Das and Jha, 2018b; Farkas and Kovács, 2017; Karimi and Moradi, 2018; Lisoviy et al., 2016; Pipinis et al., 2023). Cuttings should be planted in greenhouses. They require regular watering throughout the summer and autumn. Cuttings planted in spring have a rooting rate of around 45%, but high summer temperatures can cause some mortality. Rooting in autumn typically yields better results, with a 52% success rate. Rooting success depends largely on the genetic potential of individuals, with rooting close to 100% being obtained in some genotypes (Schneck, 1996). When possible, it is advisable to collect epicormic shoots from donor trees to avoid plagiotropism and obtain plants with a defined apical meristem.

Cuttings should be spaced 5 × 5 cm apart when planted. In the first year of open-field cultivation, cuttings should be watered twice a week from April to August and once a week from September to October. In the second year, watering must be weekly from March to August. From the third year onwards, water 12 times a year from March to August. In greenhouses, weeding is performed up to 10 times during the first year and up to 5 times in the following years.

T. baccata is rarely grafted, an activity mainly reserved for the propagation of ornamental individuals (Dagmar and Szekely, 2010; Túri-Farkas and Kovács, 2016). Three-year-old plants grown in pots are generally used as rootstock (Zachara and Lukaszewicz, 2002). Grafting success is higher if performed in early spring (February–March) in greenhouses with high air humidity. Grafting of *T. baccata* can be accomplished using various traditional methods such as cleft grafting, crown grafting, splice grafting, and approach grafting. The approach grafting method is typically performed in spring, prior to the onset of shoot growth, between March and April. A narrow film tape 1–1.5 cm wide and up to 30 cm long is used for tying. Four weeks after grafting, the strapping can be removed. The rootstock crown should be removed gradually. The first pruning of rootstock branches is recommended after 12 months. Studies of this method of grafting of *T. baccata* note a positive result when grafting *T. baccata* + *T. baccata cv. Aurea* (65%) and *T. baccata* + *T. x media cv. Hicksii* (80%).

Since this species is dioecious, Asadulaev et al. (2014) used a method that involved grafting cuttings from female plants onto the crown of a male plant. They used crown and splice grafting, both of which are performed using cuttings. To do this in natural populations, female trees are selected as mother plants, from which cuttings are harvested (September–October). These cuttings are transplanted into special containers with sand and placed in a refrigerator until the next spring. Next, young male plants of up to 15 years are selected for grafting. During the period of sap flow (in February–March), an oblique cut is made on the male tree and female cuttings 10–15 cm long, with a diameter corresponding to the diameter of the rootstock, are inserted into the 'butt'. Then the sections are tied with budding tape and the upper part of the cutting is covered with garden varnish.

Less commonly, *T. baccata* can be vegetatively propagated by air layering (Ishtiyak and Puni, 2017). In March–April, bark is stripped from a branch forming a 1.25 cm wide ring using a grafting knife. The debarked ring is smeared with rooting-stimulating hormones, covered with a handful of sphagnum moss, and then wrapped with a 17 × 12 cm plastic strip. After successful rooting, the branches are separated from the mother plants 5 cm below the rooting site and planted in plastic bags with a soil substrate, namely soil + sand + vermicompost + vermicompost (in a ratio of 2:1:1) to develop a higher number of roots.

In vitro propagation of *T. baccata* is generally challenging due to issues such as tissue recalcitrance, contamination, and poor field rooting. For the initial stage of *T. baccata* microcloning, which includes sterilization and the introduction of explants in vitro, the use of Litvay's medium or a modified Murashige and Skoog (MS) culture medium with added 2,4-D and BAP (6-benzylaminopurine) is recommended. MS-based media have proven to be highly effective in inducing callus formation in *T. baccata* and promoting callus growth (Mihaljević et al., 2002).

The highest induction of callus in *T. baccata* was achieved on half-strength MS medium containing one-fourth nitrogen (KNO_3, NH_4NO_3), enriched with glutamine, 1 mg/L 2,4-D, and 1 mg/L Kinetin from stem tissues (Baharak et al., 2014). The largest callus formation was noted in leaf cultures on half-strength MS medium with added glutamine, 2 mg/L NAA, and 0.2 mg/L Kinetin. For meristematic cell observation, tissues were cultured on half-strength MS medium supplemented with 0.4 mg/L 2,4-D and 3 BAP for 7–8 weeks. During micropropagation, the inclusion of activated charcoal (2 g/L) in the medium led to an increase in the average number of new leaves and promoted shoot elongation. The most significant shoot elongation and new leaf growth occurred in MS medium with 3 mg/L Kinetin after 1 month. Optimal rooting of the elongated shoots was achieved in WPM medium devoid of growth regulators.

Abbasin et al. (2010) developed a simple procedure for rapid callus initiation and in vitro propagation of *T. baccata* using mature shoots and germplasm. In this study, bud explants and embryos of *T. baccata* were cultured on various media, supplemented with different concentrations and combinations of cytokinin and auxin. Using 2 mg/L BAP as a cytokinin resulted in the best shoot proliferation and growth of shoot tips when cultured on MS medium. On the basis of this experience, combining 2 mg/L BAP and 0.3 mg/L NAA showed an increase in callus and the number of regenerated shoots, up to eight shoots per embryo culture on half-strength MS. Excellent formation of roots was observed in half-strength MS salts combined with forest soluble soil composition and 8 mg/L IBA. The maximum callus growth was observed using at 0.25 mg/L Kinetin and 2 mg/L NAA treatment (Mahdinejad et al., 2015).

Seedling cultivation

Container plants

Cultivating *T. baccata* from seeds in containers is challenging due to its slow growth and the difficulties associated with low, delayed seed germination, which often leads to a preference for vegetative propagation methods. Beyond ornamental purposes (see Section 3), vegetative production of yew has been used in *T. baccata* conservation programs to establish clonal banks and *ex situ* and *quasi* in situ collections to preserve the genetic heritage of the remaining yew populations in southern Spain (Vaquero-de la Cruz and Iglesias-Sauce, 2007; García-Martí, 2007).

Despite being more difficult than vegetative propagation, the production of plants from seeds potentially favors genetic variability and this stock type shows higher survival than plants produced from cuttings, at least in Mediterranean forest restoration projects (García-Martí unpublished data). This is likely due to a better conformation of the root system. In the nursery, pretreated seeds are usually sown in seed trays with good drainage. Most seedling emergence (>50%) occurs in the second spring. Once the seedlings show the two well-developed embryonic leaves, they can be carefully transplanted into 300–500 mL forest containers, 20–30 cm deep (Fig. 11.51). Container seedlings used for forest restoration are usually at least 2 years old and if well fertilized they can reach a height of about 15–25 cm and produce a well-formed plug (Nicolás Peragón et al., 2015) (Fig. 11.50). It is also possible to use 3- 4-year-old plants cultivated in 2.5–3.5 L containers. Conventional growing media formulations consist of blonde peat (Nicolás Peragón et al., 2015) or mixtures of peat and coir in equal proportions that can be supplemented with some inorganic component such as perlite or vermiculite.

For cutting-grown plants (Fig. 11.50), the volume of the container will depend on plant age as follows: 1) transplant 2-year-old rooted cuttings (2 + 0) into a container with a volume of 0.5 L; 2) transplant 3-year-old cuttings (2 + 1) into a container with a volume of 2.0 L; 3) transplant 4-year-old cuttings (2 + 2) into a container with a volume of 5.0 L.

It is recommended to shade seedlings in the nursery and fertilize them with a soluble fertilizer at an annual rate of approximately 3.5 g N/m^2 (Elton and Treaster, 1982). Navarro-Cerrillo et al. (2013) suggested mixing slow-release fertilizers within the growing medium at a rate of 2 g/L substrate (NPK proportion of 18-11-10 or 14-8-15), with a release time of 8–9 months. Nicolás Peragón et al. (2015) observed the highest outplanting growth in 2 + 0 seedlings fertilized with 376 mg N per plant distributed over two growing seasons.

Morphological attributes of container plants of *T. baccata* cultivated from cuttings depend on the microclimatic conditions in greenhouses and the source material. According to our experience in Eastern Europe, 4-year-old cuttings (2 + 2) reach a height

of 25–27 cm, and 6-year-old cuttings (4 + 2) reach 30–40 cm (Fig. 11.51). *T. baccata* container plants are especially sensitive to frost, which can damage the roots.

Bareroot plants

The technology for cultivating seedlings of various species in open ground encompasses several key operations: thorough and ongoing tillage, sowing in autumn or spring, managing weeds, and aerating the overly compacted substrate found between rows. It is recommended to sow pretreated *T. baccata* seeds in spring in rows 8–10 cm wide, 4 cm deep, and 20–25 cm between rows, with a seeding rate of 180–200 seeds/m. Seeds should be covered with a thin layer of beech forest litter and seedlings emerge 1 month after sowing (Herbut and Turys, 2008).

The care of seedlings during the first 5 years of cultivation includes weeding and loosening of the soil after emergence, fertilizing, irrigation and, if necessary, the use of chemicals to control harmful insects and diseases. Care should be performed 3-4 times every growing season, approximately once a month, starting in the third week of May. In the third year of cultivation, seedlings must be transplanted to increase spacing between plants according to a 10 × 10 cm scheme, and later, for 6- to 8-year-old seedlings to a 20 × 20 cm scheme. Some morphometric parameters of *T. baccata* bareroot plants of different ages are shown in Table 11.70.

Preparation, storage and transportation of *T. baccata* seedlings are similar to other conifers (see Landis et al., 2010). Seedlings should be transported as quickly as possible, covering the plants to prevent dehydration and protect the roots. Upon arrival at the planting site, the seedlings should be stored in trenches >25 cm deep, the roots covered with a layer of soil 8–10 cm thick and compacted gently to prevent desiccation and watered as needed.

The diseases and pests of *T. baccata* are not well known. Several diseases have been reported such as the brown felt blight (*Herpotrichia juniperi* (Duby) Petr.) and phomosis (*Phoma* spp.) (Shutko and Tuturzhans, 2016). Ivanova and Bernadovičová (2009) report the occurrence of the fungus *Cryptocline taxicola* (All.) Petr. on needles of yew.

Rhizoctonia solani Kühn (mycelial stage of *Thanatephorus cucumeris* (Frank)), *Phytophthora cinnamomi* Rands., and various *Pythium* spp. are considered to cause the late blight in yew seedlings (Hepting, 1971). In addition, various species of *Fusarium* spp. have also been reported to damage young yew plants in Poland (Siwecki, 1975). In large nurseries, the root system of yew plants can be affected by nematodes (roundworms).

Field establishment

The *T. baccata* is a valuable ornamental species widely used in garden design, green building and landscape design due to its esthetic appeal, low growing requirements, and resistance to diseases and pests. Most *T. baccata*, apart from some modern dwarf varieties,



Figure 11.51 Left: *T. baccata* sapling protected against ungulates with wire. Right: Recruited *T. baccata* juvenile more than 8 years after planting in a Mediterranean site. *(Xavier García-Martí.)*

grow as trees with one or more distinct trunks rather than shrubs with a diverse shape and growth characteristics. Its tolerance for pruning makes it ideal for topiary art. However, the high toxicity of its needles, bark, and seeds makes it unsuitable for hedges near playgrounds, schools, or areas with livestock, although deer and hares can consume yew bark and needles without harm.

General recommendations on nursery cultivation and small-stand establishment in Eastern Europe can be consulted in Herbut and Turys (2008). Two main challenges hinder active restoration of *T. baccata* populations: the complexity and high cost of producing quality planting material and the difficulty of establishing planted juveniles. Natural regeneration is often limited by current unfavorable environmental conditions in *T. baccata* populations, necessitating human intervention to assure restoration (Fig. 11.51). *T. baccata*

Table 11.70 Morphological attributes of bareroot seedlings of *Taxus baccata*.

	Age (years)				
Attribute	**1**	**2**	**3**	**4**	**5**
Shoot height (cm)	3.1 ± 0.8	8.6 ± 2.2	10.0 ± 1.0	12.7 ± 3.4	17.8 ± 4.0
Root collar diameter (cm)	0.10	0.15	0.16	0.17	0.28
Shoot dry weight (g)	0.03 ± 0.01	0.06 ± 0.05	0.09 ± 0.03	0.17 ± 0.09	0.29 ± 0.05
Root dry weight (g)	0.02 ± 0.01	0.04 ± 0.02	0.09 ± 0.04	0.17 ± 0.07	0.33 ± 0.06
Needle dry weight (g)	0.07 ± 0.01	0.10 ± 0.03	0.16 ± 0.05	0.23 ± 0.14	0.36 ± 0.04

Boiko (2014); Hnatiuk and Huz (2021); Korshykov (2019); Lisoviy et al. (2016) and unpublished results by O. Hnatiuk.

individuals ranging from 31 to 45 years grow slower than coexisting beech, oak, and firs of comparable age (Hnatiuk et al., 2020), and typically reach heights between 1.3 and 3.3 m and dbh spanning 1.5–3.1 cm. Herbivory by wild and domestic ungulates is a main environmental limitation for natural regeneration and the success of plantations across Europe (Thomas and Polwart, 2003; Piovesan et al., 2009). To prevent herbivory, individual mesh shelters (up to 1.5–2 m tall and 0.5 m in diameter) (Fig. 11.51) should be used to protect plants in small, planted patches. Larger plantations should be fenced using 2–2.5 m tall fences, preferably made of wooden posts and mesh (Serra, 2007). Shrubs in the understory can also provide protection from herbivores (García et al., 2000).

In southern Europe under Mediterranean climatic conditions, drought during the wet season can hinder *T. baccata* plantings in some years. On drought-prone sites, summer drought is a strong limitation for plantation success and dry season irrigation during the first 2–3 years is essential for the establishment of nursery-grown seedlings. As a shade-tolerant species, *T. baccata* seedlings with a minimum height of 20–30 cm are effective for planting plantation understories. In Mediterranean regions plants can be established under pine species such as *P. sylvestris* (Nicolás Peragón et al., 2015) or deciduous trees (Herbut and Turys 2008). In the eastern Iberian Peninsula with Mediterranean climate, Camprodon et al. (2016) observed that irradiance should not exceed 40%–50% for successful establishment. In line with this recommendation, Nicolás Peragón et al. (2015) observed higher survival under the canopy of a *P. sylvestris* plantation (irradiance <30%) compared with open sites in Mediterranean mountains. Once seedlings are established, these authors, however, recommend increasing irradiance by overstory thinning to promote growth (Fig. 11.51). In open sites, wire shelters wrapped with shade fabric can protect young plants from high irradiation and temperature during their early years (Camprodon et al., 2016). For soil preparation, we recommend mechanical (auger or small backhoes) or manual digging of holes 40 × 40 cm wide and 60 cm deep, avoiding very stony microsites. Planting can be done between October to December on mild winter sites, or in March on cold winter sites.

In Mediterranean mountain areas, we recommend planting yew with other species native to *T. baccata* habitats such as *Ilex aquifolium*, *Sorbus* spp., *Crataegus monogyna*, *Juniperus* spp. and spiny and fleshy fruited shrubs to increase plant-animal interactions. These companion species can attract frugivores to aid *T. baccata* seed dispersal and protect against browsers (Martínez et al., 2008; García-Martí and Ferrer, 2013).

The maintenance practices of *T. baccata* plantations should continue for at least 4–5 years, with 4–5 interventions per growing season to remove fast-growing woody species such as *Betula* and *Carpinus* spp. Silvicultural care of yew forest stands should be carried out for at least 40–60 years directed to increase light in young trees plants and provide mechanical protection until they reach a height of 3–5 m.

Chapter References

Abbasin, Z., Zamani, S., Movahedi, S., Khaksar, G., Tabatabaei, B.E.S., 2010. In vitro micropropagation of yew (*Taxus baccata*) and production of plantlets. Biotechnology 9 (1), 48–54.

Asadulaev, Z., Abdullatipov, R., Omarova, P., Aliev, H., Sadykova, G., 2014. Method of restoration of yew populations. In: Russian Patent RU 2516347 C2.

Baharak, B.S., Mansoor, O., Reza, N.M., Farhad, H.A., Sepideh, K.J., Mehdi, S., Mohammad, S., 2014. Effect of explants, salts concentration medium and hormone treatments on *Taxus baccata* in vitro culture. Int. J. Biosci. 5 (6), 1–9.

Bayraktar, A., Atar, F., Yildirim, N., Turna, I., 2018. Effects of different media and hormones on propagation by cuttings of European yew (*Taxus baccata* L.). Šumarski list 142 (9–10), 509–516. https://doi.org/10.31298/sl.142.9-10.6.

Boiko, N., 2014. Seasonal development of introduced populations of yews (*Taxus* L.) in the conditions of the state dendrological park "Alexandria". Biology 20, 17–21.

Camprodon, J., Guixé, D., Casals, P., Caritat, A., Buqueras, X., García-Martí, X., Reverté, J., Rios, A.I., Beltrán, M., Llovet, J., Taüll, M., Vives, A., Àguila, V., Casas, C., 2016. Conservation of mediterranean yew forests. In: Practice Handbook, B., Camprodon, J., Guixé, D. (Eds.), Life TAXUS Project. Forest Science Centre of Catalonia.

Casals, P., Camprodon, J., Caritat, A., Ríos, A.I., Guixé, D., Garcia-Martí, X., Martín-Alcón, S., Coll, L., 2015. Forest structure of Mediterranean yew (*Taxus baccata* L.) populations and neighbor effects on juvenile yew performance in the NE Iberian Peninsula. For. Syst. 24 (3), 42. https://doi.org/10.5424/fs/2015243-07469.

Chauhan, J., 2018. Regeneration of *Taxus baccata* through vegetative propagation using different concentrations of IBA and NAA. J. Pharmacogn. Phytochem. 7 (4), 1853–1857. https://www.phytojournal.com/archives?year=2018&vol=7&issue=4&ArticleId=5202.

Dagmar, V., Szekely, G., 2010. English yew trees in the parks of Timisoara. J. Horticult. Forest. Biotechnol. 14 (3), 154–156. http://journal-hfb.usab-tm.ro/romana/Lista%20lucrari%202010_PDF/JHFB%20-%20vol.3/31Visoiu%20Dagmar.pdf.

Das, S., Jha, L.K., 2018a. Effect of different rooting media on root proliferation of *Taxus baccata* L. stem cuttings. Curr. Agric. Res. J. 6 (1), 95–104.

Das, S., Jha, L.K., 2018b. Optimization of conditions for vegetative propagation of *Taxus baccata* by shoot cuttings - effects of IBA, growth stage and seasonal differences. Ann. Plant Sci. 7 (2), 2082–2087. https://doi.org/10.21746/aps.2018.7.2.18.

Elton, S.M., Treaster, S.A., 1982. Growth of *Taxus cuspidata* "Thayeri" produced in containers. Res. Circ. 268, 3–5. https://core.ac.uk/download/pdf/159584614.pdf#page=3.

Farkas, T.Z., Kovács, D., 2017. Propagation of *Taxus baccata* by cuttings. Manage. Agricol. 19 (2), 39–42. http://lsma.ro/index.php/lsma/article/view/1060/pdf.

García, D., Zamora, R., Hódar, J.A., Gómez, J.M., Castro, J., 2000. Yew (*Taxus baccata* L.) regeneration is facilitated by fleshy-fruited shrubs in Mediterranean environments. Biol. Conserv. 95, 31–38.

García-Martí, X., 2007. Producción de material forestal de *Taxus baccata* L. destinado a planes de conservación. In: El tejo en el Mediterráneo occidental: Jornadas Internacionales sobre el tejo y las tejeras en el Mediterráneo occidental. Conselleria de Territori i Habitatge. Generalitat Valenciana, Valencia, Spain, pp. 141–152.

García-Martí, X., Ferrer, P., 2013. La creación de núcleos de dispersión – reclamo como modelo de restauración ecológica forestal. Avances en la restauración de sistemas forestales. In: de implantación, T., Martínez-Ruiz, C., Lario, F.J., Fernández-Santos, B. (Eds.), SECF-AEET. España, Madrid, pp. 149–159.

Hepting, G.H., 1971. Diseases of Forest and Shade Trees of the United States. USDA.

Herbut, F., Turys, E., 2008. On the issue of renaturalization of yew (*Taxus baccata*) in the Carpathians. NVU Biol. 22, 63–66. https://dspace.uzhnu.edu.ua/jspui/handle/lib/15501.

Hnatiuk, O., Huz, M., 2021. Features of Growing *Taxus baccata* L. Planting Material by Seed Method. NLTU, Lviv.

Hnatiuk, O., Mohytych, V., Sułkowska, M., 2020. Distribution, protection and restoration of L. in Ukraine. Folia For. Pol. 62, 327–335. https://doi.org/10.2478/ffp-2020-0031.

Hulme, P.E., 1996. Natural regeneration of yew (*Taxus baccata* L.): microsite, seed or herbivore limitation? J. Ecol. 84 (6), 853–861. https://doi.org/10.2307/2960557.

Ishtiyak, P., Puni, L., 2017. Standardization of in-situ propagation technique for *Taxus baccata* Linn. an endangered medicinal plant of Western Himalayas. Ethno-Med. 11 (4), 332–340. https://doi.org/10.1080/09735070.2017.1305231.

Iszkuło, G., Jasińska, A.K., Sobierajska, K., 2011. Dendroecological differences between *Taxus baccata* males and females in comparison with monoecious *Abies alba*. Dendrobiology 65, 55–61. https://agro.icm.edu.pl/agro/element/bwmeta1.element.agro-45807ef4-f92d-468b-a0a6-b6c9db376282.

Ivanova, H., Bernadovičová, S., 2009. Needle disease on English yew caused by the parasitic fungus *Cryptocline taxicola* (All.) Petr. Not. Nitra Town *Folia Oecologica* 36 (2), 79–85. https://ife.sk/wp-content/uploads/2016/10/FO_36_2_Ivanova.pdf.

Karimi, M., Moradi, H., 2018. The effect of cutting texture type and hormonal treatment with combination rooting cofactors on rooting of yew (*Taxus baccata* L.). Iran. J. Forest 9 (4), 541–553. https://www.ijfisaforestry.ir/article_57290_ef28c12207f12a9d343b9515f775d244.pdf.

Korshykov, I.I., 2019. Viability of common yew (*Taxus baccata* L.) plants in conditions of Kryvyi Rih. Plant Introduct. 81 (1), 58–67. https://doi.org/10.5281/zenodo.2650461.

Landis, T.D., Dumroese, R.K., Haase, D.L., 2010. The container tree nursery manual: seedling processing, storage, and outplanting. In: Agriculture Handbook. USDA Forest Service, Washington, DC. USA.

Le Page-Degivry, M.T., 1973. An embryo-culture study of embryo dormancy in *Taxus baccata*. Biol. Plant. 15, 264–269.

Lisoviy, M., Huz, M., Hnatiuk, O., 2016. Peculiarities of seed and vegetative propagation of berry yew. Sci. Bullet. UNFU 26 (4), 99–110. https://doi.org/10.15421/40260416.

Mahdinejad, N., Fakheri, B.A., Ghanbari, S., 2015. Effects of growth regulators on in vitro callogenesis of *Taxus baccata* L. Biol. Forum 7 (1), 142–145. https://www.cabidigitallibrary.org/doi/full/10.5555/20153254892.

Martínez, I., García, D., Obeso, J.R., 2008. Differential seed dispersal patterns generated by a common assemblage of vertebrate frugivores in three fleshy-fruited trees. Ecoscience 15 (2), 189–199.

Melzack, R.N., Watts, D., 1982. Variations in seed weight, germination, and seedling vigour in the Yew (*Taxus baccata*, L.) in England. J. Biogeogr. 9 (1), 55–63. https://doi.org/10.2307/2844730.

Mihaljević, S., Bjedov, I., Kovač, M., Levanić, D.L., Jelaska, S., 2002. Effect of explant source and growth regulators on in vitro callus growth of *Taxus baccata* L. Washingtonii. Food Technol. Biotechnol. 40 (4), 299–303. https://www.academia.edu/download/87285803/263225.pdf.

Navarro-Cerrillo, R., Plaza, L., Sánchez, A., Mallofret, E., Arroyo, M., Marchal, F., Lara, M.A., 2013. *Taxus baccata* L. In: Pemán, J., Navarro-Cerrillo, R.M., Nicolás, J.L., Prada, M.A., Serrada, R. (Eds.), Producción y manejo de semillas y plantas forestales. Tomo 2, pp. 501-523, Organismo de Parques Nacionales. Ministerio de Agricultura, Alimentación y Medio Ambiente. Madrid, Spain.

Nicolás Peragón, J.L., Benito Matías, L.F., Puértolas Simón, J., 2015. Restoration of European yew (*Taxus baccata* L.) in Mediterranean mountains: importance of seedling nursery fertilization and post-planting light levels. Forest Syst. 24 (3). https://doi.org/10.5424/FS/2015243-07464 e041–e041.

Nykolaeva, M., Razumova, M., Hladkova, V., 1985. Handbook on Germination ofDormant Seeds. Science, Leningrad.

Pers-Kamczyc, E., Suszka, J., 2022. Long-term maternal fertilizer addition increased seed size but decreased germination capacity and offspring performance in *Taxus baccata* L. Forests 13 (5), 670. https://doi.org/10.3390/f13050670.

Piovesan, G., Presutti Saba, E., Biondi, F., Alessandrini, A., Di Filippo, A., Schirone, B., 2009. Population ecology of yew (*Taxus baccata*, L.) in the Central Apennines: spatial patterns and the irrelevance for conservation strategies. Plant Ecol. 205, 23–46.

Pipinis, E., Kostas, S., Hatzilazarou, S., Petropoulos, S., Mitsi, D., Stampoulidis, A., Milios, E., Smiris, P., 2023. Effects of donor tree age, cutting collection time and K-IBA application on rooting ability of *Taxus baccata* L. stem cuttings: preliminary results. Folia Oecol. 50 (1), 97–103. https://doi.org/10.2478/FOECOL-2023-0009.

Schneck, V., 1996. Studies on influence of clone on rooting ability and rooting quality in the propagation of cuttings from 40-to 350-year-old *Taxus baccata* L. ortets. Silvae Genet. 45 (5), 246–249.

Schopmeyer, C.S., 1974. Seeds of woody plants in the United States. In: Agriculture Handbook. USDA Forest Service, Washington, DC.

SER, INSR, RBGK, SID, 2023. Seed Information Database (SID). Available from. https://ser-sid.org/.

Serra, L., 2007. El tejo en el Mediterráneo Occidental. Jornadas Internacionales sobre el tejo y las tejeras en el Mediterráneo Occidental. In: Generalitat Valenciana, Conselleria de Territori i Habitatge. Spain.

Shutko, A.P., Tuturzhans, L.V., 2016. Needle diseases of yew (*Taxus baccata* L.) in the northwest Caucasus. Forest J. 13, 266–267. https://www.cabidigitallibrary.org/doi/pdf/10.5555/20173342121.

Siwecki, R., 1975. Diseases and parasitic insects of *Taxus baccata*. In: Białobok, S., Bugała, W., Boratyński, A. (Eds.), Nasze Drzewa Leśne. Monografie Popularnonaukowe. Państwowe Wydawnictwo Naukowe, Warszaw, pp. 125–133.

Stützel, T., Röwekamp, I., 1999. Female reproductive structures in Taxales. Flora 194 (2), 145–157. https://doi.org/10.1016/S0367-2530(17)30893-9.

Suszka, B., 1986. Conditions for after-ripening and germination of seeds and for seedling emergence of English yew (*Taxus baccata* L.). Arbor. Kornickie 30, 285–338. https://rcin.org.pl/Content/142544/PDF/KOR001_148545.pdf.

Thomas, P.A., Polwart, A., 2003. *Taxus baccata* L. J. Ecol. 91 (3), 489–524. https://www.jstor.org/stable/3599563.

Túri-Farkas, Z., Kovács, D., 2016. Propagation of *Taxus baccata* 'Green Diamond' by cutting. Agricult. Rural Develop. 5 (1–2), 71–76. https://doi.org/10.14232/rard.2016.1-2.71-76.

Vaquero-de la Cruz, J., Iglesias-Sauce, S., 2007. Conservación del tejo (*Taxus baccata* L.) en España. In: Serra, L. (Ed.), El tejo en el Mediterráneo occidental: Jornadas Internacionales sobre el tejo y las tejeras en el Mediterráneo occidental. Generalitat Valenciana, Conselleria de Territori i Habitatge, Spain, pp. 13–23.

Zachara, T., Lukaszewicz, J., 2002. Propozycje postepowania hodowlanego z cisem (*Taxus baccata* L.) w lasach. Sylwan 146 (04), 5–16. https://agro.icm.edu.pl/agro/element/bwmeta1.element.agro-article-04f11015-94ac-45f7-9b91-48b910a008a7.

Tilia tomentosa Moench.

Evgeni Tsavkov[1], Ina Aneva[2] and Petar Zhelev[1]
[1]University of Forestry, Sofia, Bulgaria; [2]Department of Plant and Fungal Diversity and Resources, Institute of Biodiversity and Ecosystem Research, Bulgarian Academy of Sciences, Sofia, Bulgaria

Propagation

Seed propagation

Tilia tomentosa (silver lime or silver linden) fruits are harvested manually from early September to November. Each fruit contains one or two seeds. After removing the leaf attached to the fruit cluster, no further processing is necessary. Fruits can be harvested at the physiological maturity stage, in early September for direct sowing. Fruits collected after this stage can be stored under dry or low temperature conditions. At 12–14°C and 12%–14% air relative humidity, seeds remain viable for up to 2 years; at 1–4°C and 8%–12% relative humidity they can be stored for 3–5 years. Finally, at −4 and −10°C and 7%–8% relative humidity viability extends to 5–8 years (Milev et al., 2004; Nikolova et al., 2021). Seed characteristics of Tilia tomentosa are shown in Table 11.71.

T. tomentosa seeds show deep dormancy. If collected after the physiological maturity stage, they need 5–6 months of stratification at 1–3°C in moist sand or a 1:1 mixture of sand and perlite before sowing. Treatment with gibberellic acid (GA_3) at 100–200 mg/L for 24 h and with kinetin at 1 mg/L, effectively breaks seed dormancy (Magherini and Nin, 1993, 1994; Pigott, 1996). Treatment with concentrated sulfuric acid for 5 min increases germination (Magherini and Nin, 1993, 1994). A complex protocol involving mechanical removal of the pericarp, scalding seeds for 2 h in boiling water, soaking in 200 mg/L GA_3 solution for 24 h, followed by stratification at 15–20°C for 5 days and at 0–5°C for 15 days achieves a germination rate >70% (Xiangliang et al., 2008).

Vegetative propagation

Vegetative propagation using semihardwood shoot cuttings is difficult. However, treatment with indole-butyric acid (IBA) can result in a rooting success >30% (Magherini and Nin, 1994). Cuttings taken from suckers show significantly higher rooting, up to

Table 11.71 Characteristics of seed lots of *Tilia tomentosa*.

Purity (%)	No. seeds/kg	Germination capacity (%)
95–98 (97)	8,000–17,000 (12,500)	20–70 (45)

Mean values in brackets.
Magherini and Nin (1993); Milev et al. (2004).

70% and are not influenced by IBA treatment (Magherini and Nin, 1994). Rooting can be achieved in a greenhouse or hot beds at 12–15°C. Cuttings should be about 20 cm long and inserted 2/3 of their length into the rooting medium, which can be perlite or a (3:1 v:v) mix of perlite and peat (Magherini and Nin, 1994).

Seedling cultivation

Container plants

There is little information about container production of *T. tomentosa* seedlings, with a few exceptions in ornamental nurseries. For other *Tilia* species, good cultivation results are obtained by using a substrate mixture of peat, organic fertilizer, and perlite (3:1:1) and container size of 1–1.5 L (Li et al., 2023). The roots of *Tilia* species tend to form coils when grown in containers and it is important to straighten and stretch them downwards when planting (Pigott, 2012). Mycorrhization with *Paxillus involutus* can reduce water stress and promote growth (Serafim et al., 2024).

Bareroot plants

Autumn sowing is recommended for *T. tomentosa*. Seeds can be sown immediately after harvest or in October–November using seeds collected in September at the physiological maturity stage. Until sowing, seeds can be stored covered with a moist sand layer up to 20 cm thick. Sowing in the beds should be done in rows at a depth of 3–4 cm, with 30 seeds/m. The rows should be covered with sand, and all the seedbed must be mulched with a 3–5 cm thick layer of wood bran or straw. In early Spring (March), seed status should be checked periodically. Once germination begins, the mulch should be gradually removed. Seeds stored longer need stratification as described in section 1.1.

T. tomentosa seedlings are highly sensitive to frost, direct sunlight, and drought. The minimum desired size for seedlings is 25 cm in height and 0.6–0.8 cm in root collar diameter, which is usually achieved in 2 + 0 seedlings (Milev et al., 2004). However, with suitable fertilization (N:P:K 10:10:10) and irrigation, it is possible to achieve these size standards in the first year of cultivation (Milev et al., 2004).

Field establishment

T. tomentosa requires fertile, deep, low-acid or neutral (pH 6.2–7.2) mineral soils (Radoglou et al., 2009), but generally, it can grow in a wide range of ecological conditions (Frischbier et al., 2019; Glatthorn et al., 2024). Within its range, it is often exposed to dry periods with drought being the main factor limiting seedling establishment and early growth. Intense soil preparation is desirable to facilitate root growth, as this species develops a deep root system (Pigott, 2012). Holes opened with mechanical augers can be used in small scale plantings. Planting in Spring (March–April) is preferred. The planting

density is usually 2 × 1 m or 2 × 1.5 m (Kalmukov, 2012). The species is shade-tolerant, but the seedlings can grow both under full sun and on partially shaded sites (Frischbier et al., 2019). In some regions, animals like hares, rabbits, and roe deer can cause significant damage. Plastic tree shelters with higher light transmissivity (Oliet et al., 2021) are effective for protecting the seedlings (Pigott, 2012).

Chapter References

Frischbier, N., Nikolova, P.S., Brang, P., Klumpp, R., Aas, G., Binder, F., 2019. Climate change adaptation with non-native tree species in Central European forests: early tree survival in a multi-site field trial. Eur. J. For. Res. 138, 1015–1032. https://doi.org/10.1007/s10342-019-01222-1.

Glatthorn, J., Maier, B., Binder, F., Brang, P., Frischbier, N., Horna, V., Klumpp, R., Nikolova, P.S., Aas, G., 2024. Limited influence of air temperature and precipitation on six-year survival and growth of non-native tree species in a Central European multi-site field trial. For. Ecol. Manag. 553, 121645. https://doi.org/10.1016/j.foreco.2023.121645.

Kalmukov, K., 2012. Appropriate tree species for intensive forest plantations in central Northern Bulgaria. Forest. Ideas 18, 84–91.

Li, X., Long, Z., She, Z., Shi, F.-H., Shen, Y.-B., 2023. Effects of substrate composition and container size on growth of container seedlings of *Tilia miqueliana*. J. Northeast For. Univ. 51 (6), 46–52. https://doi.org/10.13759/j.cnki.dlxb.2023.06.015 (in Chinese with English summary).

Magherini, R., Nin, S., 1993. Experiments on seed germination of some *Tilia* spp. Acta Hortic. 331, 251–258.

Magherini, R., Nin, S., 1994. Propagation of selected *Tilia* spp. by seed and semihardwood cuttings. Adv. Hortic. Sci. 8, 91–96.

Milev, M., Alexandrov, P., Petkova, K., Iliev, N., 2004. Sowing Materials of Broadleaved Species. Videnov & Son Publ, House, Sofia, Bulgaria (in Bulgarian).

Nikolova, A., Tsvetanova, Z., Koyuva, L., 2021. Guidebook LIFEFORHAB. Forest Seed Control Station, Sofia, Bulgrary (in Bulgarian). https://www.lifeforhab.eu/wp-content/uploads/2021/03/Guidebook-LIFEFORHAB-text.pdf.

Oliet, J.A., Puértolas, J., Valenzuela, P., Vázquez de Castro, A., 2021. Light transmissivity of tree shelters interacts with site environment and species ecophysiology to determine outplanting performance in Mediterranean Climates. Land 10, 753. https://doi.org/10.3390/land10070753.

Pigott, D., 1996. Induction of germination of the naturally non-germinating seeds of *Tilia petiolaris* DC sensu Hooker by treatment with gibberellic acid or kinetin. Arboric. J. 20, 213–220.

Pigott, D., 2012. Lime-trees and Basswoods. A Biological Monograph of the Genus Tilia. Cambridge University Press, Cambridge, UK, 390 pp.

Radoglou, K., Dobrowolska, D., Spyroglou, G., Nicolescu, V.-N., 2009. A review on the ecology and silviculture of limes (*Tilia cordata* Mill., *Tilia platyphyllos* Scop. and *Tilia tomentosa* Moench.) in Europe. Bodenkultur 60 (3), 9–19.

Serafim, C., Ramos, M.A., Yilmaz, T., Sousa, N.R., Yu, K., Van Geel, M., Ceulemans, T., Saudreau, M., Somers, B., Améglio, T., Honnay, O., Castro, P.M.L., 2024. Substrate pH mediates growth promotion and resilience to water stress of *Tilia tomentosa* seedlings after ectomycorrhizal inoculation. BMC Plant Biol. 24, 1001. https://doi.org/10.1186/s12870-024-05614-3.

Xiangliang, Y., Xinxiao, Y., Deshun, Z., Jinbiao, X., 2008. Techniques of quickly breaking seed dormancy of *Tilia tomentosa*. Sci. Silvae Sin. 44 (4), 161–166 (in Chinese with English summary).

Ulmus L. (*Ulmus laevis* Pall., *Ulmus minor* Mill., *Ulmus glabra* Huds.)

Jovana Devetaković
University of Belgrade, Faculty of Forestry, Belgrade, Serbia

Propagation

Seed propagation

Ulmus spp. (elms) produce samaras, winged fruits dispersed by wind and water. Flowering starts in late winter or early spring before leafing, allowing fruit maturation by spring. Samaras can be collected directly from trees or by shaking the branches with poles and collecting from the ground. As wind can easily dislodge samaras, collection is best done in calm, sunny, and dry weather. *Ulmus* produce samaras almost every year, but studies in *U. laevis* populations in central Spain have shown that seed production in a mast year is up to 24 times greater than in non-masting years (Venturas et al., 2014) with a significant proportion of samaras being empty in non-masting years (Perea et al., 2013). Seeds are kept attached to the samaras, which are not dewinged allowing the whole fruit to be used for nursery seeding (Regent, 1980; Stilinović, 1985).

On the Iberian Peninsula, *Ulmus laevis* samara abscission occurs from mid-April to the end of June (Venturas et al., 2014) and this period can be representative of other sites in Europe. The first half of May is an appropriate time for samara collection (Isajev, 1980; Nonić et al., 2012; Devetaković, 2017; Božič et al., 2021). It is recommended to dry samaras briefly (a few days in shade, spread in a thin layer) before spring seeding (Stilinović, 1985). For *U. glabra*, Aldhous (1972) recommended collecting samaras once the green pigment disappeared from the seed wings (May–June) and to sow them immediately, ideally on the same day of collection. To prevent seed displacement, seeds should be watered after sowing and before covering, with additional watering on dry days in the week following sowing.

Germination decreases significantly after 1 year, even when seeds are stored at 3°C with relatively high moisture. For storage of *U. laevis* seeds up to 5 years, Tylkowski (1987) recommended drying samaras to a moisture content of about 10% and storing them in sealed containers at −1 to −3°C, although germination rate declined after this period. Seed characteristics of the three *Ulmus* species are presented in Table 11.72.

Vegetative propagation

Ulmus species can be propagated using various vegetative reproduction methods (Grbić, 1992). *U. laevis* has been successfully propagated by leaf bud and softwood cuttings collected from mid-June to mid-July. Rooting medium can be a 3:1 mixture of peat and sand, combined with intermittent mist. Rooting is enhanced by using Rooton F powder (a mixture of 0.067% naphthalene-acetamide [NAD], 0.033% methyl-1-

Table 11.72 Characteristics of seed lots of *Ulmus* spp.

Species	Purity (%)	No. seeds/kg	Germination capacity (%)
Ulmus laevis	50–60 (55)	82,000–194,000 (138,000)	40–100 (70)
Ulmus minor	60	80,000–160,000 (120,000)	25–96 (60)
Ulmus glabra	40	36,000–125,000 (80,500)	50–91 (76)

Mean values in brackets.
Aldhous (1972); Isajev (1980); Regent (1980); Stilinović (1985); Milev et al. (2004); Çiçek and Tilki (2006); Çiçek and Tilki (2007); Nonić et al. (2012); Perea et al. (2013); Devetaković (2017); Thomas et al. (2018); Božič et al. (2021).

naphthaleneacetic acid [MNAA], 0.013% methyl-1-naphthalene-acetamide [MNAD], 0.057% indole-3-butyric acid [IBA], and 4% tetramethyl-thiuram disulfide [Thiram]) (Grbić, 2003).

U. minor can be successfully propagated using root and stem cuttings (reviewed by Venturas et al., 2013). Stem cuttings, 15–20 cm long, can be collected in winter (December to March) or early summer (mid-June to mid-July). The apical end should be cut beveled just above a bud, while the basal end is cut perpendicularly below a bud. A mixture of perlite and peat (3:1, v:v) is a recommended growing medium with an application of fungicide before establishment and weekly fungicide applications during development in the growing medium. To prevent cutting desiccation, air humidity must be around 80% and air temperature should not exceed 25°C. Optimum temperature of the growing medium is 18–20°C. For improved rooting, hormone treatment is recommended by soaking cuttings for 5 s in an IBA 6000-ppm solution prepared with ethanol 30% for winter cuttings and IBA 1000 ppm prepared with 25% ethanol for summer cuttings.

Propagation success for *U. glabra* cuttings is generally lower than for other *Ulmus* species, but rooting can be improved with 8000 ppm IBA and a perlite-peat-sand medium (Shahraji et al., 2007).

Numerous in vitro propagation methods for *Ulmus* have been developed, primarily to mass-produce trees resistant to the Dutch elm disease. There are successful protocols for the micropropagation of the three *Ulmus species* from mature trees using the axillary bud culture method (Malá et al., 2007). Dormant axillary buds, collected in winter (February), should be rinsed, sterilized in 0.1% $NaClO_3$ for 10 min, and then in 0.01% $HgCl_2$ for 15 min, followed by three rinses in autoclaved distilled water for 15 min each. Finally, apical meristems can be isolated and placed in agar-solidified (6 g/L) Murashige and Skoog (1962) medium supplemented with benzylaminopurine, IBA, glutamine, and sucrose at pH 5.8. Culturing is performed at 24°C for a 16 h photoperiod with white, fluorescent light for 4–6 weeks. Organogenesis is expected within 2 weeks.

For acclimatization, 3–4 cm long microcuttings are rooted in two steps. First, shoots are cultured for 1 week at 25°C in black boxes on one-third strength Murashige y Skoog medium with α-naphthaleneacetic acid. Then, shoots are transferred to hormone-free Murashige y Skoog medium with a 16 h photoperiod (white, fluorescent light, 30 μmol/m^2/s); roots emerge after 1 week. After 3–4 weeks, rooted shoots are transferred to a non-sterile substrate, usually perlite under continuous white, fluorescent light at 25°C and regularly irrigated. Air relative humidity needs to be 90% during the first 15 days. Then, rooted plants can be transferred to containers filled with a peat–soil–perlite mixture (2:1:1, w/w/w) and cultured for 6–8 weeks, including 15 days under controlled conditions in a greenhouse at the beginning of this phase (air temperature 25°C, 70% relative humidity). After acclimatization, the plantlets can be transplanted outdoors between May and June to protected nursery beds where they develop over the growing season and are ready for field planting in autumn or the following spring.

Seedling cultivation

Container seedlings

Nursery cultivation can start in mid-May or early June, immediately after seed collection. For low-quality seed lots, germination can occur in greenhouses using seed trays and recently emerged seedlings are transplanted into containers. Recommended container cell volume is a minimum of 300 mL, with a cultivation density below 300 plants/m^2 for one-year-old seedling production (1 + 0). The growing medium should consist of 3:1 organic (peat or coir) to mineral (perlite or vermiculite) components by volume, with seeds covered by a 1 mm layer of vermiculite. Germination typically starts within 7–14 days. Shading young seedlings for the first few weeks may be necessary in hot, sunny conditions (June). For *U. minor* seedlings, a combination of slow-release and foliar fertilization has shown satisfactory growth (Venturas et al., 2013). Recommended slow-release fertilizer options for 8–9 months include 2 g/L of 18-11-10 NPK or 2.5 g L-1 of 14-8-15 NPK. Alternatively, fertilized peat can be used with an initial 16-8-16 NPK base fertilizer and 20-7-19 NPK maintenance fertilizer, keeping nitrogen levels at 160–190 ppm.

If seedlings need to be cultivated for more than 1 year, they can be transplanted to larger containers (2–2.5 L) for one or two additional years or transplanted to nursery beds (producing 1 + 1 or 1 + 2 plants).

Bareroot seedlings

Ulmus seeds should be sown immediately after collection (during May or in early June) in prepared seedbeds. *U. minor* can be sown across the entire seedbed at 40 g of seed m^2 (Venturas et al., 2013), or in rows spaced 20–25 cm apart with 50 seeds/m.

Sowing rates for *U. laevis* are 600 seeds/m^2 (Devetaković, 2017). The suggested growing density for first-year nursery cultivation is less than 200 seedlings/m^2, or 50

seedlings/m of row (Çiçek et al., 2011). After sowing, seeds need to be covered with a 0.5 cm layer of fine soil or sand (0.5 cm) and watered regularly throughout the 2-week germination period. *U. laevis* seedlings can be successfully produced without additional fertilization if nursery soil is nutrient-rich (Çiçek and Yilmaz, 2006; Devetaković, 2017), but some studies report positive effect of foliar fertilization several times during growing season (Božič et al., 2021).

For *Ulmus* spp. seedlings older than 1 year, transplanting is needed due to the strong, branched root system that develops in the first year. Recommended spacing for *U. laevis* in the second and third year is 0.7 m between rows and 20 cm between plants within rows (Devetaković, 2017) while for *U. minor* 0.5 m between rows and plants is adequate (Venturas et al., 2013). Božić et al. (2021) suggested 0.8 m between rows and 25 cm within rows for both *U minor* and *U. laevis,* allowing adjustments based on mechanization, but not exceeding 50 cm between rows and 20 cm between seedlings within rows.

For seedlings growing 3 years in the nursery (1 + 2), root pruning is necessary due to extensive root growth. Pruning should be done in the spring of the third year using a plow, followed by immediate watering and soil compaction around the plants. Regular irrigation and weeding are essential throughout the nursery period. Morphological characteristics of container and bareroot stock of U. minor and U. laevis seedlings are presented in Table 11.73.

Table 11.73 Morphological and physiological attributes of high quality seedlings of *Ulmus* species.

	Container seedlings		**Bareroot seedlings**	
	Observed values	**Recommended values**	**Observed values**	**Recommended values**
Attributes	***Ulmus minor* 1 + 0**		***Ulmus laevis* 1 + 0**	
Shoot height (cm)	25–50	>30	7–63	>30
Root collar diameter (cm)	0.4–0.6	>0.4	0.09–0.8	>0.4
Shoot dry weight (g)			0.02–7.7	>2
Root dry weight (g)			0.01–7.7	>2
Seedling dry weight (g)			5.4–18.9	>4
Shoot/root dry weight ratio			0.3–4.22	>0.3
	***Ulmus minor* 1 + 1**		***Ulmus laevis* 1 + 1**	
Shoot height (cm)	100–150	>50	16–107	>50
Root collar diameter (cm)	0.8–1.2	>0.8	0.2–1.6	>0.8
			***Ulmus laevis* 1 + 2**	
Shoot height (cm)			54–494	>130
Root collar diameter (cm)			0.9–8.6	>2.0

Çiçek and Yilmaz (2006), Venturas et al. (2013), Devetaković et al. (2013), Devetaković (2017).

Field establishment

Seedlings of different ages can be successfully planted in the field. *U. laevis* 1 + 0 bareroot seedlings have shown survival rates >95% (Çiçek and Yilmaz, 2006), although one-year-old seedlings may have lower survival (50%–60%, unpublished data). Older plants (1 + 1 or 1 + 2) generally achieve higher survival, especially on flooded sites common for *U. laevis* (Devetaković, 2017). *U. minor* and especially *U. laevis* seedlings are somewhat flood tolerance (Li et al., 2015). Our experience indicates that 3-year-old (1 + 2) *U. laevis* seedlings can show survive rates >87% after 3 weeks of flooding in the year after outplanting. In addition, *U. laevis* tolerates various soil types better than *U. minor*, with slightly higher survival in clay-rich soils (70% clay, pH 7.5–7.9; Çiçek et al., 2007).

For 1 + 0 *Ulmus* plants in forest plantations, a root collar diameter >4 mm and shoot height >40 cm are recommended. Taller seedlings (65–75 cm) perform better on sites with high weed competition. One-year-old rooted cuttings *of U. minor* have shown successful survival and growth in the first years after outplanting (Çiçek et al., 2010).



Figure 11.52 A 6-year-old *Ulmus laevis* tree in a plantation with a regular 3 × 3 m spacing. Seedlings were 3 years-old at the time of planting. *(Jovana Devetaković.)*

Herbivores such as rabbits and roe deer are the main environmental limitation for seedling establishment; therefore, using tree shelters or fences is advised.

Planting can be performed in late autumn or early spring when vegetation is dormant. For two- or three-year-old seedlings, planting holes should be at least 30 cm wide and 50 cm deep. Tillage is recommended on abandoned cropland, while holes can be used for planting spots on sites where weeds are not vigorous (Devetaković et al., 2015). Fertilizing within the planting hole and irrigation during the first two growing seasons enhances *U. minor* establishment, especially on dry sites (Venturas et al., 2013). If necessary, branch pruning is recommended 2 or 3 years after outplanting to promote a well-formed crown.

Planting density varies depending on the planting aim. *Ulmus* species are mainly planted for ecological restoration and species conservation. In these cases, planting at 6×6 m spacing (278 seedlings/ha) would promote growth and early flowering. A higher density can accommodate seedling mortality, although thinning may be necessary if survival rates are high (Fig. 11.52). For restoration and protection objectives, irregular planting patterns that create a more natural spatial distribution are preferable to traditional regular grids (Venturas et al., 2013).

Chapter References

Aldhous, J.R., 1972. Nursery Practice. Forestry Commission, London, Bull, p. 43.

Božič, G., Westergren, M., Heino, K., Lanšćak, M., Nagy, L., Novčić, Z., Rukavina, S., Schuler, S., Stojni', S., Železnik, P., 2021. Guidelines for the Production of Planting Material for Restoration of Riparian Forests. Slovenian Forestry Institute, Silva Slovenica Publishing Centre.

Çiçek, E., Tilki, F., 2006. Effects of temperature, light and storage on seed germination of *Ulmus glabra* Huds. and U. *laevis* Pall. Pakistan J. Biol. Sci. 9 (4), 697–699.

Çiçek, E., Tilki, F., 2007. Seed germination of three *Ulmus* species from Turkey as influenced by temperature and light. J. Environ. Biol. 28 (2), 423–425.

Çiçek, E., Yilmaz, M., 2006. Effect of seedbed density on morphological characteristics and field performance of *Ulmus laevis* seedlings. J. Balkan Ecol. 9 (2), 167–173.

Çiçek, E., Tilki, F., Özbayram, A.K., Çetin, B., 2010. Three-year growth comparison between rooted cuttings and seedlings of *Fraxinus angustifolia* and *Ulmus laevis*. J. Appl. Sci. Res. 6 (3), 199–204.

Çiçek, E., Tilki, F., Kulaç, Ş., Yilmaz, M., Yilmaz, F., 2007. Survival and growth of three hardwood species (*Fraxinus angustifolia, Ulmus laevis and U. minor*) on a bottomland site with heavy clay soil. J. Plant Sci. 2 (2), 233–237.

Çiçek, E., Çiçek, N., Tilki, F., 2011. Four-year field performance of *Fraxinus angustifolia* Vahl. and *Ulmus laevis* Pall. seedlings grown at different nursery seedbed densities. Res. J. Forest. 5 (2), 89–98.

Devetaković, J., 2017. Genetički Potencijal Veza (*Ulmus laevis* Pall.) Za Proizvodnju Namenskog Sadnog Materijala. Beograd, Šumarski Fakultet (Doctoral Dissertation, 1–244).

Devetaković, J., Mitrović, B., Milosavljević, M., Nonić, M., Stanković, D., 2015. European white elm: potential for wetlands reforestation. In: International Conference Reforestation Challenges, 03-06 June 2015, Belgrade, Serbia, pp. 144–148.

Devetaković, J., Šiljačić-Nikolić, M., Ivetić, V., 2013. Variability of morphometric characteristics of one-year seedlings of different half-sib European white elm (*Ulmus effusa* wild.) from the great war island. Biologica Nyssana 4 (1–2), 87–92.

Grbić, M., 1992. Unapređenje Rasadničke Proizvodnje Nekih Brestova (*Ulmus* L.) Autovegetativnim Metodama Razmnožavanja. Doctoral dissertation. University of Belgrade - Faculty of Forestry).

Grbić, M., 2003. Uporedna analiza oživljavanja jednonodusnih i standardnih zelenih reznica brestova. Bullet. Facult. Forestry-Belgrade 88, 55–64.

Isajev, V., 1980. Results of the five-year study of some Russian white elm (*Ulmus laevis* Pallas) seed properties. Bullet. Facul. Forest. Belg. 54, 101–110.

Li, M., López, R., Venturas, M., Pilar, P., Guillermo, G., Luis, G., Rodríguez-Calcerrada, J., 2015. Greater resistance to flooding of seedlings of *Ulmus laevis* than *Ulmus minor* is related to the maintenance of a more positive carbon balance. Trees 29, 835–848. https://doi.org/10.1007/s00468-015-1163-x.

Malá, J., Cvikrová, M., Chalupa, V., 2007. Micropropagation of mature trees of *Ulmus glabra, Ulmus minor* and *Ulmus laevis*. In: Jain, S.M., Häggman, H. (Eds.), Protocols for Micropropagation of Woody Trees and Fruits. Springer, Dordrecht. https://doi.org/10.1007/978-1-4020-6352-7_22.

Milev, M., Aleksandrov, P., Petkova, K., Iliev, N., 2004. Posevni Materiali Ot Širokolisni Vidove Sofia, pp. 1–262.

Nonić, M., Devetaković, J., Šijačić-Nikolić, M., Milovanović, J., 2012. Yield variability as a basis for conservation and directed utilization of European White Elm (*Ulmus e*ffusa Willd.) gene pool at Great War Island. Agric. For. 58 (3), 105–113.

Perea, R., Venturas, M., Gil, L., 2013. Empty seeds are not always bad: simultaneous effect of seed emptiness and masting on animal seed predation. PLoS One 8 (6), e65573. https://doi.org/10.1371/annotation/4690ecf8-cf07-4e28-9d81-855f8113fef7.

Regent, B., 1980. Šumsko sjemenarstvo. Jugoslovenski poljoprivredno šumarski centar. In: Služba Šumske Proizvodnje, pp. 1–201.

Shahraji, T.R., Farokhnia, E., Hatamzadeh, A., 2007. Effect of IBA on the rooting ability of hardwood and semi-hardwood cuttings of *Ulmus glabra* (Huds.). Indian J. For. 30 (2), 229–232. https://doi.org/10.54207/bsmps1000-2007-A02153.

Stilinović, S., 1985. Semenarstvo šumskog i ukrasnog drveća i žbunja. Institut za šumarstvo Šumarskog fakulteta 1–399.

Thomas, P.A., Stone, D., La Porta, N., 2018. Biological flora of the British Isles: *Ulmus glabra*. J. Ecol. 106 (4), 1724–1766. https://doi.org/10.1111/1365-2745.12994.

Tylkowski, T., 1987. Storing of Russian elm (*Ulmus laevis* Pall.) seed over many years. Arbor. Kornickie 32, 297–305.

Venturas, M.D., Lopez Rodriguez, R.A., Perea García-Calvo, R., Fernandez Fernandez, V., Guzman Delgado, P., Rodriguez Calcerrada, J., Miranda, E., Domínguez, J., González-Gordaliza, G., Zafra, E., Fajardo-Alcántara, M., Martín, J.A., Ennos, R., Nanos, N., Lucena, J.J., Iglesias, S., Collada, C., Gil, L., 2014. *Ulmus laevis* in the Iberian Peninsula: a review of its ecology and conservation. iFor. Biogeosci. For. 8, 1–8.

Venturas, M., Iglesias Sauce, S., Nicolás Peragón, J.L., Martín García, J.A., Gil Sánchez, L., 2013. *Ulmus minor* mill. In: Pemán García, J., Navarro-Cerrillo, R.M., Nicolás Peragón, J.L., Prada Sáez, M.A., Serrada Hierro, R. (Eds.), Producción y Manejo de Semillas y Plantas Forestales, Organismo Autónomo Parques Nacionales. Serie Forestal, Madrid, Spain, vol II, pp. 575–597.