Verbenaceae—Verbena family

Callicarpa americana L.

American beautyberry

Franklin T. Bonner

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Other common names. French-mulberry, Spanish-mulberry, sour-bush, sow-berry.

Growth habit, occurrence, and uses. American beautyberry—*Callicarpa americana* L.—is a small, woody shrub of the pine forests in the southern coastal plain. It seldom grows taller than 2 or 3 m. The shrub is common underneath the pine overstory and along roads and forest edges, where it grows best. It is found from Virginia to Florida and west to Texas and Oklahoma; it also occurs in the West Indies (Vines 1960). American beautyberry is an important food plant for wildlife, especially birds and eastern white-tailed deer *(Odocoileus virginianus)* (Blair and Epps 1969; Grelen and Duvall 1966; Halls 1973). The shrub's well-branched root system and drought resistance make it desired for erosion control in some areas (Brown 1945), and it is frequently grown as an ornamental because of the colorful fruits (Dirr and Heuser 1987).

Flowering and fruiting. The small, inconspicuous flowers are borne in axillary, dichotomous cymes about 8 to 36 mm long. Flowering starts in early June and may continue into the fall months, even as the fruits mature in August to November (Dirr and Heuser 1987; Vines 1960). The fruit is a berrylike, globose drupe, about 3 to 6 mm in diameter, that is borne in conspicuous axillary clusters on the current season's growth. The rose to purple, or sometimes white (Brown 1945), fruit color gives this plant its ornamental value. A single fruit cluster may contain as many as 300 fruits, although about 100 is typical. Each fruit usually contains 4 small flattened seeds that are light brown in color and about 1 to 1.5 mm in length (Grelen and Duvall 1966; Vines 1960) (figures 1 and 2). Plants begin to bear fruit as early as 2 years of age, and mature plants may yield over $^1/_2$ kg (about 1 lb) annually (Halls 1973).

Collection of fruits; extraction and storage of seeds. Fruits can be easily collected by hand in autumn, when their rose to purple color indicates maturity. The soft fruits quickly disintegrate when they are macerated with water. Filled

Figure 1—*Callicarpa americana*, American beautyberry: seeds.

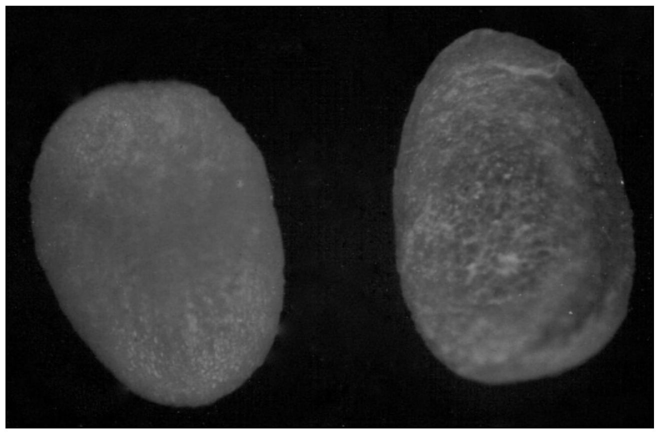
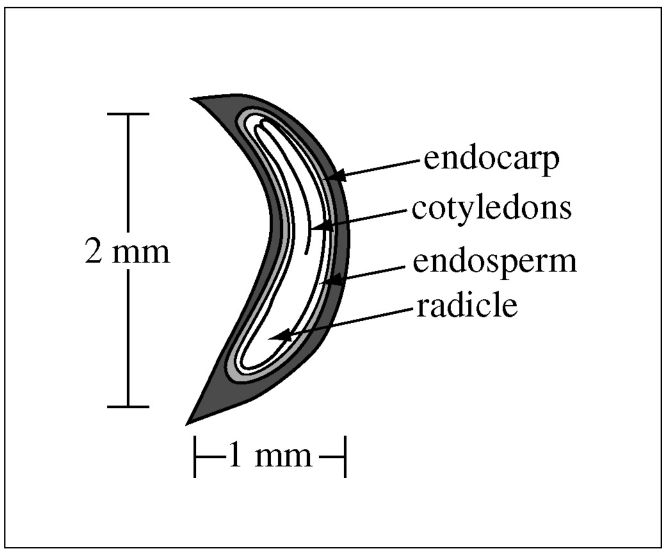


Figure 2—*Callicarpa americana*, American beautyberry: longitudinal section through a seed.



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seeds sink in water, and the pulp can be floated off. Any type of macerator should work, even laboratory or kitchen blenders for small lots. There are about 600 seeds/g (17,000/oz), and good cleaning should yield a purity of practically 100%. There are no known storage data for this species, but soil seed bank studies show that the seeds will survive for at least 1 year buried in the soil. This fact, plus the hard seedcoat, suggest that these seeds are orthodox in storage behavior. Long-term storage at temperatures near or below freezing should be successful with seeds that are dried to below 10% moisture content.

Pregermination treatments and germination tests. The seeds have a hard seedcoat, and germination is relatively slow. One sample stratified for 30 days yielded only 22% germination in 90 days when tested at an alternating temperature of 20 °C at night and 30 °C in the light. Untreated seeds sown in the fall, however, were reported to give excellent germination in the spring (Dirr and Heuser 1987). There are no official test prescriptions for American beautyberry.

Nursery practice. No details of nursery practices for American beautyberry are available, except the successful fall-sowing mentioned above. The small seed size suggests that soil or mulch covers after sowing must be very light. Vegetative propagation is not difficult with this species. Softwood cuttings taken anytime from June to September root well if treated with IBA (1,000 ppm) and placed in a mist bed (Dirr and Heuser 1987).

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Cupressaceae—Cypress family

Calocedrus decurrens (Torr.) Florin

incense-cedar

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Synonyms. *Libocedrus decurrens* Torr., *Heyderia decurrens* (Torr.) K. Koch.

Other common names. California incense-cedar, pencil cedar, pecky cedar.

Growth habit, occurrence, and uses. Incense-cedar was once classified as the only species in the genus *Libocedrus* native to the United States (Harlow and others 1979; Little 1979), but recent taxonomic changes have included it as 1 of 3 species in the genus *Calocedrus* Kurz. Regional genetic variation within incense-cedar is small, but 12-year growth of trees from southern California was less than that of trees from more northerly regions (Rogers and others 1994). Recognized cultivars under the former classification include *L. decurrens* cv. *aureovariegata* Beissner, *L. decurrens* cv. *columnaris* Beissner, *L. decurrens* cv. *compacta* Beissner, and *L. decurrens* cv. *glauca* Beissner (Harrison and Dallimore 1966; Rehder 1940).

Mature trees of this evergreen conifer vary in height from 15 to 46 m and from 0.3 to 2.13 m in diameter (Jepson 1910; Sargent 1961; Sudworth 1908). A maximum circumference of 12.9 m (van Pelt 2001) and a maximum height of 68.6 m have been reported (Stein 1974). Young trees generally have dense pyramidal to columnar crowns; older trees are characterized by more open, irregular crowns; rapidly tapering trunks with buttressed bases; and deeply furrowed and ridged bark.

The range of incense-cedar spans about 15 degrees of latitude, from the southeastern slopes of Mount Hood in Oregon southward within and adjacent to the Cascade, Siskiyou, coastal, and Sierra Nevada ranges to the Sierra de San Pedro Martír in northwestern Mexico (Griffin and Critchfield 1976; Sudworth 1908). It extends eastward from the coastal fog belt to arid inland parts of central Oregon, northern California, and westernmost Nevada. In elevation, incense-cedar is found from 50 to 2,010 m in the north and from 910 to 2,960 m in the south (Peattie 1953; Powers and Oliver 1990; Sudworth 1908). Incense-cedar grows on many kinds of soil and is one of the most prominent conifers on serpentine soils. Typically, it is a component of mixed conifer forest and may make up as much as 50% of the total stand (Powers and Oliver 1990).

Trees are harvested primarily for lumber and for round or split wood products. The wood is variable in color, durable, light, moderately soft, uniformly textured, easy to split and whittle, and finishes well. Incense-cedar is also used as a pulp additive and for making a variety of specialty items, the best known being the wooden pencil (Betts 1955; Panshin and others 1964). Boughs, particularly those bearing staminate cones, are harvested commercially for decorations (Schlosser and others 1991), and young trees are a minor component of the Christmas tree trade.

First cultivated in 1853, ornamental specimens with shapely crowns have grown well in many places outside of their native range in the Pacific Northwest—in New England and in the mid-Atlantic region of the United States and western, central, and southern Europe (Edlin 1968; Harrison and Dallimore 1966; Jelaska and Libby 1987; Sargent 1961). Within its native range, incense-cedar is commonly planted for highway landscaping, screenings, and home-site improvement.

Young incense-cedars are sometimes browsed extensively (Stark 1965), but in general, the species rates low to moderate in value as wildlife browse (Longhurst and others 1952; Sampson and Jespersen 1963; Van Dersal 1938). Its seeds are eaten by small mammals (Martin and others 1951) but are not a preferred food of chipmunks (Tevis 1953). Dense understory incense-cedars provide an important source of cover and food for overwintering birds in the western Sierra Nevada (Morrison and others 1989).

Flowering and fruiting. Yellowish green staminate flowers develop terminally on twigs as early as September even before the current year's cones on the same twigs have opened (Stein 1974). These flowers, 5 to 7 mm long, are prominently present "...tingeing the tree with gold during

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the winter and early spring..." (Sargent 1961). The inconspicuous pale yellow ovulate flowers also develop singly at tips of twigs. Flowering has been reported to occur as early as December and as late as May (Britton 1908; Hitchcock and others 1969; Mitchell 1918; Peattie 1953; Sargent 1961; Sudworth 1908), but it is not clear how well observers distinguished between flower appearance and actual pollen dissemination. Unopened staminate flowers and open or nearly open ovulate flowers were present on branches collected in the first week of April west of Klamath Falls, Oregon (Stein 1974).

Individual cones (figure 1), each containing up to 4 seeds, are scattered throughout the crown, and mature in a single growing season. As they ripen, their color changes from a medium green to a yellowish green or yellow tinged with various amounts and shades of brown. During opening, the cone becomes reddish brown and acquires a purplish cast. Insect-attacked cones are among the first to change color. Generally, cones of many color shades are found on a tree as opening commences.

Seed dispersal may extend over a lengthy period, from late August through November or later (Fowells and Schubert 1956; McDonald 1992; Mitchell 1918; Powers and Oliver 1990; Sudworth 1908). For example, in 1937 and 1940, respectively, 11 and 32% of the seed had fallen by early October at 1 or 2 California locations, yet 47 and 66% of the total fell after November 11 (Fowells and Schubert 1956). Cutting tests have shown that 14 to 65% of the naturally dispersed seeds appear sound, with higher values coincident with heavy crops (Fowells and Schubert 1956).

The oft-repeated generalization that incense-cedars bear some seeds every year and abundant crops frequently (Betts 1955; Mitchell 1918; Sudworth 1908; Van Dersal 1938) has not been confirmed by systematic observations made in 3 locations. During a 35-year period on the Stanislaus National Forest in California, incense-cedars bore a heavy or very heavy crop in 7 of those years, a medium crop in 11 years, and a light crop in 17 years (Schubert and Adams 1971). On the Challenge Experimental Forest in central northern California, there were 1 medium to heavy and 9 light to very light crops in 24 years (McDonald 1992). During 15 years on the South Umpqua Experimental Forest in southwest Oregon, there were 2 abundant crops, 1 medium crop, and 12 years with light or no crops (Stein 1974). Generalized statewide reports for California and Oregon show that incense-cedar cone crops are often light and that there is wide geographic variability in crop abundance (Schubert and Adams 1971). During years when crops are reported as light or a failure, scattered cones, even an occasional heavily loaded tree, may be found somewhere.

Figure 1—*Calocedrus decurrens*, incense-cedar: cones hang singly from branch tips well-dispersed over the crown and contain up to 4 seeds each.

Flowers and young cones may be damaged or killed occasionally by adverse climatic factors, and squirrels cut some mature cones (Fowells and Schubert 1956). Losses are also caused by sawflies (*Augomonoctenus libocedrii* Rohw.), juniper scale (*Carulaspis juniperi* Bouche), and leaf-footed bugs (*Leptoglossus occidentalis* Heidemann) that feed on developing cones and seeds (Furniss and Carolin 1977; Koerber 1963).

Collection of cones. Cones are generally hand-picked from standing or felled trees. Stripping cones or using a cone rake will expedite collection because cones hang dispersed over the crown. The ideal time for collection is the short period when cleavages appear between the scales of many cones on a tree. If large quantities of seeds are needed, both collecting them from plastic sheets spread beneath or enclosing the tree and vacuum-harvesting seeds from the ground merit consideration. Dispersed seeds should be collected promptly to minimize heat damage. To facilitate later seed cleaning, foliage intermixed with cones or seeds should be removed during collection or shortly afterward, before it dries and crumbles.

Cones are normally handled and transported in partly filled open-mesh sacks that facilitate cone expansion and air exchange. Good aeration should be provided around each sack to keep the cones from overheating during storage.

Extraction and storage of seeds. To maintain high seed viability, cones should not be exposed to high temperatures. Under warm, dry conditions, cones will air-dry outdoors or indoors in 3 to 7 days if layered thinly in trays or on sheeting or tarps. Turning or stirring layered cones will facilitate drying and opening. They may also be kiln-dried at 27 °C or lower (Lippitt 1995).

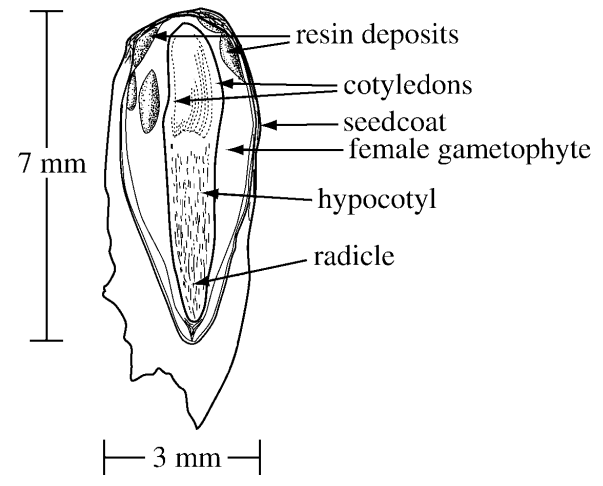
Seeds separate readily from well-opened cones; moderate tumbling or shaking is helpful. Whether done by improvised methods or in commercial machines, tumbling or shaking should be done gently, preferably at less than 27 °C, because seedcoats of incense-cedar are thin and easily broken.

The winged seeds are about 2.5 cm long and nearly one-third as wide (figure 2). Although appearing to have only 1 wing, each seed actually has 2 wings—a long, wide wing extending lengthwise beyond the seed on one side and a narrow, much shorter wing barely merging alongside the first from the opposite side. The wings are persistent and project past the narrow radicle end of the seed rather than from the cotyledon end as in many other conifers (figure 3).

The persistent wings should be left intact. When seeds are run through mechanical de-wingers, the narrow radicle ends may break off along with the wings. This type of damage was the probable cause of the very low viability observed in some lots of de-winged seeds. Damaging effects should be evaluated before using any proposed hand or mechanical de-winging technique.

Figure 2—*Calocedrus decurrens*, incense-cedar: each seed has 2 wings, a long, wide wing on one side (**right**) and a narrow, much shorter one on the other side (**left**).

Figure 3—*Calocedrus decurrens*, incense-cedar: longitudinal section showing the radicle located at the narrow end of the seed.



Small particles of debris can be removed from among winged seeds by screening. Sensitive adjustment of an air stream or gravity separator will permit further cleaning and adequate separation of empty from full seeds with wings intact. Purities of 85 to 98% or more have been obtained (Lanquist 1946; Lippitt 1995; Rafn 1915; Toumey and Korstian 1942).

Thirty-five liters (1 bu) of cones weigh 18 to 23 kg (40 to 50 lbs) and yield from 0.45 to 1.36 kg (1 to 3 lb) of seeds (CDF 1969; Tillotson 1925; Toumey and Korstian 1942). A minimum of 14,110 and a maximum of 63,930 seeds/kg (6,400 and 29,000 seeds/lb) were found among 55 samples from northern California weighed by Show in 1918. More recent collections indicate that seeds per weight values differ by seed zone (Lippitt 1995):

	Average		Range		
Region & seed zone series no.	**/kg**	**/lb**	**/kg**	**/lb**	**Samples**
Siskiyou Mtns. & inland north coastal range (SZ #300)	27,270	12,368	24,820–29,960	11,260–13,588	41
Sierra Nevada (SZ #500)	31,820	14,433	21,540–45,330	9,768–20,562	36
Southern California & Central Valley (SZ #900)	33,420	15,160	24,120–38,760	10,940–17,583	5

Reported averages representing collections made largely in northern and central parts of the species' range vary from 27,270 to 44,450 seeds/kg (12,368 to 20,160 seeds/lb) (CDF 1969; Lanquist 1946; Lippitt 1995; Mitchell 1918; Rafn 1915; Show 1918; Stein 1963; Sudworth 1900; Tillotson 1925; Toumey and Korstian 1942). The smaller averages are probably the most realistic, for samples weighed by several investigators contained only 60 to 67% full seeds either winged or wingless (Lanquist 1946; Show 1918).

Incense-cedar seeds do not keep well in dry storage at room temperature (Shaw 1918), but high viability can be maintained for several years in cool storage. In limited tests, 2 seedlots retained 98% and 74% viability after storage in closed metal containers at 5 °C for 2 and 3 years, respectively, but lost all viability after 8 years (Schubert 1954). It is now common practice to store incense-cedar seeds dried to low moisture content near –18 °C in cloth or plastic bags or in plastic-lined fiberboard containers. Mature, undamaged seedlots have retained viability in cold storage for 10 years at 5 to 9% moisture content (Lippitt 1995); maximum duration before such lots begin losing viability has not been determined.

Pregermination treatments and germination tests. Standard procedures prescribed by the Association of Official Seed Analysts (1999) for testing incense-cedar seeds include chilling them for 30 days at 2 to 5 °C before germination. Comparison tests showed that prechilling markedly improved total germination and rate of germination of some but not all lots (Stein 1974). Short of making a paired test, there is no way to identify which lots benefit from prechilling and which ones do not. To prepare them for prechilling, seed samples are either (1) placed on a moist substratum in a closed dish; (2) placed in a loosely woven bag or screen surrounded by moist peat, sand, or vermiculite; or (3) allowed to soak for 24 hours in tap water at room temperature, drained, and then placed in a glass or plastic container.

Following prechilling, germination of incense-cedar is determined by subjecting seeds for 28 days to alternating temperatures—16 hours at 20 °C and 8 hours at 30 °C with 750 to 1250 lux (75 to 125 foot-candles) exposure to cool-white fluorescent illumination at least during the high-temperature period (AOSA 1999). Tests should be carried out on cellulose paper wadding or blotters in closed germination boxes. Germination for 85 lots now in storage at one nursery has averaged 72% following 8 weeks of naked stratification (Lippitt 1995).

The viability of incense-cedar seeds can also be determined by a tetrazolium test (AOSA 2000). The preparation sequence involves removal of wings from dry seeds followed by soaking in water at room temperature for 6 to 18 hours (overnight). Shallow longitudinal cuts are then made on both ends of the seed to expose the embryo. Cut seeds are immersed in a 1% tetrazolium solution and kept in darkness for 6 to 18 hours at 30 to 35 °C. Seeds having a completely stained embryo and a completely stained endosperm are considered viable. Viability determined by the tetrazolium test reveals the seeds' maximum potential and generally is somewhat higher than indicated by a germination test.

Nursery practice and seedling development. Soil fumigation of outdoor beds to combat damping-off and other diseases may or may not be necessary before sowing incense-cedar seeds. Maintenance or replacement of endomycorrhizal fungi is of concern if beds are fumigated. Spring-sowing is now most common even though fall-sown seeds germinated earlier and more uniformly than those sown in the spring and resulting seedlings grew larger in the first season if they escaped damage by late spring frosts (Show 1930). An intermediate approach is to prepare seedbeds in the fall to facilitate early sowing in February or March. Before sowing, seeds are usually stratified naked or in a moist medium at 2 to 5 °C for 30 to 60 days (Lippitt 1995). Well-timed spring-sowings of unstratified seeds have produced satisfactory crops (Show 1930; Stein 1974), but results are less certain. Some spring-sown seeds may hold over to produce seedlings the following spring (Show 1930).

The winged seeds are usually hand-sown in rows. They should be covered about 6 to 12 mm ($^1/_4$ to $^1/_2$ in) deep (Show 1930). Burlap mulch proved satisfactory to keep seedbeds moist (Show 1930); sawdust or other mulch material and frequent sprinkler irrigation are currently used.

Incense-cedar can readily be grown in containers to plantable size in one season. Containers about 15 cm (6 in) deep with a volume of 165 to 215 cm^3 (10 to 13 in^3) are recommended. The seedlings may be started about February in a greenhouse and moved outdoors after 4 to 8 weeks or they may be germinated and grown entirely outdoors.

Germination is epigeal and the radical emerges from the narrow winged end of the seed (figure 4). Young seeds usually have 2, rarely 3, cotyledons (Harlow and others 1979). Leaves about 1.2 cm (0.5 in) long develop along the epicotyl (figure 5). On the first branches, awl-shaped transitional leaves grade into the normal scalelike leaves (Jepson 1910). Seedlings grow 5 to 20 cm (2 to 8 in) tall in the first season and develop a well-branched root system. Young seedlings are fairly resistant to frost and drought (Fowells and Stark 1965; Pharis 1966; Stone 1957). They are preferentially attacked by cutworms, however, and need protection from damping-off (Fowells 1940; Fowells and Stark 1965; Show

Figure 4—*Calocedrus decurrens*, incense-cedar: germinating seed with radicle and hypocotyl emerging from the winged end.

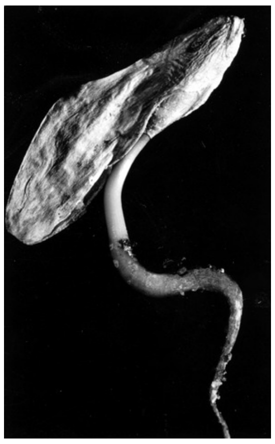
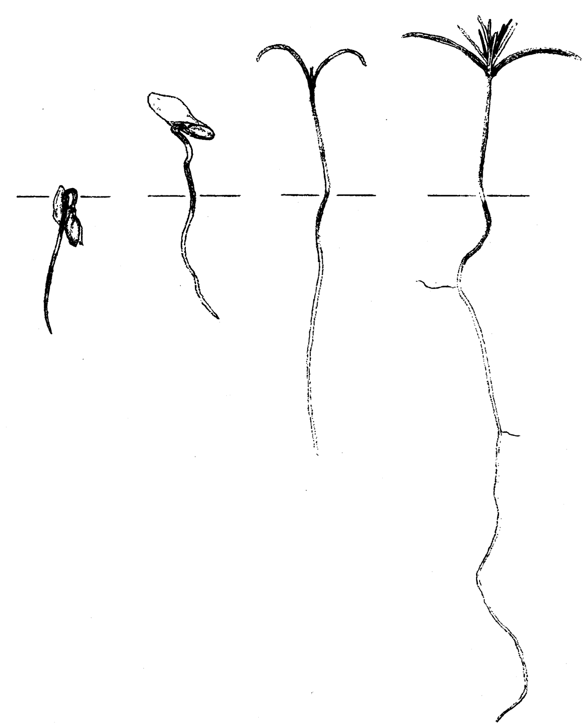


Figure 5—*Calocedrus decurrens*, incense-cedar: seedling development 4, 7, 10, and 17 days after germination.



1930; Stein 1963). In the north-central Sierra of California, they grew about as well unshaded as with one-fourth shade (Show 1930). In current practice, both bareroot and container seedlings are grown without shade. They should be watered regularly but not to excess. Beds may be weeded entirely by hand or with mechanical and chemical assistance.

Seedbed densities of 270 to 325 seedlings/m^2 (25 to 30/ft^2) are satisfactory for producing 1+0 stock. Densities of 160 to 215 seedlings/m^2 (15 to 20/ft^2) are used for 2+0 stock. Tree percents range from 20 to 75 (Show 1930; Stein 1974). Generally, 2+0 bareroot seedling stock is used for outplanting, but 1+0, 1+1, 2+1, and 1+2 transplants have also been used. Some of the target sizes now used for producing stock include 1+0 (stem caliper 3 mm and top length 13 cm), 2+0 (stem caliper 3.5 cm and top length 20 cm), and 1+1 (stem caliper 4 mm and top length 25 cm). Outplanting in the spring proved best in long-ago tests (Show 1930) and continues to be favored.

Incense-cedar also can be reproduced from cuttings started in November (Nicholson 1984), and responds better than most conifers to cell and tissue culture (Jelaska and Libby 1987).

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Bignoniaceae—Trumpet-creeper family

Campsis radicans (L.) Seem. ex Bureau

common trumpet-creeper

Franklin T. Bonner

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Synonyms. *Bignonia radicans* L., *Tecoma radicans* (L.) Juss.

Other common names. trumpetvine, cowitch vine, trumpet-flower.

Growth habit, occurrence, and uses. Common trumpet-creeper—*Campsis radicans* (L.) Seem. ex Bureau, a deciduous vine—is native from Texas to Florida and north to Missouri, Pennsylvania, and New Jersey (Vines 1960). It has also been introduced into New England (Bonner 1974). The vine is sometimes used in erosion control and as an ornamental, but its greatest value is for wildlife food. Hummingbirds are common visitors to trumpet-creeper flowers.

Flowering and fruiting. The large, orange-to-scarlet, perfect flowers are 5 to 9 cm long and appear from May through September (Bonner 1974; Vines 1960). This species is largely self-sterile, but pollinates well when self and cross pollen are mixed (Bertin and Sullivan 1988). The fruit is a 2-celled, flattened capsule about 5 to 15 cm long (figure 1) that matures from September to November (Vines 1960). The capsules turn from green to gray brown as they mature, and the small, flat, winged seeds (figures 1 and 2) are dispersed chiefly by wind as the mature capsules split open on the vine from October through December (Bonner 1974; Vines 1960). Good seed crops are borne annually.

Collection and extraction. Ripe capsules should be gathered when they turn grayish brown in the fall before splitting open. Seeds can be extracted by hand-flailing. There are approximately 300,000 cleaned seeds/kg (136,000/lb) (Bertin 1990; Bonner 1974). One sample had a purity of 98%, with 52% sound seeds (Bonner 1974). The longevity of common trumpet-creeper seeds in storage is not known, but if the seeds are dried to about 10% moisture content, they should store as well as other orthodox seeds.

Germination and nursery practice. The seeds exhibit some embryo dormancy. Pretreatment is not necessary for germination, but cold, moist stratification for 60 days at 5 to

Figure 1—*Campsis radicans*, common trumpet-creeper: fruit (**top**) and seed (**bottom**).

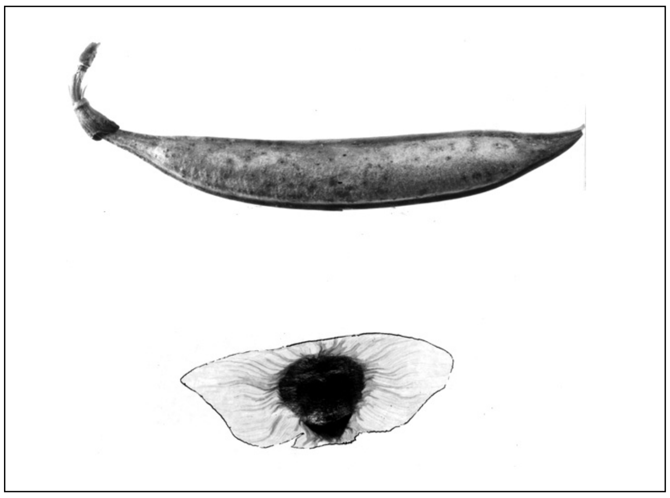
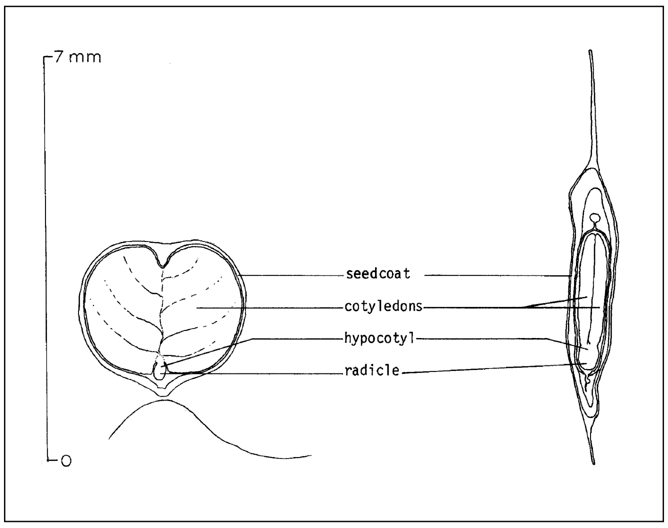


Figure 2—*Campsis radicans*, common trumpet-creeper: longitudinal section through a seed.



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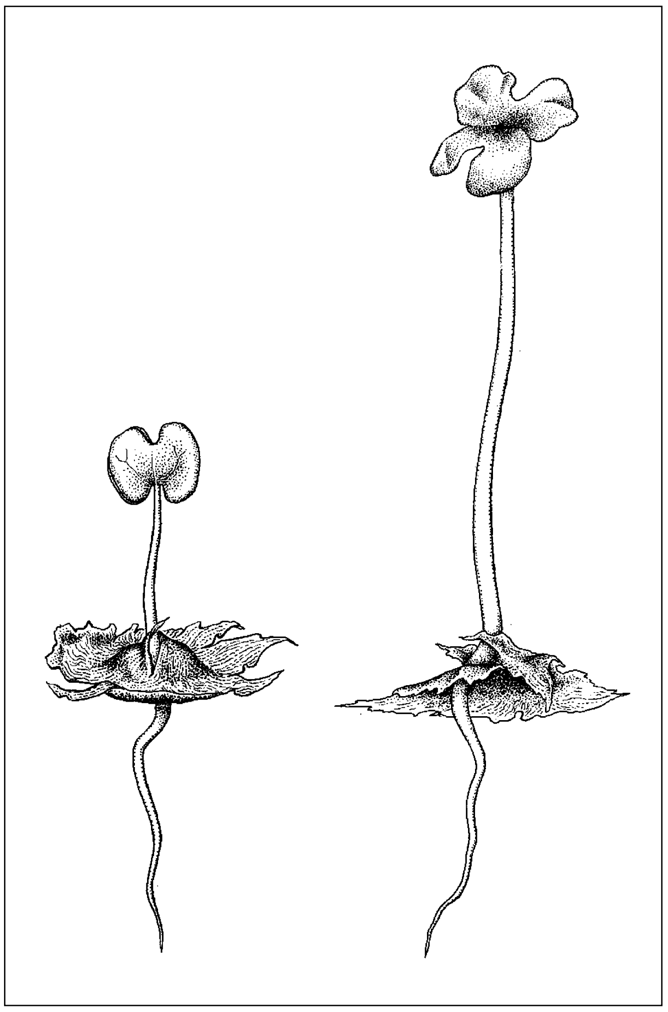
10 °C is recommended for quick and uniform germination (Bonner 1974; Dirr and Heuser 1987). Germination tests in sand have been run for 30 days at 20 °C night and 30 °C day temperatures. Four tests with stratified seeds averaged 66% germination, and germination rate was 51% in 19 days (Vines 1960). Germination is epigeal (figure 3). Seedlings can be grown in nurserybeds from either untreated seeds sown in the fall or from stratified seeds sown in the spring. Some horticultural cultivars are propagated by stem and root cuttings and layering. Softwood cuttings taken in June to September are easily rooted without hormone treatments (Dirr and Heuser 1987).

Figure 3—*Campsis radicans*, common trumpet-creeper: seedling development at 1 and 9 days after germination.



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Fabaceae—Pea family

Caragana arborescens Lam.

Siberian peashrub

Donald R. Dietz, Paul E. Slabaugh, and Franklin T. Bonner

Drs. Dietz and Slabaugh (deceased) retired from the USDA Forest Service's Rocky Mountain Forest and Range Experiment Station; Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station, Mississippi State, Mississippi

Synonym. *Caragana caragana* Karst.

Other common names. caragana, pea-tree.

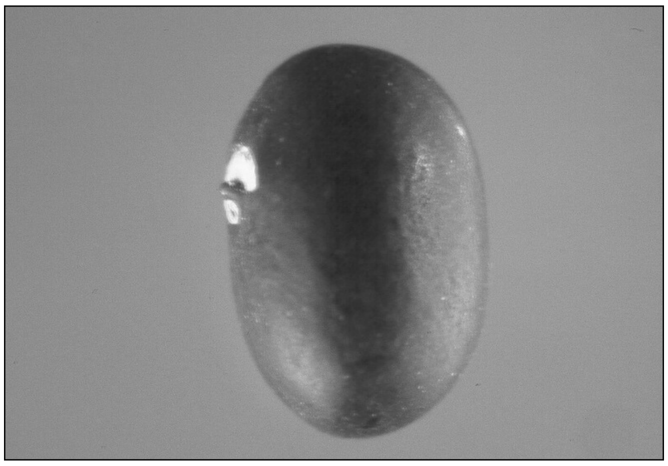
Growth habit, occurrence, and use. Siberian peashrub—*Caragana arborescens* Lam.—is one of the most hardy small deciduous trees or shrubs planted on the northern Great Plains (George 1953; Rehder 1940). Introduced into North America in 1752 (Rehder 1940), Siberian peashrub is native to Siberia and Manchuria and occurs from southern Russia to China *(Graham 1941)*. Varieties include the dwarf (*C. a. nana* Jaeg.) and Lorberg (*C. a. pendula* Carr.) peashrubs (Kelsey and Dayton 1942). The species readily adapts to sandy, alkaline soil and open, unshaded sites on the northern Great Plains, where it grows to heights of 7 m. It has been planted extensively for shrub buffer strips and windbreaks on farmlands and for hedges and outdoor screening in many towns and cities of the upper mid-West (Dietz and Slabaugh 1974; George 1953). It was also planted for wildlife and erosion control in the Great Lakes region (Graham 1941) and for deer-range revegetation programs in the Black Hills of South Dakota (Dietz and Slabaugh 1974). It is now considered invasive.

Flowering and fruiting. The yellow bisexual flowers appear from April to June. The fruit is a legume (pod) that measures 2.5 to 5 cm (figure 1) and contains about 6 reddish-brown, oblong to spherical seeds 2.5 to 4.0 mm in diameter (Lindquist and Cram 1967; Ross 1931) (figures 2 and 3). Fruits change in color to amber or brown as they ripen from June to July (Rehder 1940). Seed dispersal is usually completed by mid-August in most areas on the Great Plains. Shrubs take about 3 to 5 years to reach commercial seed-bearing age, and good crops occur nearly every year (Dietz and Slabaugh 1974).

Collection of fruits. The optimum seed collection period for Siberian peashrub is less than 2 weeks—usually in July or early August. Because the fruits begin to split open and disperse the seeds as soon as they are ripe, the legumes should be gathered from the shrubs by hand as soon as the first ones begin to open (Dietz and Slabaugh 1974).

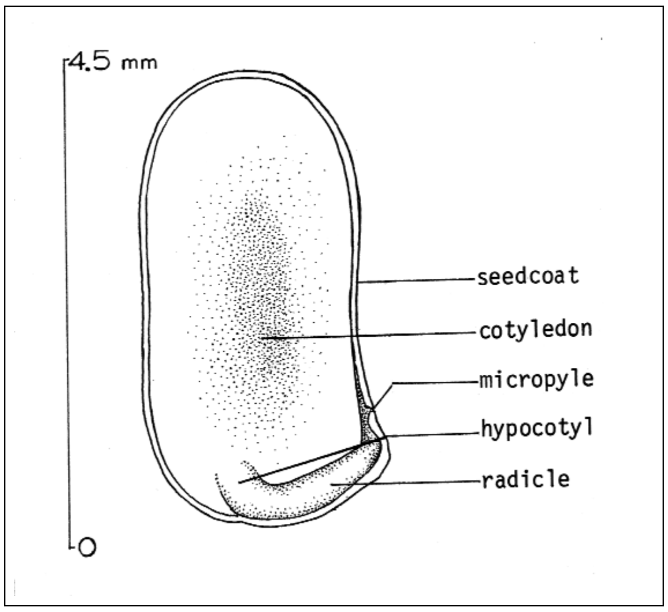
Figure 1—*Caragana arborsecens*, Siberian peashrub: legume.

Figure 2—*Caragana arborsecens*, Siberian peashrub: seed.



Extraction and storage of seeds. The legumes should be spread out to dry in a protected area until they pop open. The seeds can then be extracted easily by light maceration or beating. Legume fragments and other trash can be removed with aspirators, air-screen cleaners, or fanning mills. The average number of cleaned seeds per weight ranges from 28,700 to 48,500/kg (13,000 to 22,000/lb), with a purity of 97 to 100% (Dietz and Slabaugh 1974). A yield

Figure 3—*Caragana arborsecens*, Siberian peashrub: longitudinal section through a seed.



of 13 to 20 kg of seeds/100 kg (13 to 220 lb/100 lb) of fresh legumes has also been reported.

Seeds of Siberian peashrub, like those of other legumes, are orthodox in storage behavior. Studies in Canada have shown that the seeds remain viable for at least 5 years when stored dry at room temperatures. Germination of seedlots stored this way was 94% after 1 and 2 years and 93% after 5 years (Cram 1956). For the best long-term storage, seeds should be stored dry in polyethylene bags (or other sealed containers) at –18 to 4 °C, with a moisture content between 9.6 and 13.5% (Lindquist and Cram 1967).

Pregermination treatments and germination testing. For a leguminous species, Siberian peashrub does not have a very impermeable seedcoat. Untreated seeds will germinate in 15 days after sowing, but the best germination (87 to 100% in 5 days) can be obtained by soaking seeds for 24 hours in cold or hot (85 °C) water (Dirr and Heuser 1987). Successful germination has also been reported after acid scarification, cold stratification for 2 weeks, or fall planting (Dietz and Slabaugh 1974; Dirr and Heuser 1987; Hamm and Lindquist 1968; Lindquist 1960). Certain pesticides, such as captan and thiram, can apparently increase germination, possibly by inhibiting seed-borne disease (Cram 1969). The official testing prescription for Siberian peashrub seeds calls for clipping or filing through the seedcoat on the cotyledon end, soaking these seeds in water for 3 hours, then germinating them for 21 days at alternating temperatures of 20/30 °C (ISTA 1993). Germination tests have also been carried out in flats of sand or perlite and in Jacobsen germinators for 14 to 60 days at the same alternating temperatures (Dietz and Slabaugh 1974; Hamm and Lindquist 1968). Germination after 25 to 41 days averaged 45 to 72%, and 55 to 100% after 60 days (Dietz and Slabaugh 1974).

Nursery practice. Seeds of Siberian peashrub may be drilled or broadcast in late summer or spring. In a North Dakota nursery, Siberian peashrub is seeded during the last week in July or the first week in August. A cover crop of oats is seeded between the tree rows early enough to give winter protection. The shrubs are large enough to dig the following fall (Dietz and Slabaugh 1974). Many nurseries recommend drilling 80 to 160 seeds/m (25 to 50/ft) at 6, 9, or 12 mm ($^1/_4$ to $^1/_2$ in) depth; percentages of seeds growing into seedlings have varied from 35 to 50 (Dietz and Slabaugh 1974; Lindquist and Cram 1964).

Grading seeds for size has greatly increased the percentage of plantable seedlings. To be plantable, seedlings should be 30 cm (12 in) or more in height at the time of lifting. Only 87% of the seedlings grown from seeds measuring 2.5 mm in diameter were plantable, whereas 77% of seeds measuring 4.0 mm in diameter were plantable (Lindquist and Cram 1967). Inoculation of seeds with *Rhizobium* ssp. before sowing has been recommended (Wright 1947), but other workers report no significant effect on 1+0 seedlings (Cram and others 1964). Commercial nurseries have recommended anywhere from 1+0 to 3+0 stock for outplanting (Dietz and Slabaugh 1974).

Spraying to control insects in the nursery may be necessary. Grasshoppers are especially destructive to Siberian peashrub, sometimes completely defoliating the plants (Kennedy 1968). Plants have also been extensively damaged by deer browsing.

Vegetative propagation of Siberian peashrub is also possible. Untreated cuttings taken in late July rooted 80% in sand, whereas cuttings taken earlier (May to June) responded well to indole-butyric acid (IBA) in talc (Dirr and Heuser 1987).

C

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C

Cactaceae Cactus family

Carnegiea gigantea (Engelm.) Britton & Rose

saguaro or giant cactus

Susan E. Meyer

Dr. Meyer is a research ecologist at the USDA Forest Service's Rocky Mountain Research Station, Shrub Sciences Laboratory, Provo, Utah

Growth habit, occurrence, and use. Saguaro—*Carnegiea gigantea* (Engelm.) Britton & Rose—has the northernmost distribution of any of the large, columnar cacti of the tropical and subtropical Americas. Formerly regarded as a member of the genus *Cereus,* it is now considered the single species of its own genus, *Carnegiea*. It is a principal indicator species of the Sonoran Desert and is found at elevations below 1,200 m from extreme southeastern California east to south central Arizona and south into northern Sonora (Kearney and Peebles 1960; Munz 1964). Saguaro is an arborescent, sometimes branched, stem succulent that reaches 10 m in height. It is found primarily in desert upland communities with coarse, gravelly, well-drained soils.

Saguaro is an important component of the communities where it occurs, providing food and shelter to a host of desert animals. Its wood has been used for fence and hogan construction by indigenous people of the area, and its fruits provided one of their most reliable wild food sources. The sweet, fleshy fruit pulp can be eaten raw or used to make confections or jams. The nectar is reported to be a source of excellent honey (Alcorn and Martin 1974). In addition, saguaro is one of the most well-known and beloved plants in the country, recognizable at a glance by most Americans.

Flowering and fruiting. Saguaro plants normally produce fruit on a yearly basis, even if the winter has been dry (Steenbergh and Lowe 1977). They have sufficient water and energy reserves in their succulent stems to buffer fruit production from the yearly vagaries of water availability. Even plants that have been severed at the base are capable of fruit production for 2 subsequent years. Plants in the wild reach reproductive maturity at a height of about 2 m and an age of about 40 years (Steenbergh and Lowe 1983). Flowering occurs from March through May, depending on latitude and elevation, with later flowering on cooler sites. The fruit crop may be damaged or destroyed by frost during flowering. The large, fragrant, epigynous flowers are borne at the stem apices. They open in the evening, and each lasts only until midday the following day. The flowers produce copious pollen and nectar and are pollinated primarily by nectar-feeding bats, although many other visitors take advantage of the rich resource (Hevly 1979; Steenbergh and Lowe 1977).

Fruits ripen from June through August. A single fruit contains 2,000 to 2,500 seeds. The succulent fruits usually split open while still attached to the plant, exposing the tiny seeds (figures 1 and 2) to removal by rain, but the fruits eventually fall to the ground. Many animals utilize the fruits and seeds. Larger mammals such as coyotes (*Canis latrans*) may act as dispersers, but most users, especially harvester ants (*Pogonomyrmex* spp.) and doves (*Zenaida* spp.), are consumers only. Most of the seeds are consumed before the beginning of summer rains, especially in years when fruits ripen early or initiation of the summer rainy season is delayed (Steenbergh and Lowe 1977).

Figure 1—*Carnegiea giganteus*, saguaro: seeds.

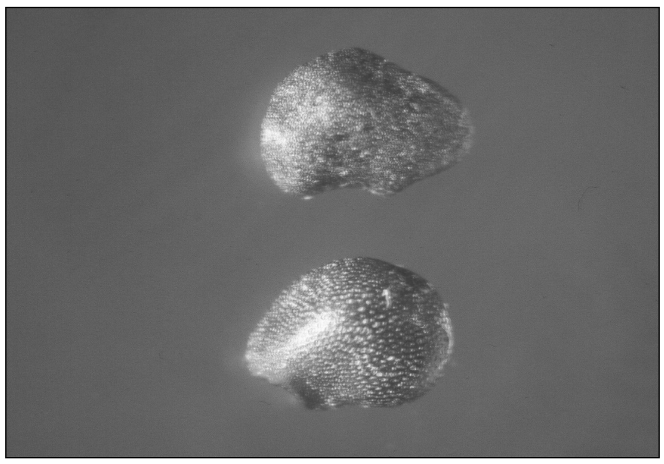
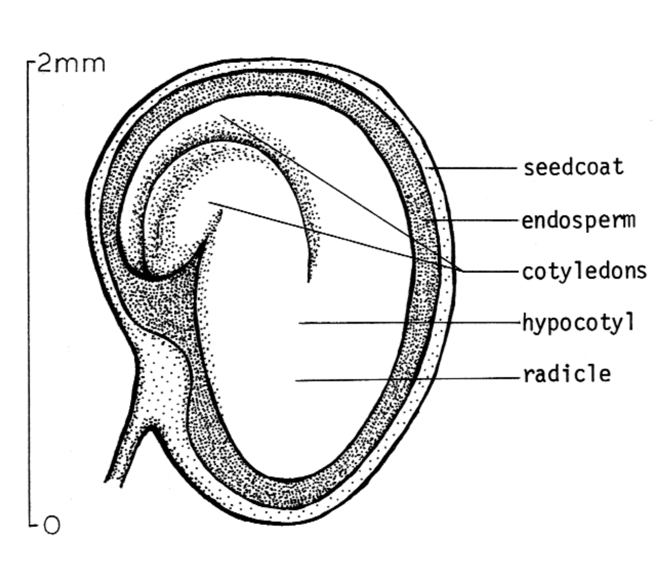


Figure 2—*Carnegiea giganteus*, saguaro: longitudinal section through a seed.



Seed collection, cleaning and storage. Ripe fruits turn from green to purple and can be collected by cutting with long-handled knives prior to dehiscence on the plant. The seeds can be removed from the fruits using standard procedures for fleshy fruited species, such as maceration in a macerator; forcing the fruits through an appropriately sized sieve; removing the pulp by flotation; drying the seeds and cleaning them in a fanning mill or aspirator. Extra care must be taken because of the small size of the seeds. The average number of seeds per weight is 990/g (450,000/lb) (Alcorn and Martin 1974). The seeds are usually of high quality (>95% germination of seedlots). Seeds apparently may be stored at room temperature for several years without much loss of viability, and germination values as high as 51% have been recorded, even after 10 years (Alcorn and Martin 1974).

Germination. Saguaro seeds are readily germinable when the fruits are ripe. Their germination is suppressed by the fruit pulp, but once they are washed free of the pulp they germinate freely, as long as temperatures are high (25 °C is optimum) and the seeds are exposed to light (Alcorn and Martin 1974; Steenburgh and Lowe 1977). Official testing calls for germination on moist blotter paper for 20 days at alternating temperatures of 20/30 °C, with light during the 8 hours at the higher temperature; no pretreatment is needed (AOSA 1993). In the field, seeds germinate soon after dispersal in response to adequate summer storms. Time to 50% germination of initially air-dried seeds is about 72 hours. If seedlots are first exposed to high-humidity air (without condensation) for 24 hours, they can reach 50% germination in 48 hours, an apparent adaptation for speeding germination during closely spaced summer storms (Steenbergh and Lowe 1977). Saguaro seeds do not form persistent seed banks; all viable seeds germinate soon after dispersal.

Because of apparently poor recruitment in natural stands, factors affecting survival of saguaro seedlings and young plants have been studied in some detail (Despain 1974; Nobel 1980; Steenbergh and Lowe 1969, 1976, 1977, 1983). New seedlings are highly susceptible to drought and herbivore damage, and young plants are at risk both of overheating in summer and freezing in winter. Nurse plants and other sheltering objects such as rocks greatly decrease these risks.

Nursery practice. Cleaned saguaro seeds may be surface-sown on coarse potting medium. The seedlings are highly susceptible to fungal pathogens, and care must be taken to provide good drainage and avoid overwatering (Alcorn and Martin 1974). They must be protected from freezing and also from full sunlight. Shade is probably beneficial for plants up to a meter (39 in) in height. The seedlings grow very slowly at first—only a few millimeters a year for the first several years.

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Hydrangeaceae—Hydrangea family

Carpenteria californica Torr.

carpenteria

Donald L. Neal

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Forest and Range Experiment Station

Growth habit. Carpenteria (bush-anemone or tree-anemone)—*Carpenteria californica* Torr.—is an erect evergreen shrub that is 1 to 3 (sometimes 4 m) tall with large showy white flowers. Plants are usually multi-stemmed and about as wide as tall. The leaves are oblong–lanceolate on short petioles and placed opposite. The leaves are leathery and in response to moisture stress, they turn yellow and twist and their edges roll under. They return to normal appearance and color when moisture is available (Kottcamp 1983; Sanwo 1997).

Occurrence. The range of carpenteria is extremely limited, occurring only between 300 and 1,500 m on the west slope of the Sierra Nevada, between the San Joaquin and Kings Rivers in eastern Fresno County, California. The shrub is found in scattered stands over an area 20 by 30 km or about 60,000 ha. The total number of plants has been estimated to less than 5,000 (Clines 1995).

Most stands are in small drainages on dry rocky slopes, mixed with Digger pine (*Pinus sabiniana* Dougl. ex Dougl.), interior live oak (*Quercus wislizeni A.* DC.), chaparral whitethorn (*Ceanothus leucodermis* Greene), and other representatives of the foothill woodlands at the lower elevational limits of its range. At their upper elevational limit, carpenteria plants can be found growing with ponderosa pine (*Pinus ponderosa* P. & C. Lawson), interior live oak, and other species of the lower yellow pine zone.

About two-thirds of the existing plants occur on lands of the USDA Forest Service's Sierra National Forest, and carpenteria is classified as a sensitive plant by the Forest Service. It receives some protection on 2 areas set aside by the Sierra National Forest and 1 owned by The Nature Conservancy. Carpenteria is a threatened species under the California Endangered Species Act, and in October of 1994 it was proposed for listing as endangered under the Federal Endangered Species Act (Federal Register 1994).

Natural reproduction. Until recently, all observed natural reproduction was by stump-sprouting after fire (Stebbins 1988; Wickenheiser 1989). However, after a large wildfire in 1989, an abundance of naturally occurring seedlings were found where mineral soil was exposed (Clines 1994) and many of them later had become established plants. In one study, hand-seeding in mineral soil after a fire produced abundant seedlings (Clines 1994). Stem layering and adventitious rooting have also been observed (Clines 1994).

Use. Carpenteria was first collected by the John C. Fremont expedition of 1845. It drew the attention of horticulturalists early and was found in gardens in the United States, England, and Europe by the early 1880s (Cheatham 1974). It is raised commercially for the garden in several California nurseries (Laclergue 1995).

Flowering and fruiting. Flowers are large (3 to 7 cm in diameter) and appear in May and June in a terminal cyme. The calyx is 5 (or 6) parted and there are 5 to 8 large white petals. The ovary is incompletely 5 (2 to 8)–celled. Up to 1,500 (2,000) ovules are attached to axile placentas that protrude into the locule (Clines 1994). The style has 5 to 7 closely grouped branches topped with numerous spreading yellow stamens. Pollination seems to be mostly outcrossing by insects but geitonogamy also produces viable seeds (Clines 1994).

Extraction and cleaning. Capsules (figure 1) can be collected by hand in July and August from native or commercially grown stands. The capsules are hard and must be cut open when collected intact. They can be found partially open on the shrubs later in the season. Each capsule may contain over 1,000 viable seeds (figure 2). Germination occurs easily without treatment and ranges from 70 to 100% (Mirov and Kraebel 1939; Clines 1994). There are 33 to 47.2 million seeds/kg (15.0 to 21.5 million/lb) (Mirov and Kraebel 1939).

Nursery practice. Carpenteria is grown in commercial nurseries both from seeds and cuttings. One common practice involves direct seeding into flats of well-drained soil. Damping-off is a problem and top dressing with perlite reduces but does not eliminate this problem. Cuttings root readily, especially those taken from the terminal few inches of the branches and treated with rooting compound.

C

Figure 1—*Carpenteria californica*, carpenteria: exterior view of fruit (**upper left**), cross section of fruit (**upper right**), exterior view of seeds in 2 planes (**bottom**).

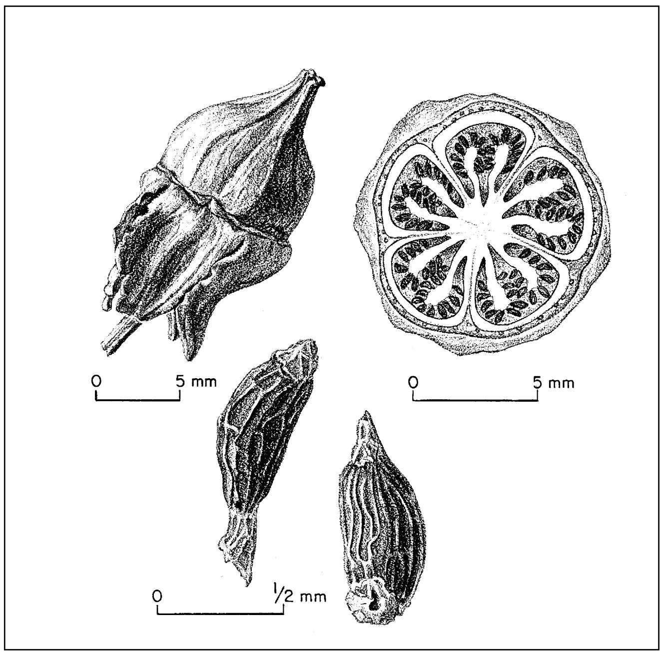
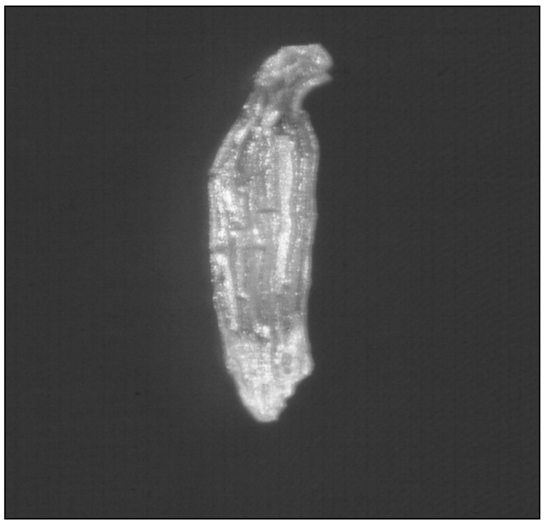


Figure 2—*Carpenteria californica*, carpenteria: seed.



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C

Betulaceae—Birch family

Carpinus L.

hornbeam or ironwood

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Growth habit, occurrence, and use. The hornbeam genus—*Carpinus* L.—includes about 35 species of deciduous, monoecious, small to large trees, that are native to the Northern Hemisphere from Europe to eastern Asia, south to the Himalayas, and in North and Central America (Furlow 1990; Hillier 1991; Krüssmann 1984; LHBH 1976; Suszka and others 1996). Five species are considered here (table 1). Hornbeams occur mainly as understory trees in rich, moist soils on bottomlands and on protected slopes (Metzger 1990; Rudolf and Phipps 1974). European hornbeam is an important forest tree species throughout Europe (Furlow 1990). In Mexico and Central America, *Carpinus tropicalis* (J.D. Sm.) Lundell forms a dominant canopy component (Furlow 1990). American hornbeams, which are native to the eastern United States and Canada, are smaller trees that grow in the mixed hardwood forest understory (Furlow 1990; Metzger 1990). Several geographic races of American hornbeam exist in North America (Fernald 1935; Furlow 1990). The races are morphologically variable and difficult to distinguish on the basis of independent characters. Furlow (1987a), using multivariate analysis, analyzed this geographical variation. The northern American hornbeam species is divided into the subsp. *caroliniana* from along the Atlantic and Gulf Coastal Plains of the southeastern United States and the subsp. *virginiana* of the Appalachian Mountains and northern interior regions to the West (Furlow 1987b). The Latin American *C. tropicalis* is divided into subsp. *tropicalis* of the highlands of southern Mexico and north Central America and subsp. *mexicana* of the mountains in northeastern Mexico and the trans-Mexican volcanic belt (Furlow 1987b).

The wood of hornbeams is extremely hard—hence the common name "ironwood"—and is used for making tool handles and mallet heads. It is also used to produce the high-quality charcoal used in gunpowder manufacture (Bugala 1993; Furlow 1990). Species of ornamental interest in the United States are listed in table 1. Most of the information presented in this chapter deals with European and American hornbeams, unless noted otherwise.

European hornbeam is a slow-growing tree (about 3 m over 10 years) that is pyramidal in youth but oval-rounded to rounded at maturity (Dirr 1990; Suszka and others 1996). This species is planted in the landscape as single specimen trees or as screens or hedges. It tolerates a wide range of soil and light conditions but grows and develops best in full sun on rich, moist sites with good drainage (Dirr 1990; Metzger 1990). Several cultivars produce excellent color, form, and texture. The cultivar 'Fastigiata' is the most common one in cultivation, with foliage more uniformly distributed along the branches than on other cultivars (Dirr 1990; Hillier

Table 1—*Carpinus,* hornbeam: nomenclature, occurrence, height at maturity, and date of first cultivation

Scientific name	Common name(s)	Occurrence	Height at maturity (m)	Year first cultivated
***C. betulus* L.**	**European hornbeam**	Europe, Asia Minor, & SE England	12–21	1800s
***C. caroliniana* Walt.**	**American hornbeam,** musclewood, blue beech, ironwood	Nova Scotia S to Florida, W to Texas, & N to Minnesota & Ontario; also in central & S Mexico & Central America	6–9	1812
***C. cordata* Blume**	**heartleaf hornbeam**	Japan, NE Asia, & China	6–15	1879
***C. japonica* Blume**	**Japanese hornbeam**	Japan	6–9	1895
***C. orientalis* Mill.**	**Oriental hornbeam**	SE Europe & SW Asia	6–8	1739

Sources: Dirr (1990), Hillier (1991), Krüssmann (1984), LHBH (1976), Metzger (1990).

1991; Krüssmann 1984). This cultivar is used primarily as a screen hedge because of its dense, compact, ascending branches (Dirr 1990). The bark on older trees is gray and beautifully fluted.

American hornbeam is a small, multi-stemmed, bushy shrub or single-stemmed tree with a wide-spreading, flat or round-topped crown, that grows slowly; averaging 2.5 to 3 m over a 10-year period (Dirr 1990; Metzger 1990). This species has considerable fall color variation, from yellow to orange-red, and is planted in the landscape in groups or as an understory tree (Beckett 1994; Dirr 1990). The bark on older trees is slate gray, smooth, and irregularly fluted; the overall appearance is comparable to the flexed bicep and forearm muscles—hence another common name, "muscle-wood" (Dirr 1990).

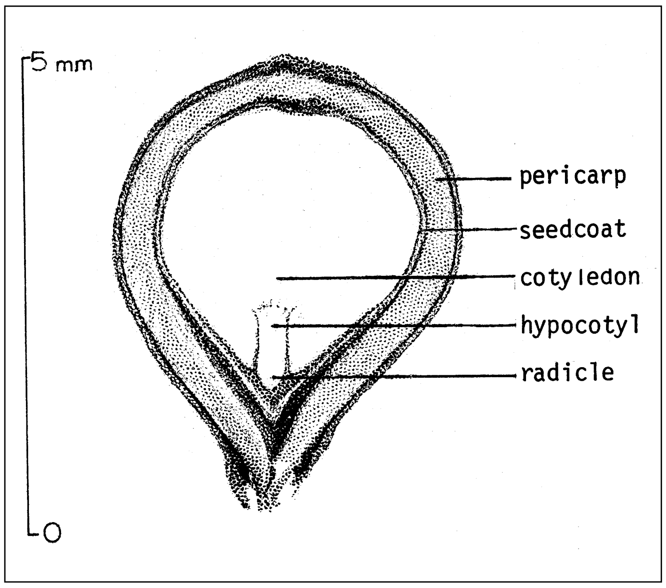
Heartleaf hornbeam is a small tree of rounded habit with leaves that are large with deeply heart-shaped bases, and with large, rich brown winter buds (Dirr 1990; Hillier 1991; Krüssmann 1984). The bark is slightly furrowed and scaly (Dirr 1990). The fruits are borne in cigar-shaped catkins (Dirr 1990). Japanese hornbeam is a wide-spreading small tree or large shrub with prominently corrugated leaves and with branches that radiate like the ribs on a fan (Dirr 1990; Hillier 1991). Oriental hornbeam grows as a large shrub or small tree with an overall U-shaped branching pattern (Dirr 1990). The bracts of this species are unlobed, differentiating it from European and American hornbeams (Dirr 1990). The main branches and stems are twisted, giving this species an interesting winter appearance (Dirr 1990).

Flowering and fruiting. In most species, the staminate and pistillate catkins appear in the spring concurrently as the trees are leafing out (Dirr 1990; Furlow 1990; Metzger 1990; Suszka and others 1996). The fruits are ovoid, ribbed, single-seeded nutlets (figures 1 and 2), each borne at the base of a distinctive 3-lobed involucre (bract) (Metzger 1990; Rudolf and Phipps 1974). The fruits ripen from late summer to fall. They are dispersed from fall to spring and are carried only a short distance by the wind or may be dispersed farther by birds (Rudolf and Phipps 1974). Details of flowering and seeding habits for European and American hornbeams are described in tables 2 and 3.

Collection of fruits; extraction, cleaning, and storage of seeds. Fruits harvested while they are still green (when the wings are turning yellow and are still soft and pliable) can be fall-sown for germination the following spring (Bugala 1993; Dirr 1990; Hartmann and others 1990). These seeds should not be allowed to dry out, as a hard seedcoat will develop, and they should be checked before sowing for the presence of well-developed embryos (Bugala 1993;

Figure 1—*Carpinus caroliniana*, American hornbeam: nutlet with involucre removed

Figure 2—*Carpinus caroliniana*, American hornbeam: longitudinal section through a nutlet.



Leiss 1985). Green seeds can also be stratified for 3 to 4 months over winter and sown the following spring (Hartmann and others 1990).

Mature seeds (with hardened seedcoats) should be collected, spread out in thin layers in a cool, well-aerated room or shed, and allowed to dry superficially (Macdonald 1986; Rudolf and Phipps 1974; Suszka and others 1996). The bracts do not need to be removed if the seeds are to be broadcast (Macdonald 1986). They should be removed, however, from large quantities of seeds (to aid in mechanical sowing) by placing the seeds in a de-winging machine or beating the seeds in bags (Rudolf and Phipps 1974; Suszka

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Table 2—*Carpinus*, carpinus: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening	Seed dispersal
C. betulus	Europe & NE US	Apr–May	Aug–Nov	Nov–spring
C. caroliniana	NE US	Mar–June	Aug–Oct	Nov–spring

Source: Rudolf and Phipps (1974).

Table 3—*Carpinus*, hornbeam: seed-bearing age, seedcrop frequency, seed weight, and fruit ripeness criteria

Species	Minimum seed-bearing age (yrs)	Years between large seedcrops	Average no. cleaned seeds /kg	Average no. cleaned seeds /lb	Preripe color	Ripe color
C. betulus	10–30	1–2	28,660	13,000	Green	Brown
C. caroliniana	15	3–5	66,138	30,000	Green	Greenish brown

Sources: Allen (1995), Rudolf and Phipps (1974).

and others 1966). The debris can be removed from seedlots by screening and fanning (Macdonald 1986). European hornbeam seeds with bracts weigh 15 to 18 kg/0.35 hl (33 to 40 lb/bu). Fruits weighing 45 kg (100 lb) yield about 23 kg (50 lb) of cleaned seed (Rudolf and Phipps 1974). The average numbers of cleaned seeds per weight of European and American hornbeam are listed in table 3.

Hornbeam seeds stratified immediately after extraction can be stored up to 2 years (Rudolf and Phipps 1974). European hornbeam seeds in nuts partially dried to 8 to 10% moisture content can be stored in sealed containers at a temperature of –3 °C for at least 5 years (Bugala 1993). Seeds of this species stored at 10% moisture content in sealed containers at 3 °C lost no viability after 14 months (Suszka and others 1969).

Pregermination treatments. Hornbeam seeds that are allowed to mature and become dry will develop a hard seedcoat. Dormancy, caused by conditions in the embryo and endosperm, may be overcome by stratification treatments (a warm period followed by a cold period). In general, 1 to 2 months of warm stratification followed by 2 to 3 months of cold stratification are necessary to break dormancy of the European hornbeam. The International Seed Testing Association (1993) prescribes 1 month of moist incubation at 20 °C, followed by 4 months at 3 to 5 °C, for laboratory testing of European hornbeam. Results of stratification treatments vary for different species of hornbeam, so several are presented in table 4. Bretzloff and Pellet (1979) reported that gibberellic acid treatment at 0.025, 0.1, and 0.5 g/liter (25, 100, and 500 ppm) generally increased germination of American hornbeam seeds stratified at 4 °C for 6, 12, or 18 weeks, compared to stratification alone. Scarification of the seedcoat plus gibberellic acid also improved germination (Bretzloff and Pellet 1979). Gordon and others (1991) and Suszka and others (1996) provide extensive information on the sampling, seed pretreatment, purity, viability, and germination testing, seedling evaluation, and storage of forest tree and shrub seeds. Specific procedures are presented for a number of species.

Germination tests. Germination percentage of stratified seeds is low, usually less than 60% and occasionally as low as 1 to 5% (Metzger 1990). Germination tests may be made on pretreated seeds in germinators, or in flats of sand, or sand plus peat (Rudolf and Phipps 1974). Viability of European and American hornbeams is best determined by using the tetrazolium test for viability (Chavagnat 1978; Gordon and others 1991; ISTA 1993; Suszka and others 1996). Details of germination test results are shown in table 5. Germination of hornbeam seeds is epigeal.

Nursery practice and seedling care. The optimum seedbed is continuously moist, rich loamy soil protected from extreme atmospheric changes (Rudolf and Phipps 1974; Suszka and others 1966). Germination of many naturally disseminated seeds is delayed until the second spring after seed dispersal (Rudolf and Phipps 1974). If germination is expected the first spring, seeds should be collected while they are still green (the wings turning yellow and still soft and pliable) and sown in the fall, or stratified immediately and sown the following spring (Bugala 1993; Dirr 1990; Hartmann and others 1990; Rudolf and Phipps 1974). Macdonald (1986) suggested collecting European hornbeam seeds in the fall, followed by extraction, stratification for 8

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Table 4—*Carpinus,* hornbeam: stratification treatments for breaking embryo dormancy

	Warm period		Cold period		
Species	Temp (°C)	Days	Temp (°C)	Days	Percentage germination
C. betulus	20	28	3–5	90–112	NS
	20	14	5	210	65
	20	30	4	120	65
C. caroliniana	20–30	60	5	60	10
	—	—	4.5	126	58
C. orientalis	20	60	5	90–120	NS

Sources: Allen (1995), Blomme and Degeyter (1977), Bretzloff and Pellet (1979), Bugala (1993), Rudolf and Phipps (1974), Suszka and others (1996).
NS = not stated.

Table 5—*Carpinus,* hornbeam: germination test conditions and results with stratified seed

	Test conditions*			Germination rate		% Germination		Purity	Soundness
	Temp (°C)								
Species	Day	Night	Days	%	Days	Avg	Samples	(%)	(%)
C. betulus	20	20	70	30	7	18–90	50	97	60
C. caroliniana	27	16	60	2	12	1–5	2	96	62

Source: Rudolf and Phipps (1974).
*Tests were made in sand or soil.

weeks at 18 to 21 °C and then for 8 to 12 weeks at 0.5 to 1 °C, and then spring-sowing. Seeds collected later should be partially dried, stratified, and sown the next fall or the following spring to avoid having seedbeds with germination spread out over 2 years (Rudolf and Phipps 1974). Seeds should be sown in well-prepared beds at a rate of 323 to 431/m^2 (30 to 40/ft^2) and covered with 0.6 to 1.3 cm ($^1/_8$ to $^1/_4$ in) of soil (Rudolf and Phipps 1974). Macdonald (1986) suggested sowing seeds at a rate of 250/m^2 (23/ft^2) for lining-out stock and 150 to 250/m^2 (14 to 23/ft^2) for rootstocks. Fall-sown beds should be mulched with burlap, pine straw, or other material until after the last frost in spring (Rudolf and Phipps 1974). The soil surface should be kept moist until after germination, and beds should shaded lightly for the first year (Rudolf and Phipps 1974). Davies (1987, 1988) demonstrated that growth of European hornbeam transplants was greatly increased by using a chemical for weed control and various synthetic sheet mulches. Black polythene sheets (125 m thick) gave the best results for controlling weeds and aiding in tree establishment (Davies 1988).

Cultivars of hornbeam may be grafted (side whip or basal whip) or budded onto seedlings of the same species (Hartmann and others 1990; Macdonald 1986; MacMillan-Browse 1974). Hornbeam can also be propagated by cuttings, but with variable success. Stem cuttings of European hornbeam 'Fastigiata' rooted when treated with 2% (20,000 ppm) indole-3-butyric acid (IBA); American hornbeam 'Pyramidalis' with 1 and 1.6% IBA; heartleaf hornbeam (var. *chinensis*) with 1.6 and 3% IBA–talc; and Japanese hornbeam with 3 g/liter (3,000 ppm) IBA–talc plus thiram (Cesarini 1971; Dirr 1990; Dirr and Heuser 1987; Obdrzalek 1987). After rooting, the cuttings require a dormancy period (Dirr 1990). Placing cuttings at a temperature of 0 °C during the winter months satisfies the dormancy requirements (Dirr 1990). Stock plant etiolation and stem banding have been shown to improve the rooting of hornbeam (Bassuk and others 1985; Maynard and Bassuk 1987, 1991, 1992, 1996). Chalupa (1990) reported the successful micropropagation of European hornbeam by using nodal segments and shoot tips as initial explants. Oriental hornbeam has been established in bonsai culture (Vrgoc 1994).

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Juglandaceae—Walnut family

Carya Nutt.

hickory

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Growth habit, occurrence, and use. Of the dozen or so species of hickories native to the United States, 9 are valuable for timber and the food they provide for wildlife (table 1). All are deciduous trees. Pecan and its many horticultural varieties and hybrids are widely cultivated for nuts in large plantations in the southern and southwestern United States, as well as in many other countries. The first known selections were made in 1846, and many cultivars were available by the late 19th century (Madden and Malstrom 1975). Budding and grafting have been the primary means of improvement, but new provenance studies (Grauke and others 1990) and advanced research on the reproductive biology and genetics of pecan (Graves and others 1989; McCarthy and Quinn 1990; Yates and Reilly 1990; Yates and Sparks 1990) demonstrate the promise for future improvements in nut production and disease resistance. Shellbark and shagbark hickories have also been planted for nut production.

Flowering and Fruiting. Hickories are monoecious and flower in the spring (table 2). The staminate catkins develop from axils of leaves of the previous season or from inner scales of the terminal buds at the base of the current

Table 1—*Carya,* hickory: nomenclature and occurrence

Scientific name & synonym(s)	Common name	Occurrence
C. alba **(L.) Nutt. ex Ell.** *C. tomentosa* (Lam. ex Poir.) Nutt. *Hicoria tomentosa* (Lam. ex Poir.) Raf.	**mockernut hickory,** bullnut, white hickory, whiteheart hickory, hognut, mockernut	S New Hampshire to S Michigan, S to E Texas & N Florida Valley to Illinois
C. aquatica **(Mich f.) Nutt.** *Hicoria aquatica* (Michx. f.) Britt.	**water hickory,** bitter pecan swamp hickory	Coastal plain from Virginia to S Florida & E Texas; N in Mississippi Valley to Illimois
C. cordiformis **(Wangenh.) K. Koch.** *Hicoria cordiformis* (Wagenh.) Britt.	**bitternut hickory,** bitternut, swamp hickory, pignut	New Hampshire to Minnesota, S to E Texas & Georgia
C. glabra **(P. Mill.) Sweet** *Hicoria glabra* (Mill.) Britt. *C. microcarpa* (Nutt.) Britt.	**pignut hickory,** sweet pignut, pignut, swamp hickory	New Hampshire to NE Kansas, S to Arkansas & NW Florida
C. illinoensis **(Wangenh.) K. Koch** *Hicoria pecan* (Marsh.) Britt. *C. oliviformis* (Michx. f.) Nutt. *C. pecan* (Marsh.) Engl & Graebn.	**pecan,** sweet pecan, *nuez encarcelada*	S Indiana to SE Iowa; S to Texas & E to Mississippi & W Tennessee; local to Ohio, Kentucky, & Alabama
C. laciniosa (Michx. f.) G. Don *Hicoria laciniosa* (Michx. f.) Sarg.	**shellbark hickory,** bigleaf shagbark hickory, big shellbark, kingnut, bottom shellbark, big shagbark hickory	Ohio & Mississippi Valleys; W New York to E Kansas, E to Georgia & Virginia; local in Louisiana, Alabama, & Virginia
C. myristiciformis **(Michx. f.) Nutt.** *Hicoria myristicaeformis* (Michx. f.) Britt.	**nutmeg hickory,** bitter water hickory, swamp hickory	Mississippi W to SE Oklahoma, S to E Texas & Louisiana; also E South Carolina & central Alabama
C. ovata **(P. Mill.) K. Koch** *Hicoria alba* Britt. p.p.; *H. ovata* (P. Mill.) Britt.	**shagbark hickory,** scalybark hickory, shagbark, shellbark hickory	Maine to SE Minnesota, S to E Texas & Georgia
C. pallida **(Ashe) Engl. & Graebn.** *Hicoria pallida* Ashe	**sand hickory,** pale hickory, pallid hickory	New Jersey & Illinois, S to Florida & SE Louisiana

Sources: Little (1979), Sargent (1965).

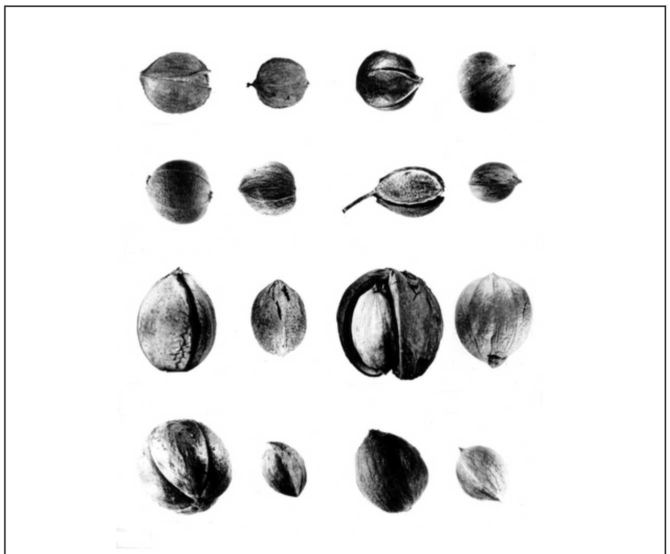
Table 2—*Carya,* hickory: phenology of flowering and fruiting

Species	Flowering	Fruit ripening	Seed dispersal
C. alba	Apr–May	Sept–Oct	Sept–Oct
C. aquatica	Mar–May	Sept–Nov	Oct–Dec
C. cordiformis	Apr–May	Sept–Oct	Sept–Dec
C. glabra	Apr–May	Sept–Oct	Sept–Oct
C. illinoensis	Mar–May	Sept–Oct	Sept–Oct
C. laciniosa	Apr–June	Sept–Nov	Sept–Oct
C. myristiciformis	Apr–May	Sept–Oct	Sept–Oct
C. ovata	Apr–June	Sept–Oct	Sept–Oct
C. pallida	Mar–Apr	Sept–Oct	Sept–Oct

Source: Bonner and Maisenhelder (1974).

growth. The pistillate flowers appear in short spikes on peduncles terminating in shoots of the current year. Hickory fruits are ovoid, globose, or pear-shaped nuts enclosed in husks developed from the floral involucre (figure 1). Husks are green prior to maturity and then turn brown to brownish black as they ripen (Bonner and Maisenhelder 1974). The husks become dry at maturity in the fall (table 2) and split away from the nut into 4 valves along sutures. Husks of mockernut, nutmeg, shagbark, and shellbark hickories, as well as those of pecan, split to the base at maturity, usually releasing the nuts. Husks of pignut, bitternut, sand, and water hickories split only to the middle or slightly beyond and generally cling to the nuts. The nut is 4-celled at the base and 2-celled at the apex. The edible portion of the embryonic plant is mainly cotyledonary tissue (figure 2) and has a very high lipid content (Bonner 1971; Bonner 1974; Short and Epps 1976).

Figure 1—*Carya*, hickory: nuts with husks attached and removed (the size and shape of individual nuts varies greatly within a species and may differ from the examples shown here); *C. aquatica*, water hickory (**first row left**) and *C. cordiformis*, bitternut hickory (**first row right**); *C. glabra*, pignut hickory (**second row left**) and *C. myristiciformis*, nutmeg hickory (**second row right**); *C. illinoensis*, pecan (**third row left**) and *C. laciniosa*, shellbark hickory (**third row right**); *C. ovata*, shagbark hickory (**fourth row left**) and *C. alba*, mockernut hickory (**fourth row right**).



Collection, extraction, and storage. Hickory nuts can be collected from the ground after natural seedfall or after shaking the trees or flailing the limbs. Persistent husks may be removed by hand, by trampling, or by running the fruits through a macerator or a corn sheller. Several studies have shown that the larger nuts of pecan make larger seedlings (Adams and Thielges 1977; Herrera and Martinez 1983), so sizing of nuts may be beneficial. Shagbark and shellbark hickory trees have been known to produce 0.5 to 0.75 hl ($1^1/_2$ to 2 bu) and 0.75 to 1.1 hl (2 to 3 bu) of nuts, respectively (Bonner and Maisenhelder 1974). Good crops of all species are produced at intervals of 1 to 3 years (table 3). Some typical yield data are presented in table 4.

Figure 2—*Carya ovata*, shagbark hickory: longitudinal section through the embryo of a nut with husk removed.

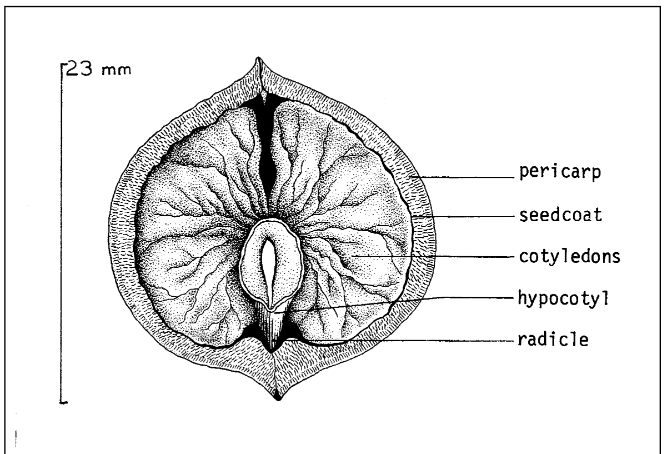


Table 3—*Carya,* hickory: height, seed-bearing age, seedcrop frequency, and year first cultivated

Species	Height at maturity (m)	Year first cultivated	Minimum seed-bearing age (yrs)	Years between seedcrops
C. alba	30	1766	25	2–3
C. aquatica	30	1800	20	1–2
C. cordiformis	15–30	1689	30	3–5
C. glabra	24–27	1750	30	1–2
C. illinoensis	34–43	1766	10–20	1–2
C. laciniosa	37	1800	40	1–2
C. myristiciformis	24–30	—	30	2–3
C. ovata	21–30	1911	40	1–3
C. pallida	12-30	—	—	2–3

Source: Bonner and Maisenhelder (1974).

Table 4—*Carya,* hickory: seed data

						Cleaned seeds/weight			
	Place	Fruits/vol		Seed wt/fruit vol		Range		Average	
Species	collected	/hl	/bu	kg/hl	lb/bu	/kg	/lb	/kg	/lb
C. alba	—	—	—	—	—	75–249	34–113	200	90
	Mississippi	5,040	1,776	57	44	71–106	32–48	79	36
C. aquatica	Mississippi	—	—	—	—	305–419	138–140	360	164
C. cordiformis	—	—	—	51	40	275–408	125–185	344	156
C. glabra	—	—	—	51	40	386–496	175–225	441	200
	Mississippi	10,100	3,552	—	—	—	—	143	65
C. illinoensis	—	—	—	—	—	121–353	55–160	220	100
	Mississippi	20,800	7,330	—	—	333–384	151–174	357	162
	Texas	—	—	—	—	—	—	311	141
C. laciniosa	—	—	—	—	—	55–77	25–35	66	30
C. myristiciformis	Mississippi & Arkansas	14,500	5,110	—	—	207–375	94–170	273	124
C. ovata	—	17,600	6,200	38–49	30–38	176–331	80–150	220	100
	Wisconsin	—	—	—	—	—	—	291	32
	Mississippi	12,100	4,264	—	—	—	—	207	94

Source: Bonner and Maisenhelder (1974).

Storage tests with pecan and shagbark hickory have demonstrated that the hickories are orthodox in storage behavior, that is, they should be dried to low moisture contents and refrigerated. Seedlots of nuts of both species dried to below 10% moisture and stored at 3 °C in sealed containers retained viability well for 2 years before losing half to two-thirds of their initial viability after 4 years (Bonner 1976b). The poor results after 4 years are probably due to the high lipid levels in these seeds, which places them in the sub-orthodox storage category (Bonner 1990). There are no storage data for other species of hickory, but it is reasonable to think that they can be stored in a similar fashion.

Pregermination treatments. Hickories are generally considered to exhibit embryo dormancy, although work with pecan suggests that mechanical restriction by the shell is the reason for delayed germination in that species (van Staden and Dimalla 1976). Other research with pecan has shown that there is a clinal gradient in stratification requirement. Seedlots from southern sources are practically nondormant, whereas those from northern sources require treatment for prompt germination (Madden and Malstrom 1975). The common treatment is to stratify the nuts in a moist medium at 1 to 4 °C for 30 to 150 days (table 5). Stratification of

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imbibed nuts in plastic bags without medium is suitable for most species (Bonner and Maisenhelder 1974), and good results have been reported for pecans from southern sources by soaking the nuts at 20 °C for 64 hours (Goff and others 1992). There are indications that stratification should be shortened for stored nuts; this was the case in one storage test on pecan and shagbark hickory (Bonner 1976b). If cold storage facilities are not available, stratification in a pit with a covering of about 0.5 m of compost, leaves, or soil to prevent freezing will suffice. Prior to any cold stratification, nuts should be soaked in water at room temperature for 2 to 4 days with 1 or 2 water changes each day to ensure full imbibition (Eliason 1965). There is evidence that germination of pecan can be increased by treatment with gibberellins (Bonner 1976a; Dimalla and van Staden 1977), but practical applications have not been developed.

Germination tests. Official testing rules for North America (AOSA 1993) prescribe testing pecan and shagbark hickory at alternating temperatures of 20 °C (dark) for 16 hours and 30 °C (light) for 8 hours on thick creped paper for 28 days. Stratification for 60 days as described above is also recommended. Adequate germination tests can also be made on stratified nuts in flats of sand, peat, or soil at the same temperature regime (table 5). Quick tests with tetrazolium salts can also be used with hickories (Eliason 1965).

Nursery practice. Either fall-sowing with untreated seed or spring-sowing with stratified seed may be used. Excellent results with fall-sowing have been reported for shagbark hickory, but good mulching is necessary (Heit 1942). Drilling in rows 20 to 30 cm (8 to 12 in) apart and 2 to 4 cm ($^3/_4$ to 1 $^1/_2$ in) deep with 20 to 26 nuts/m (6 to 8/ft) is recommended; about 100 seedlings/m^2 (10/ft^2) is a good density (Williams and Hanks 1976). Mulch should remain until germination is complete. Shading is generally not necessary, but shellbark hickory may profit from shade. Protection from rodents may be required for fall-sowings.

Table 5—*Carya, hickory:* stratification period, germination test conditions, and results

Species	Cold stratification (days)	Germination test conditions: Medium	Temp (°C) Day	Temp (°C) Night	Days	Germination Rate (%)	Germination Days	Germination % Avg (%)	Samples
C. alba	90–150	Sand, peat, soil	30	20	93	54	64	66	4
C. aquatica	30–90	Soil	27–32	21	63	76	28	92	1
C. cordiformis	90	Sand, peat, soil	30	20	250	40	30	55	3
	90	Soil	27	21	50	60	50	60	1
C. glabra	90–120	Sand, peat, soil	30	20	30–45	—	—	85	2
C. illinoensis	30–90	Sand, peat	30	20	45–60	—	—	50	9
	30–90	Kimpak	30*	20	60	80	33	91	6
	30	Soil	32	21	35–97	—	—	75	2
C. laciniosa	90–120	Sand, peat, soil	30	20	45–60	—	—	—	—
C. myristiciformis	60–120	Kimpak	30*	20	60	53	50	60	2
C. ovata	90–150	Sand, peat	30	20	45–60	75	40	80	6
	60–120	Kimpak	30*	20	60	65	35	73	2

Source: Bonner and Maisenhelder (1974).
* Daily light period was 8 hours.

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Fagaceae—Beech family

Castanea P. Mill.

chestnut

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Growth habit, occurrence, and use. The genus *Castanea*—the chestnuts—comprises 11 species of small to medium-sized deciduous trees found in southwestern and eastern Asia, southern Europe, northern Africa, and the eastern United States. Five species are covered in this chapter; only 2 are native to the United States (table 1). American chestnut formerly ranked as one of the most valuable timber species in the Appalachian region, and the nuts were an important wildlife food as well as being extensively marketed for human consumption. In the years since the chestnut blight—*Cryphonectria parasitica* (Murr.) Barr—was discovered in New York in 1904, the disease has spread throughout the range of the American chestnut and completely destroyed it as a commercial species. Many rootstocks still survive and send up multiple sprouts that grow to the size of a small tree (table 2) before dying. Some of these sprouts occasionally produce a few seeds, but they usually do not live long enough for significant production (Sander 1974).

Japanese, Chinese, and European chestnuts (table 1) were introduced into the United States in the 18th and 19th centuries (Anagnostakis 1990; Sander 1974). The Asian species demonstrated good resistance to the chestnut blight, and breeding programs were started as early as the 1890s to transfer the resistance to American chestnut (Jaynes 1975). Chinese chestnut, the most promising of these introductions, has been widely planted throughout the eastern United States, mostly in orchards for nut production. Allegheny chinkapin is somewhat resistant to the blight and might be useful as a rootstock in grafting; its other good features are small size, precocity of fruiting, and heavy seedcrops (Payne and others 1994). Breeding for resistance has not been highly successful, but advances in tissue culture offer new promise (Dirr and Heuser 1987).

Flowering and fruiting. Chestnuts are monoecious, but some trees produce bisexual catkins also (Sander 1974). Unisexual male catkins, 15 to 20 cm long, appear near the base of the flowering branches. The pistillate flowers occur singly or in clusters of 2 to 3, near the end of the branches (Brown and Kirkman 1990; Sander 1974), with the female catkins at the base of the shoot (Payne and others 1994). Flowering begins in April or May in the Southeast (Hardy 1948) and in June in the Northeast (Sander 1974).

Chestnut fruits are spiny, globose burs, from 2.5 to 7.5 cm in diameter, borne singly or in spikelike clusters (Sander 1974; Vines 1960). The fruits each contain from 1 to 3 seeds (nuts); Allegheny chinkapins have 1 seed and American chestnuts (figure 1) have 3 seeds/fruit (Brown and Kirkman 1990; Sander 1974). The nuts are flattened on one side and range from light to dark brown or black in color (Brown and Kirkman 1990; Rehder 1940). Nuts of American chestnut are 12 to 25 mm wide and about 25 mm long. The exotic chestnuts bear larger nuts that are 19 to 38

Table 1—*Castanea,* chestnut: nomenclature and occurrence

Scientific name	Common name(s)	Occurrence
C. crenata **Siebold & Zucc.**	**Japanese chestnut**	Japan
C. dentata **(Marsh.) Borkh.**	**American chestnut**	S Maine to Michigan; S to S Mississippi & Georgia
C. mollissima **Blume**	**Chinese chestnut**	China & Korea
C. pumila **(L.) P. Mill.**	**Allegheny chinkapin**	Pennsylvania S to central Florida & W to E Texas & Oklahoma
C. sativa **P. Mill.**	**European chestnut,** Spanish chestnut	S Europe, W Asia, & N Africa

Sources: Little (1979), Sander (1974).

Table 2—*Castanea,* chestnut: height, year first cultivated, and seed weights

Species	Height at maturity (m)	Year first cultivated in US	Cleaned seeds/weight /kg	Cleaned seeds/weight /lb
C. crenata	10	1876	33	15
C. dentata	20–25*	1800	220–360	100–162
C. mollissima†	21	1853	50–220	23–100
C. pumilla	15	—	300	136
C. sativa	21	Pre 1880	33	15

Sources: Payne and others (1994), Sander (1974).
* Height refers to sprouts from living rootstocks of trees killed by the blight; before the blight this species obtained heights of 21 to 30 m.
† Bears large crops annually in orchards beginning at about 8 years of age.

Figure 1—*Castanea dentata*, American chestnut: fruit (bur) and nut.

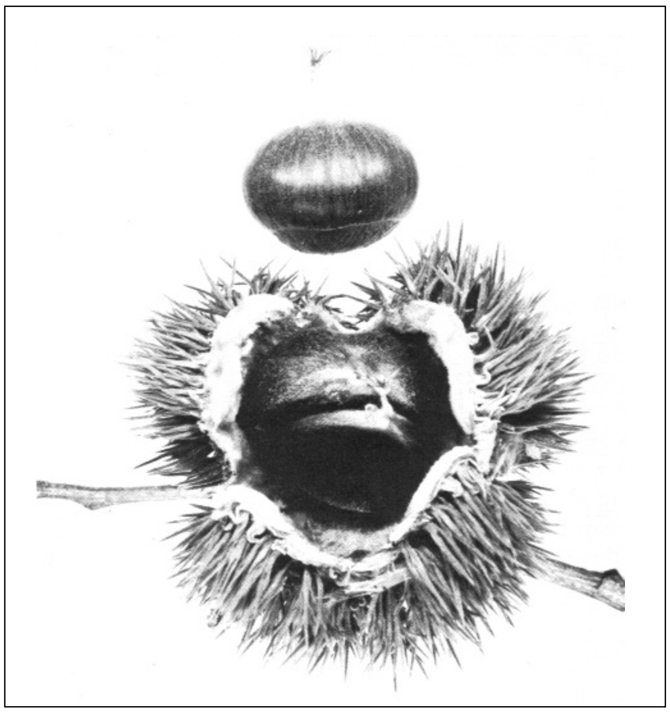
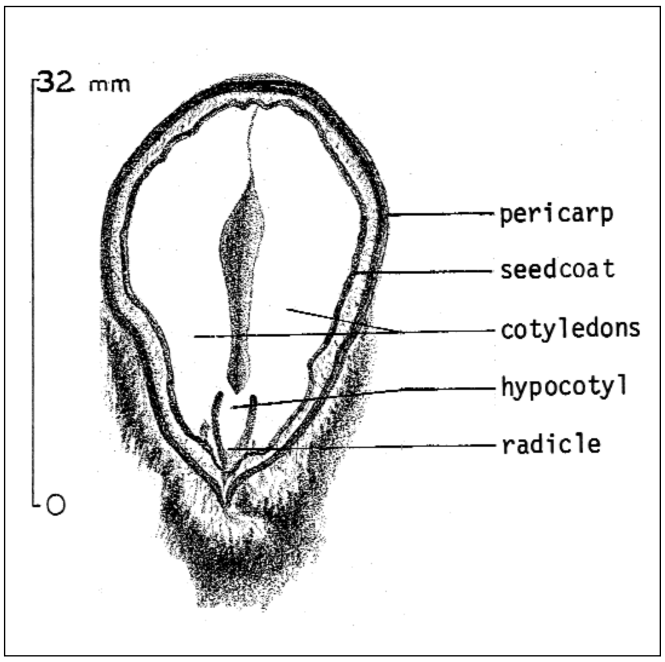


Figure 2—*Castanea dentata,* American chestnut: longitudinal section through a nut.



mm wide (Sander 1974). Food reserves, primarily starch, are stored in the large cotyledons (figure 2). Fresh nuts are 40 to 45% starch by weight, with very little lipid content (Jaynes 1975; Payne and others 1994; Wainio and Forbes 1941). Seeds ripen in August to October, depending on species and location (Hardy 1948; Sander 1974). Seed weights are listed in table 2.

Superior strains and hybrids. There are no identified superior strains of native chestnuts, but many cultivars and hybrids have been developed with the exotic chestnuts, primarily in Europe. The search for blight-resistant American chestnuts continues, however, with breeding, tissue culture, and innovative budding and grafting techniques (Ackerman and Jayne 1980; ACF 2002).

Collection of fruits. Chestnuts can be picked from the trees, collected from the ground by hand, or shaken from the trees onto ground cloths. Burs of Allegheny chinkapin do not open widely, and the seeds are difficult to shake out. Some remain on the trees throughout winter (Payne and others 1994). Harvesting should begin as soon as the burs begin to split open. The nuts are intolerant of desiccation (recalcitrant) (Aldous 1972; Pritchard and Manger 1990), so collections from the ground should be done very soon after dissemination to prevent excessive drying. Frequent collection

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is especially important if the weather is hot and dry, as nuts can lose viability within a week on the ground (USDA 1951). If the weather is wet, Allegheny chinkapin nuts will sometimes germinate on the trees (Payne and others 1994).

Storage of seeds. Because of their recalcitrant nature, chestnuts are normally stored no longer than 6 months (overwinter). With good care, however, storage for 18 months is not difficult, and some have been successfully stored for 3.5 years (Jaynes 1975). Immediately after collection, the nuts should be floated in water to remove trash and immature and damaged nuts. If collected from the ground in a dry condition, they should be left in water overnight to restore their naturally high moisture content. Upon removal from water, the nuts should be spread to dry in a cool, well-ventilated place to remove all surface moisture. The nuts should be placed in containers that inhibit drying, such as polyethylene bags, and stored at 1 to 3 °C; however, the containers should not be airtight so that some gas exchange between nuts and the storage atmosphere is possible. Moisture content of the nuts should be about 40 to 45% during storage (Sander 1974). Too much moisture can result in loss of seeds to microorganisms (Woodruff 1963).

Pregermination treatments. Chestnut seeds are dormant and require a period of cold, moist stratification for prompt germination. In normal nursery practice, overwinter storage of fully imbibed nuts at 1 to 3 °C will satisfy the chilling requirement to overcome dormancy. For nuts that have not been stored moist, or if a deeper dormancy than usual is suspected, then stratification should be used; 1 to 3 months is the recommended period for American and Chinese chestnuts (Dirr and Heuser 1987; Jaynes 1975). If nuts are planted in the fall, stratification is not necessary, but the nuts should be kept in cold storage until planted (Sander 1974).

Chestnuts are commonly infested with the larvae of the seed weevils *Curculio sayi* Gyllenhall and *C. caryatrypes* Bohemon (Gibson 1985). A simple method to kill the larvae is to submerge the nuts for 20 to 40 minutes in water at 49 °C (Payne and Wells 1978).

Germination tests. The standard laboratory testing procedure for European chestnut is to (1) soak the seeds in water for 24 hours; (2) cut off a third of the seed at the cup-scar end; (3) remove the testa; and (4) germinate the seeds for 21 days in or on top of sand at the standard test regime of alternating 20 and 30 °C (ISTA 1993). If only constant temperatures are available, 28 °C is recommended for this species, which also has no specific light requirement of germination (Pritchard and Manger 1990). Data are lacking on other chestnut species with this procedure, but it quite likely will work for any of them. There are alternate procedures for whole nuts. Stratified nuts of Chinese chestnut have been germinated in a moist medium at 15 to 21 °C; germination reached 100% in 42 days (Berry 1960).

Nursery practice. Chestnuts may be planted in autumn or spring. Nuts that have been kept in cold storage from the time they are harvested should be planted in September or October (Sander 1974). Fall-sown beds should be mulched and protected as much as possible against rodents (Williams and Hanks 1976). Nuts for spring-planting should be stratified for 2 to 3 months.

In both fall- and spring-plantings, nuts should be sown 2 to 4 cm ($^3/_4$ to 1 $^1/_2$ in) deep and spaced 7.5 to 10 cm (3 to 4 in) apart in rows 7.5 to 15 cm (3 to 6 in) apart in the nursery beds. Nuts can be either sown or drilled by hand, or broadcast mechanically (Sander 1974; Williams and Hanks 1976). Some growers recommend planting by hand so that the nuts can be placed on their sides to promote better seedling form (Jaynes 1975). European chestnuts are normally broadcast at a density of 100 nuts/m^2 (9 to 10/ft^2) (Aldous 1972). One should expect 75 to 80% germination in beds with good seeds (Aldous 1972; Sander 1974). A study with Chinese chestnuts found that grading nuts by size had no influence on time of emergence, although larger seeds did tend to produce larger seedlings (Shepard and others 1989).

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C

Casuarinaceae—Casuarina family

Casuarina Rumph. ex L.

casuarina

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Growth habit, occurrence, and use. The genus *Casuarina*—the only genus in the Casuarina family—comprises about 50 species, chiefly Australian, with a few having native ranges extending from Bangladesh to Polynesia. Casuarina trees are evergreen angiosperms, resembling conifers, with thin crowns of drooping branches and leaves reduced to scales (Little 1949; Little and Wadsworth 1964). Three species of this genus have been introduced successfully into continental United States, Hawaii, and Puerto Rico (table 1) (Bailey 1949; Rockwood and others 1990). Beach she-oak, especially, is planted as a windbreak throughout its native and introduced ranges and as an ornamental in parks and gardens (Parrotta 1993; Rockwood and others 1990). It was first introduced into Hawaii in 1882 (Neal 1965). The bark has been used in tanning, in medicine, and for the extraction of dye (Parrotta 1993). The fruits are made into novelties and Christmas decorations (Little and Wadsworth 1964). The wood is hard and heavy and is difficult to work, hence the common name "ironwood." It was once heavily used for building poles and firewood but now is seldom used commercially in the United States (Parrotta 1993). Beach and gray she-oaks are considered invasive pests in southern Florida and gray she-oak in Hawaii.

Flowering and fruiting. Casuarinas are monoecious or dioecious. Minute male flowers are crowded in rings among grayish scales. Female flowers lack sepals but have pistils with small ovaries and threadlike dark red styles (Little and Wadsworth 1964). The multiple fruit is conelike, about 8 to 20 mm in diameter (figure 1), and composed of numerous individual fruits. Each fruit is surrounded by 2 bracteoles and a bract that splits apart at maturity and releases a 1-winged light brown samara (figures 2 and 3). The immature fruits of the genus are green to gray-green, becoming brown to reddish brown when ripe (Neal 1965). In warm climates, flowering and fruiting occur throughout the year. Consequently, time of seed collection varies from place to place (Little and Wadsworth 1964; Olson and Petteys 1974). In Hawaii, Florida, and Puerto Rico, the peak of the flowering period appears to be April through June, with fruiting from September through December (Magini and Tulstrup 1955; Neal 1965; Olson and Petteys 1974; Rockwood and others 1990). Minimum seed-bearing age is 2 to 5 years, and good seedcrops occur annually (Magini and Tulstrup 1955; Olson and Petteys 1974; Parrotta 1993).

Collection, extraction, and storage. The multiple fruits may be picked from the trees or shaken onto canvas or

Table 1—*Casuarina,* casuarina: nomenclature, occurrence, and uses

Scientific name(s) & synonym	Common name(s)	Occurrence (native & introduced)
C. cunninghamiana **Miq.** *C. tenuissima* Hort.	**river she-oak,** river-oak casuarina, Cunningham beefwood, ironwood	Australia & New Caledonia; Hawaii, S US, & California
C. equisetifolia **L.** *C. litorea* L.	**beach she-oak,** Australian pine, horsetail casuarina, horsetail beefwood	Burma through Australia & Polynesia; Hawaii, Florida, & Puerto Rico
Casuarina glauca **Sieb. ex Spreng.**	**gray she-oak,** longleaf casuarina, longleaf ironwood	Australia; Hawaii

Sources: Olson and Petteys (1974), Parrotta (1993).

Figure 1—*Casuarina cunninghamiana*, river she-oak: multiple fruit.

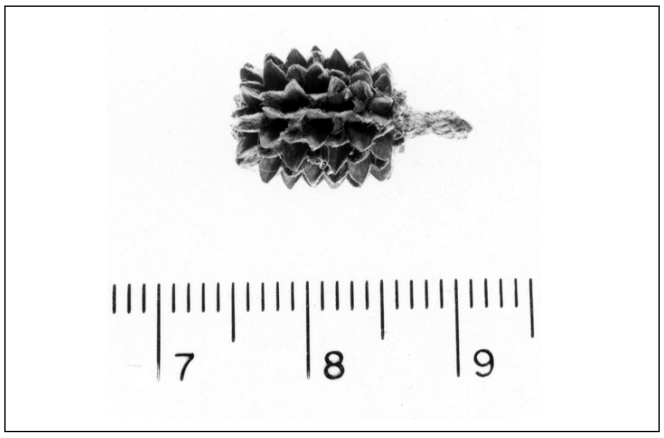


Figure 2—*Casuarina cunninghamiana*, river she-oak: longitudinal section through a seed.

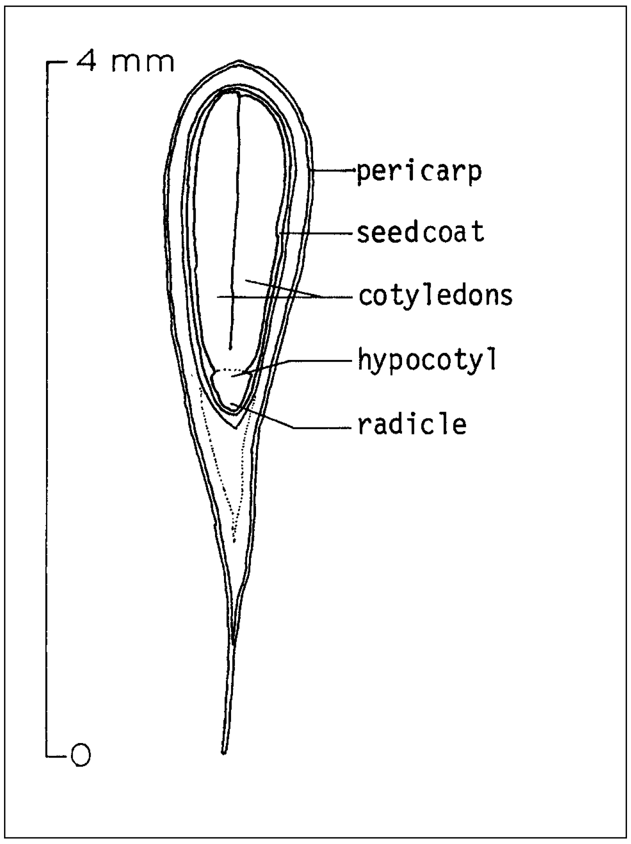
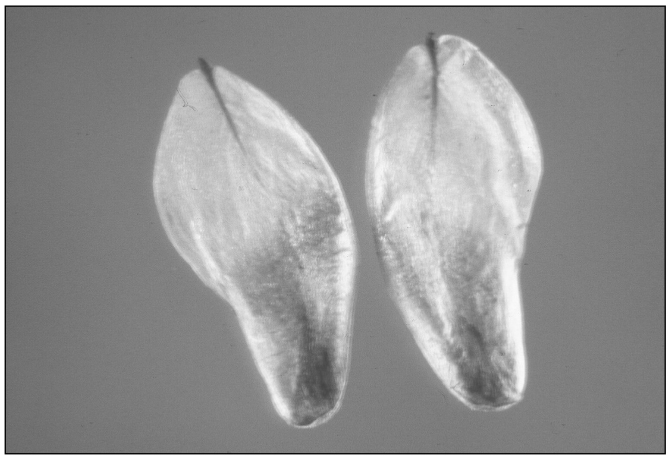


Figure 3—*Casuarina cunninghamiana*, river she-oak: winged seeds.



plastic sheets. Seeds reach maximum weight and germinability 18 weeks after anthesis, or when cones change in color from green to brown (Rai 1990). The samaras, which are used as seeds, may be separated from the fruits by shaking and screening (Olson and Petteys 1974). Cones placed in trays, covered by a thin cloth, and dried under full sunlight will soon begin to release their seeds, usually within 3 days (Kondas 1990). A kilogram of fruits (about 250 cones) yields between 20 and 60 g of seeds (1 lb of cones yields 1.5 to 2.4 oz of seeds). There are about 650 to 760 seeds/g (300,000 to 350,000 seeds/lb) (Kondas 1990; Turnbull and Markensz 1990). The application of an insect repellant effective against ant predation is advisable during the drying process (Kondas 1990). Seeds do not retain their viability for more than 3 months at ambient temperatures (Kondas 1990), but appear to be orthodox in storage behavior (Jones 1967). Seeds stored at subfreezing (–7 °C) or close to freezing (3 °C) temperatures, with moisture contents ranging from 6 to 16%, retain viability for up to 2 years (Turnbull and Markensz 1990). In Hawaii, seeds have been successfully stored in sealed polyethylene bags at 1 °C (Olson and Petteys 1974).

Germination. Germination in beach she-oak is epigeal; it takes place 4 to 22 days after sowing and is optimal at 30 °C under well-lighted conditions (Parrotta 1993). Casuarina seeds are usually sown without pretreatment (Magini and Tulstrup 1955; Olson and Petteys 1974), although soaking seeds for 36 hours in a 1.5% solution of potassium nitrate reportedly enhances germination (Rai 1990). Germination ranges from 40 to 90% for fresh seeds and from 5 to 25% for seeds stored in airtight containers at 4 °C for 1 year (Parrotta 1993). Official test prescriptions for casuarina species call for a 14-day test at alternating temperatures of 20/30 °C on the top of moist blotter paper (AOSA 1993). In the Philippines, germination of seedlots collected from different trees within a single plantation ranged from 33 to 75% for fresh seeds (Halos 1983). A significant positive relationship between cone size and seed germination rate was also noted in this study.

C

Nursery practice. In the nursery, seeds are generally germinated in trays under full sunlight at an optimal density of 1,000 to 7,500 seeds (weighing 2 to 10 g) /m^2 (93 to 700 seeds/ft^2), covered with about 0.5 cm of soil (Olson and Petteys 1974; Parrotta 1993). In South Africa, seedling yield averages are 18,000 plants/kg (8,200/lb) of river she-oak seeds (Magini and Tulstrup 1955). Nursery soils should be light textured, optimally sandy loams or a mixture of sand and peat moss. Seedlings are transferred from germination trays to containers when they reach a height of 10 to 15 cm (4 to 6 in), usually within 6 to 10 weeks after germination. Seedling containers measuring about 15 cm (6 in) in diameter and 20 cm (8 in) in depth are recommended. Seedlings may also be transplanted to new beds at densities of 100 to 400 seedlings/m^2 (9 to 37/ft^2) to obtain bareroot planting stock. Seedlings should be kept under partial shade until shortly before outplanting. Seedlings reach plantable size of 20 to 50 cm (8 to 20 in) in height in 4 to 8 months (Parrotta 1993). It is recommended that seedlings be inoculated in the nursery with pure cultures of effective strains of *Frankia* (a nitrogen-fixing actinomycete) or an inoculum from a nodule suspension prepared from fresh, healthy nodules collected in the field. Roots can be inoculated by dipping them into the suspension or by directly applying the suspension to the soil. Alternatively, crushed, fresh nodules, leaf litter, or soils from the vicinity of effectively inoculated trees may be incorporated directly into the nursery potting mix (Parrotta 1993).

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C

Bignoniaceae—Trumpet-creeper family

Catalpa Scop.

catalpa

Franklin T. Bonner and David L. Graney

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Growth habit, occurrence, and use. The catalpas include about 10 species of deciduous or rarely evergreen trees native to North America, the West Indies, and eastern Asia (Rehder 1940). Two deciduous species, southern catalpa and northern catalpa (table 1), are native to the continental United States and have been planted quite widely outside their native range, especially northern catalpa. Mature trees of both species attain heights of 9 to 18 m (Little and Delisle 1962; Sargent 1965). Both have been grown to some extent in Europe. Catalpas are used in shelterbelts and ornamental planting and have minor value as timber trees, mainly for posts and small poles. Haitian catalpa, or yokewood, a native of the West Indies, has also been widely planted for forestry and ornamental purposes (table 1).

Flowering and fruiting. Attractive clusters of showy, white perfect flowers with purplish and orange blotches and stripes in the throat are borne in May and June on southern and northern catalpas (Brown and Kirkman 1990; Sargent 1965). Fruits of these species ripen in October, and good crops are borne every 2 to 3 years beginning at about age 20 (Bonner and Graney 1974; Sargent 1965; Vines 1960). Haitian catalpa flowers, which vary from white to solid rose in color, appear irregularly throughout the year. Even 6-month-old seedlings flower, and abundant seed crops are borne by the age of 18 months (Francis 1993). Mature fruits are round, brown, 2-celled capsules, 15 to 75 cm long (figure 1). In late winter or early spring, the capsules of northern and southern catalpas split into halves to disperse the seeds (Sargent 1965). Each capsule contains numerous oblong, thin, papery, winged seeds 1 to 5 cm long and about 1 to 6 mm wide (figure 2). Removal of the papery outer seedcoat reveals an embryo with flat, round cotyledons (figure 3). Southern and northern catalpas are separate from each other. The most consistent identification feature is the relative thickness of the fruit walls. Northern catalpa fruit walls are considerably thicker than those of southern catalpa (Brown and Kirkman 1990).

Collection, extraction, and storage. Fruits should be collected only after they have become brown and dry. When dry enough, the seeds can be separated by light beating and shaking. Pods of northern catalpa collected in February and March had seeds of higher quality than those collected in

Table 1—*Catalpa,* catalpa: nomenclature and occurence

Scientific name & synonym(s)	Common name(s)	Occurrence	Year first cultivated
***C. bignonioides* Walt.** *C. catalpa* (L.) Karst.	**southern catalpa,** common catalpa, Indian-bean, catawba, cigar-tree	SW Georgia & Florida to Louisiana; naturalized to New England, Ohio, Michigan, & Texas	1726
***C. longissima* (Jacq.) Dum.-Cours.**	**Haitian catalpa,** yokewood, *roble de olor, chenn*	Hispaniola & Jamaica; naturalized in Martinique, Guadeloupe, & the Grenadines; planted in Florida, Hawaii, & the West Indies	—
***C. speciosa* (Warder) Warder ex Engelm.**	**northern catalpa,** hardy catalpa, western catalpa, western Catawba-tree, Indian-bean, catawba	SW Indiana & Illinois to NE Arkansas & W Tennessee; widely naturalized in NE & SE US	1754

Sources: Bonner and Graney (1974), Francis (1990), Little (1979).

Figure 1—*Catalpa bignonioides*, southern catalpa: capsule and leaf.

Figure 2—*Catalpa speciosa,* northern catalpa: seed.

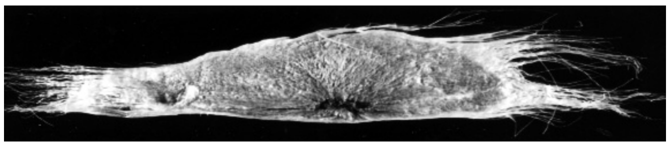
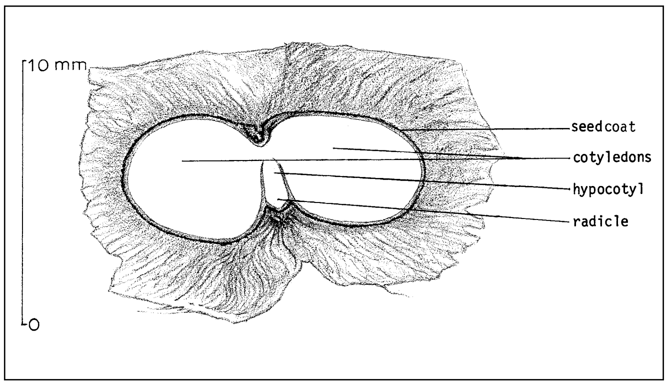


Figure 3—*Catalpa speciosa*, northern catalpa: longitudinal section through a seed.



the fall (Bonner and Graney 1974). In terms of size, seeds of northern catalpa are slightly smaller than seeds of southern catalpa, and seeds of Haitian catalpa are by far the smallest of these three (table 2). Seeds of all 3 species dried to about 10% moisture content can be stored under refrigerated conditions. Successful storage for 2 years has been reported for southern catalpa (Bonner and Graney 1974) and 1 year for Haitian catalpa (Francis 1990). Long-term storage has not been studied, but this genus appears to be orthodox in storage behavior and capable of extended storage at low moisture contents and temperatures.

Germination tests. Seeds of all 3 species germinate promptly without pretreatment. Tests should be made on wet germination paper for 21 days with 20 °C night and 30 °C day temperatures. Other moist media also are satisfactory. Although northern catalpa is known to be photosensitive (Fosket and Briggs 1970), light is not necessary for germination tests (AOSA 1993). Germination in excess of 90% (25+ samples) has been obtained in about 12 days with good quality seeds of southern and northern catalpas (Bonner and Graney 1974). Francis (1993) has reported 40% germination of Haitian catalpa in 8 days on potting mix. Germination is epigeal, and the emerging 2-lobed cotyledons look like 4 leaves (figure 4).

Nursery practice. Catalpa seeds should be sown in late spring in drills at the rate of about 100/linear m (30/ft), and covered lightly with 4 mm ($^1/_8$ in) of soil or sown on the surface. A target bed density of 108 to 215 seedlings/m^2 (10 to 20/ft^2) is recommended (Williams and Hanks 1990). Stratification or other pretreatment is not needed. A pine needle mulch has been recommended for southern catalpa (Bonner and Graney 1974). In Louisiana, this species starts germination about 12 days after March sowing and germination is about 80%. In Puerto Rico, Haitian catalpa seeds can be spread thinly on a shaded bed of moist, sterile soil or

Figure 4—*Catalpa bignonioides*, southern catalpa: seedling development at 1, 5, 8, and 20 days after germination.

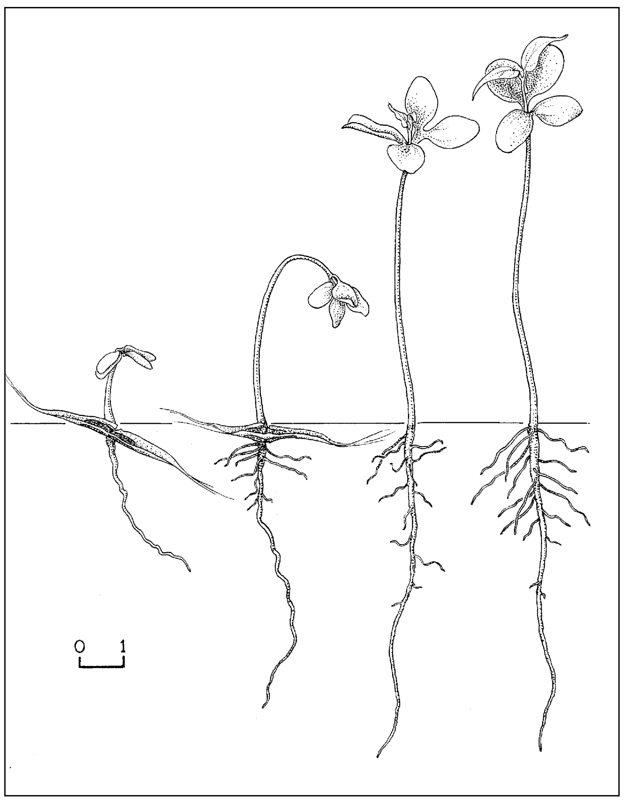


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Table 2—*Catalpa,* catalpa: seed data

Species	Place collected	Wt yield of seeds/ 100wt fruit	Cleaned seeds/weight Range /kg	/lb	Average /kg	/lb
C. bignonioides	Florida	—	32,600–40,10	14,800–18,200	36,400	16,500
		35	30,900–81,600	14,000–37,000	44,100	20,000
C. longissima	Puerto Rico	—	572,000–618,000	259,460–280,325	600,000	272,160
C. speciosa	Minnesota	—	29,450–48,300	13,359–21,910	—	—
	Prairie states	10–25	30,000–80,700	13,600–36,600	—	—
		25–35	35,300–66,150	16,000–30,000	46,300	21,000

Sources: Bonner and Graney (1974), Francis (1990).

sand and covered lightly with sand (Francis 1990). This species can also be sown directly into containers; germination begins in about 10 days. Nematodes, powdery mildews, and the catalpa sphinx—*Ceratonia catalpae* (Boisduval)—may give trouble in the nursery. Southern and northern catalpas are normally planted as 1+0 stock (Bonner and Graney 1974). Haitian catalpa seedlings should be ready for planting 10 to 14 weeks after sowing in containers. Untreated woody cuttings can also be used for vegetative propagation of Haitian catalpa (Francis 1990).

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C

Rhamnaceae—Buckthorn family

Ceanothus L.

ceanothus

Susan G. Conard and Merton J. Reed

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Growth habit, occurrence, and use. Van Rensslaer and McMinn (1942) recognized 55 species of ceanothus, 25 varieties, and 11 named natural hybrids, all restricted to the North American continent. Most of them are found along the Pacific Coast of the United States, and only 2 are found east of the Mississippi River (Hitchcock and others 1961; Kearney and Peebles 1951; Munz and Keck 1968; Rowntree 1948; Sampson and Jesperson 1963; Schmidt 1993; Van Rensslaer and McMinn 1942). Forty-three species and 21 varieties are described in the most recent flora of California, which does not recognize hybrid forms (Schmidt 1993). Although hybridization appears to be common in nature, there are few named hybrids (Lentz and Dourley 1981; Schmidt 1993). Ceanothus species are mainly evergreen or deciduous shrubs, some of which may attain the height of small trees. In the West, they occur in a diversity of habitats, ranging from interior desert chaparral to moist redwood forest along the Pacific Coast (table 1). Ceanothus species are important as wildlife food and shelter, for erosion control, as hedges and shelterbelts, and in soil development and soil nitrogen regimes (Conard and others 1985; Graham and Wood 1991). Deerbrush ceanothus is rated as one of the most important summer browse species in California for deer and cattle (Sampson and Jesperson 1963), and redstem ceanothus is a key winter browse plant for deer (*Odocoileu* spp.) and elk (*Cervus cervus*) in parts of Idaho, Washington, and Oregon (Hickey and Leege 1970). All species that have been investigated bear root nodules containing a nitrogen-fixing *Frankia* symbiont (for example, Delwiche and others 1965); species from both forest and chaparral systems have been associated with accretion of soil nitrogen over time (Binkley and Husted 1983; Binkley and others 1982; Conard and others 1985; Hellmers and Kelleher 1959; Youngberg and Wollum 1976; Youngberg and others 1979; Zavitkovski and Newton 1968; Zinke 1969).

On forest sites, ceanothus species have alternately been considered a problem because they compete with commercial conifers and a benefit because of their nitrogen-fixing ability and their wildlife value (Conard and others 1985). Although there was early experimentation with planting ceanothus species for erosion control on chaparral sites (DFFW 1985) and there has been some interest in ceanothus establishment for browse or general site improvement in forest sites (Hickey and Leege 1970; Radwan and Crouch 1977), the dominant horticultural uses have continued to be for domestic, commercial, and right-of-way landscaping—particularly in California and the Pacific Northwest. Ceanothus species are valued particularly for their showy flowers (they are sometimes called "California lilacs"), relatively rapid early growth, drought adaptation, and ability to tolerate landscape watering. Some species have been cultivated for many years (table 2), and the potential for hybridization has led to the development of numerous cultivars, many of which are available from commercial native plant nurseries (for example, Lentz and Dourley 1981; Perry 1992). Distribution and uses of some of the more common species are described in table 1.

Flowering and fruiting. Flowers are small, bisexual, and regular, and are borne in racemes, panicles, or umbels. The 5 sepals are somewhat petal-like, united at the base with a glandular disk in which the ovary is immersed. The 5 petals are distinct, hooded, and clawed; the 5 stamens are opposite the petals, with elongated filaments. Petals and sepals can be blue, white purple, lavender, or pink. The ovary is 3-celled and 3-lobed, with a short 3-cleft style. Fruits are drupaceous or viscid at first but soon dry up into 3-lobed capsules (figure 1) that separate when ripe into 3 parts. Seeds are smooth, varied in size among species (figures 2 and 3; table 3), and convex on one side.

Flowering and fruiting dates for several species are listed in table 2. Feltleaf ceanothus is reported to begin bearing seeds at 1 year (Van Rensslaer and McMinn 1942), deerbrush ceanothus at 3 years (McDonald and others 1998),

Table 1—*Ceanothus,* ceanothus: nomenclature and occurrence

Scientific name & synonym(s)	Common name	Occurrence
C. americanus **L.**	**New-Jersey-tea,** Jersey-tea,	Dry woods, Ontario to Manitoba, Maine to North Dakota, S to Florida & Texas
C. arboreus **Greene** *C. arboreus* var. *glabra* Jepson	**feltleaf ceanothus,** island myrtle, Catalina ceanothus	Larger islands of Santa Barbara Channel, California (up to 300 m on dry slopes & chaparral)
C. cordulatus **Kellogg**	**mountain whitethorn,** snowbush, whitethorn ceanothus	Baja California & mtns of S California, N to SW Oregon, E to Nevada (900–2,900 m on rocky ridges & ponderosa pine to red fir forests)
C. crassifolius **Torr.**	**hoaryleaf ceanothus**	Cis-montane southern California & Baja California (to 1,100 m on dry slopes & ridges, chaparral)
C. cuneatus **(Hook.) Nutt.**	**buckbrush ceanothus,** wedgeleaf ceanothus, hornbrush, buckbrush	Inner Coast Range & Sierra Nevada foothills, California into Oregon, S to Baja California (100–1,800 m in chaparral & ponderosa pine forests)
C. cuneatus **var.** ***rigidus*** **(Nutt.) Hoover** *C. rigidus* Nutt.	**Monterey ceanothus**	San Luis Obispo Co., N through Mendocino Co., California (up to 200 m on coastal bluffs, in closed-cone pine forests)
C. diversifolius Kellogg	**trailing ceanothus,** pinemat, Calistoga ceanothus	Westside Sierra Nevada, spotty in northern Coast Range, California (900–1,800 m, under oaks & pines)
C. fendleri **Gray**	**Fendler ceanothus,** buckbrush	South Dakota to New Mexico, Arizona, & Mexico (1,500 to 3,000 m, in ponderosa pine to dry Douglas-fir forests)
C. greggii **Gray**	**desert ceanothus,** mountain buckbrush	W Texas to S California, Utah, & N Mexico (300–2,300 m, chaparral & desert chaparral)
C. impressus **Trel.**	**Santa Barbara ceanothus**	Coastal areas in Santa Barbara & San Luis Obispo Cos., California (to 200 m in dry, sandy flats & slopes)
C. integerrimus **Hook & Arn.** *C. andersonii* Parry	**deerbrush ceanothus,** sweet-birch, blue bush, deer brush	N California, Oregon, Washington to S California, Arizona, & New Mexico (300–2,100 m, in ponderosa pine to western hemlock, white fir forests; chaparral in SW)
C. leucodermis **Greene**	**chaparral whitethorn**	S California to N Baja California (to 1,800 m in chaparral, dry slopes)
C. megacarpus **Nutt.**	**bigpod ceanothus**	California
C. oliganthus **Nutt.** *C. hirsutus* Nutt. *C. divaricatus* Nutt.	**hairy ceanothus,** jimbrush	Coast Ranges, San Luis Obispo & Santa Barbara Cos. & San Gabriel Mtns to Humboldt Co., California (to 1,300 m in chaparral)
C. prostratus **Benth.**	**prostrate ceanothus,** squaw-carpet, mahala mat, squaw mat	Sierra Nevada & N Coast Range S to Calaveras Co., California; higher mtns of Oregon & Washington, W Nevada (900–2,200 m, common in ponderosa & Jeffrey pine forests)
C. sanguineus **Pursh** *C. oreganus* Nutt.	**redstem ceanothus,** Oregon-tea tree	N California, Oregon, Idaho, Washington, & W Montana to S British Columbia (around 1,200 m in ponderosa pine Douglas-fir/mixed conifer, western hemlock zones)
C. sorediatus **Hook. & Arn.** *C. intricatus* Parry *C. oliganthus* var. *sorediatus* (Hook. & Arn.) Hoover	**jimbrush ceanothus,** jimbrush	Coast Range in Los Angeles & Riverside Cos., Parry to Humboldt Co., California (150–1,000 m, in chaparral)
C. thyrsiflorus **Eschsch.** *C. thyriflorus* var. *repens* McMinn	**blueblossom,** wild lilac	Coastal mountains Santa Barbara Co., California, to Douglas Co., Oregon (from sea level to 600 m in coast redwood, mixed-evergreen, Douglas-fir forest, & chaparral
C. velutinus **Dougl. ex Hook.**	**snowbrush ceanothus,** mountain balm, sticky-laurel, tobacco brush	Coast Ranges, British Columbia to Marin Co., California, Siskiyou Mtns, Sierra Nevada/Cascade axis E to SW Alberta, Montana, South Dakota, & Colorado (to 3,000 m, in many forest types, ponderosa pine to subalpine)
C. velutinus **var.** ***hookeri*** **(M.C. Johnston)** *C. velutinus* var. *laevigatus* Torr. & Gray	**varnish-leaf ceanothus**, Hooker ceanothus, snowbrush	Same as above, but more common near coast

Sources: Franklin and others (1985), Hitchcock and others (1961), Lenz and Dourley (1981), McMinn (1964), Munz and Keck (1968), Reed (1974), Sampson and Jesperson (1963), Schmidt (1993).

C

Figure 1—*Ceanothus*, ceanothus: capsules of *C. americanus*, New-Jersey-tea (**top**) and *C. velutinus*, snowbrush (**bottom**).

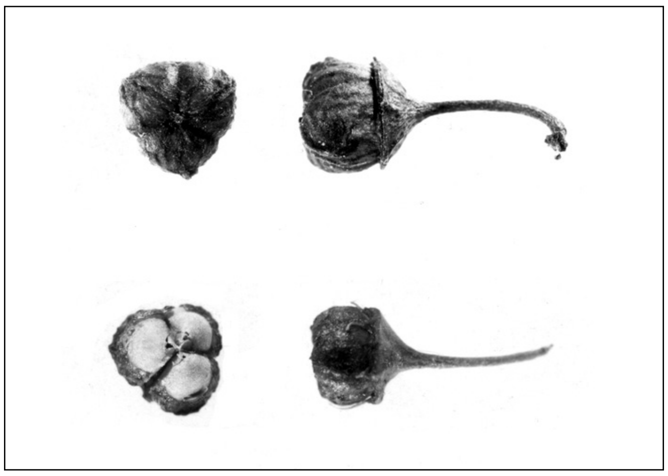
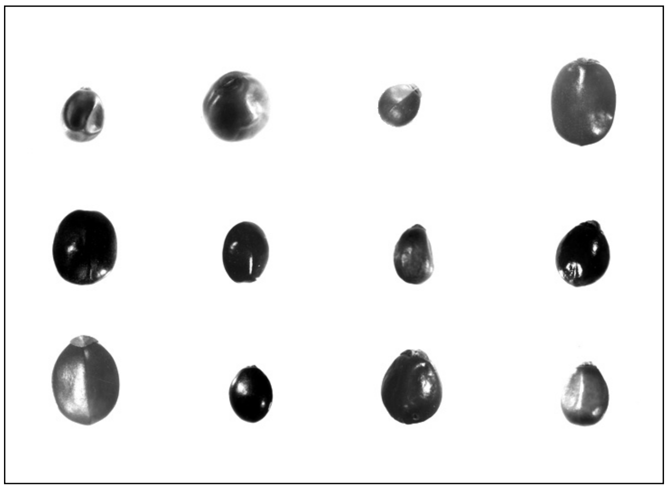
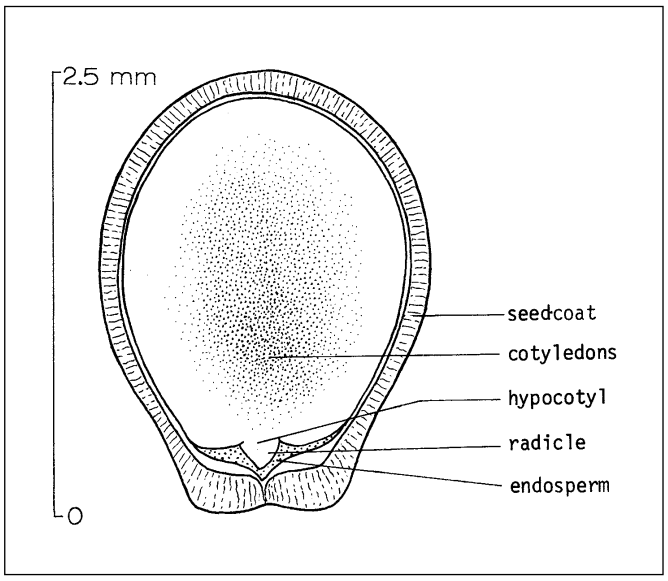


Figure 2—*Ceanothus*, ceanothus: seeds of *C. americanus*, New-Jersey-tea; *C. arboreus*, feltleaf ceanothus; *C. cordulatus*, mountain whitethorn; and *C. crassifolius* (**top, left to right**); *C. cuneatus*, buckbrush ceanothus; *C. impressus*, Santa Barbara ceanothus; *C. integerrimus*, deerbrush ceanthosus; and *C. oliganthus*, hairy ceanothus (**middle, left to right**); *C. prostratus*, prostrate ceanothus; *C. sorrediatus*, jimbrush ceanothus, *C. thyrsiflorus*, blueblossom; and *C. velutinus*, snowbrush ceanothus (**bottom, left to right**).



hoaryleaf ceanothus at 5 years (Everett 1957), desert ceanothus at 6 to 8 years (Zammit and Zedler 1992), and snowbrush ceanothus at 6 to 10 years (McDonald and others 1998). Thus it appears that most species can be expected to begin producing seeds by about 5 to 10 years of age. Fendler ceanothus has been reported to bear good seedcrops annually (Reed 1974). However, in hoaryleaf ceanothus, desert ceanothus, chaparral whitethorn, and other species,

Figure 3—*Ceanothus americanus*, New-Jersey-tea: longitudinal section through a seed.



both annual seed production and the amount of seeds stored in the soil may be quite variable (Conard and others 1985; Keeley 1977, 1987b; Zammit and Zedler 1988).

Collection, extraction, and storage. Several useful points on collecting ceanothus seeds have been described (Van Rensslaer and McMinn 1942; Emery 1988). Seeds should be collected only from vigorous plants, as weak, diseased plants do not produce sound seeds. To obtain plants similar to mature specimens, seeds should be collected in single-species stands in the wild or from isolated garden plants. Because many species hybridize freely, asexual propagation is the only certain way of maintaining species or varieties free from hybridization (Lenz and Dourley 1981). As the capsules split, ripe seeds are ejected with considerable force, such that about two-thirds of the seeds fall outside the shrub canopy, to distances up to 9 m (Evans and others 1987). Therefore, a common method of seed collection is to tie cloth bags—preferred to paper—securely over clusters of green seedpods. It is also possible to cut seedpod clusters before capsules have split, but the degree of maturity of the seeds is critical, as few prematurely collected seeds will germinate. Seeds that contain milky or gelatinous substances are not mature enough to harvest (Emery 1988). Green seeds should be air-dried at 29 to 38 °C.

If necessary, the seeds can be separated from capsule fragments by screening and blowing (Reed 1974), or seeds can be passed through a mill and floated (Plummer and others 1968). Average number of cleaned seeds per weight ranges from 90,000 to over 350,000/kg (41,000 to

Table 2—*Ceanothus, ceanothus:* phenology of flowering and fruiting, height, and year of first cultivation

Species	Flowering	Fruit-ripening	Height at maturity (m)	Year first cultivated
C. americanus	May–July	Aug–early Oct	0.5–1	1713
C. arboreus	Feb–Aug	May–early Oct	3–9	1911
C. cordulatus			0.6–2.5	—
California	May–June	July–Sept	—	—
Oregon	June–July	Aug–Sept	—	—
C. crassifolius	Jan–June	May–June	1.2–3	1927
C. cuneatus	Mar–June	Apr–July	1–4.5	1848
C. cuneatus var. *rigidus*	Dec–Apr	May–June	1–2.1	1847
C. diversifolius	Spring	June–July	0.3 or less	1941
C. fendleri (Arizona)	Apr–Oct	Aug–Dec	0.2–1	1893
C. greggii (Arizona*)	Mar–Apr	July	0.6–1.8	—
C. impressus	Feb–Apr	June	—	—
C. integerrimus	Apr–Aug	June–Aug	1–5.5	1850
C. leucodermis	—	July–Aug	—	
C. oliganthus	Feb–Apr	May–June	1.2–7.5	—
C. prostratus	Apr–June	July	.05–.15	—
C. sanguineus	Apr–June	June–July	1.5–3	1812
C. sorediatus	Mar–Apr	May–July	1–5.5	—
C. thyrsiflorus	Jan–June	Apr–July	1.2–8	1837
C. velutinus			0.6–2.4	1853
California	June–Aug	July–Aug	—	—
N Idaho†	May 20–July 25	July 15–Aug 1	—	—
W Montana‡	June 25–July 15	Aug 10–Sept 10	—	—
SW Oregon	May–July	July–Sept	—	—
Utah	—	Aug 1–Aug 30	—	—

Sources: Evans and others (1987), Furbush (1962), Hitchcock (1961), Hubbard (1958), Kearney (1951), McMinn (1964), Mirov and Kraebel (1939), Plummer and others (1968), Reed (1974), Rowntree (1948), Sampson and Jesperson (1963), Swingle (1939), Van Dersal (1938), Van Rensslaer (1942).
Elevations: * 900–1,500 m. † 700 m. ‡ 1,650 m.

178,000/lb), depending on the species (table 3). Adequate information on long-term storage is not available, but the seeds are apparently orthodox in storage behavior. Dry storage at around 4.5 °C should be satisfactory. Quick and Quick (1961) reported good germination in seeds of a dozen ceanothus species that had been stored for 9 to 24 years, with no apparent effect of seed age on viability. Seeds are apparently long-lived in litter; viable seeds of snowbrush ceanothus have been found in the surface soil of forest stands that were between 200 to 300 years old (Gratkowski 1962).

Germination. The long-term viability of seeds of ceanothus species apparently results from a strong seed coat dormancy, which in nature is typically broken by fire but may occasionally be broken by solar heating or mechanical scarification, such as from forest site preparation activities (Conard and others 1985). Germination of ceanothus seeds generally increases with increasing fire intensity (Conard and others 1985; Moreno and Oechel 1991), although at very high intensities, soil temperatures may be high enough to kill substantial numbers of seeds, resulting in decreased germination (Lanini and Radosevich 1986). In varnish-leaf ceanothus, Gratkowski (1962) observed that when seeds were exposed to drying conditions at normal air temperature, the hilum (the attachment scar on the seed, through which the radicle would normally emerge) functioned as a one-way hygroscopic valve that allowed moisture to pass out but prevented moisture uptake by the seeds. Heat caused a permanent, irreversible opening of the hilar fissure, which rendered the seed permeable to water. However, the seedcoat itself remained impermeable to moisture even after heating. This mechanism likely accounts for the abundant germination of ceanothus species that often occurs after fire on both chaparral and forest sites (Conard and others 1985).

In the laboratory, germination has been induced by soaking in hot water or heating in an oven, with or without a subsequent period of cold stratification (table 4). The typical pattern is that germination increases with the temperature of heat treatments up to a maximum, at which point seed mortality begins to occur. Seed germination and mortality are a function of both temperature and length of exposure, but for most species these optima are poorly defined. For hoaryleaf ceanothus, for example, maximum germination was obtained after heat treatments of 10 minutes to 1 hour at

C

Table 3—*Ceanothus,* ceanothus: thousands of cleaned seeds per weight

	Range		Average		
Species	**/kg**	**/lb**	**/kg**	**/lb**	**Samples**
C. americanus	212–291	96–132	247	112	5
C. arboreus	106–110	48–50	108	49	2
C. cordulatus	311–396	141–179	366	166	4
C. crassifolius	73–143	33–65	117	53	3
C. cuneatus	80–123	36–56	108	49	3
C. cuneatus var. *rigidus*	—	—	159	72	1
C. diversifolius	—	—	185	84	1
C. greggii	—	—	51	23	—
C. impressus	—	—	245	111	1
C. integerrimus	128–179	58–81	154	70	2
C. oliganthus	137–161	62–73	148	67	2
C. prostratus	82–98	37–45	90	41	3
C. sanguineus	282–291	128–132	287	130	2
C. sorediatus	267–269	121–122	—	—	2
C. thyrsiflorus	106–400	48–181	—	—	—
C. velutinus	135–335	61–152	207	94	5

Sources: Emery (1964), Hubbard (1958), Mirov and Kraebel (1939), Plummer and others (1968), Quick (1935), Quick and Quick (1961), Reed (1974), Swingle (1939).

70 to 80 °C. At higher temperatures, germination dropped off increasingly rapidly with duration of treatment, until at 100 °C there was a linear decrease in germination with times over 5 minutes (Poth and Barro 1986). In the wild, this range of time and temperature optima gives the advantage of allowing dormancy to be broken at a range of soil depths as a function of fire temperature and residence times. Quick and Quick (1961) reported that germination of mountain whitethorn and, to a lesser extent, deerbrush ceanothus began to drop off rapidly after a few seconds to several minutes in boiling water. Although "steeping" treatments at cooler temperatures (for example, 70 to 95 °C) were also found effective on several species (Quick 1935; Quick and Quick 1961), many investigators have continued to use treatments of boiling water (table 4). Dry heat treatments may be less damaging at higher temperatures than wet heat (table 4), although careful comparisons have not been made. In place of hot water treatments, seeds can also be immersed in sulfuric acid for 1 hour (Reed 1974).

Seeds of species found at high elevations also require cold stratification for good germination (Quick 1935; Van Rensslaer and McMinn 1942). Although some lower-elevation species from chaparral sites can germinate reasonably well without this cold treatment, their germination rates generally increase with stratification (table 4). Cold stratification is accomplished by storing seeds in a moist medium for periods of 30 to 90 days at temperatures of 1 to 5 °C. In general, longer periods of cold stratification are more effective than short ones. For example, Radwan and Crouch (1977) observed increasing germination of redstem ceanothus as cold stratification was increased from 1 to 3 or 4 months; no germination occurred without stratification. Similar patterns were observed by Quick and Quick (1961) for deerbrush ceanothus (increased germination up to 2 months of stratification) and Bullock (1982) for mountain whitethorn (increased germination up to 3 months). In lieu of cold stratification, a chemical treatment with gibberellin and thiourea was used to induce germination of buckbrush ceanothus (Adams and others 1961). Treatment with potassium salts of gibberellin also sucessfully replaced cold stratification in germination tests on redstem ceanothus seeds (Radwan and Crouch 1977). Following chemical treatments, seeds may then be germinated or dried again and stored (Adams and others 1961). Although emphasis has been on more natural methods of stimulating germination, seeds of snowbrush ceanothus and other species can be germinated quite successfully with acid scarification followed by a gibberellin treatment (Conard 1996).

Specific germination test conditions have not been well defined for most species of ceanothus. Sand or a mixture of sand and soil has been used as the moisture-supplying medium in most of the reported germination tests (Emery 1964; Quick 1935; Reed 1974), but filter paper has also been used successfully (Keeley 1987a). Diurnally alternating tempera-

Table 4—*Ceanothus*, ceanothus: pregermination treatments and germination test results

	Pregermination treatments				Germination rate	
	Hot water soak		Cold			
Species	Temp (°C)	Time* (min)	stratification (days)	Germination test days	Avg (%)	Samples
C. americanus	—	0	90	50	65	4
	77–100	ttc	60	30	32	1
C. arboreus	71–91	ttc	0	40–112	90	3+
C. cordulatus	90	ttc	94	35	74	4
	85	ttc	94	35	90	4
	80	ttc	94	35	74	4
C. crassifolius	71	ttc	90	21–90	76	1+
	71	ttc	0	90	48	1+
C. cuneatus	71	ttc	90	21–90	92	1+
	120†	5	30	21	28	3
	100	5	30	21	38	3
	70	60	30	21	3	3
	—	0	30	21	10	3
C. cuneatus var. *rigidus*	71	ttc	0	60–112	85	2+
C. diversifolius	77–100	ttc	60	60	61	1+
C. fendleri	—	0	0	—	16	—
C. greggii	100	1	30–60	17	51	—
C. impressus	77–100	ttc	60	30	73	1+
C. integerrimus	85	ttc	56	—	100	1
	71	ttc	90	20	85	1+
C. leucodermis	120†	5	30	21	68	3
	100	5	30	21	50	3
	70	60	30	21	47	3
	—	0	30	21	7	3
C. megacarpus	120†	5	30	21	88	3
	100	5	30	21	53	3
	70	60	30	21	54	3
	—	0	30	21	6	3
C. oliganthus	71	ttc	0	70	62	1+
C. prostratus	100	0.5	115	—	92	—
	77–100	ttc	90	30	71	1+
C. sanguineus	100	1	120	32	97	3
	100	5	120	32	92	3
	100	15	120	32	41	3
	100	1–5	0	32	0	3
C. sorediatus	100	5	90	30	100	1+
	100	5	0	30	38	1
C. thyrsiflorus	71	ttc	90	60	83	1+
	71	ttc	0	60	73	1
C. velutinus	90	ttc	63–84	—	82	1
	71	ttc	90	30	70	2+

Sources: Emery (1964), Keeley (1987a), Mirov and Kraebel (1939), Quick (1935), Quick and Quick (1961), Radwan and Crouch (1977), Reed (1974), Van Dersal (1938).
* ttc = "time to cool" (to room temperature) varied from several hours to overnight.
† Results reported here are for dry heat treatments, with germination in the dark; see Keeley (1987) for data on light germination.

tures of 30 °C in light and 20 °C in darkness have been effective, but constant temperatures of 10 °C (Reed 1974) and 24.5 °C (Emery 1988) also have been suitable for germination. A need for light has not been reported (Keeley 1991), and at least 1 species (deerbrush ceanothus) appears to germinate significantly better in the dark (Keeley 1987a). Germination rates resulting from selected pregermination treatments are listed in table 4 for 19 species.

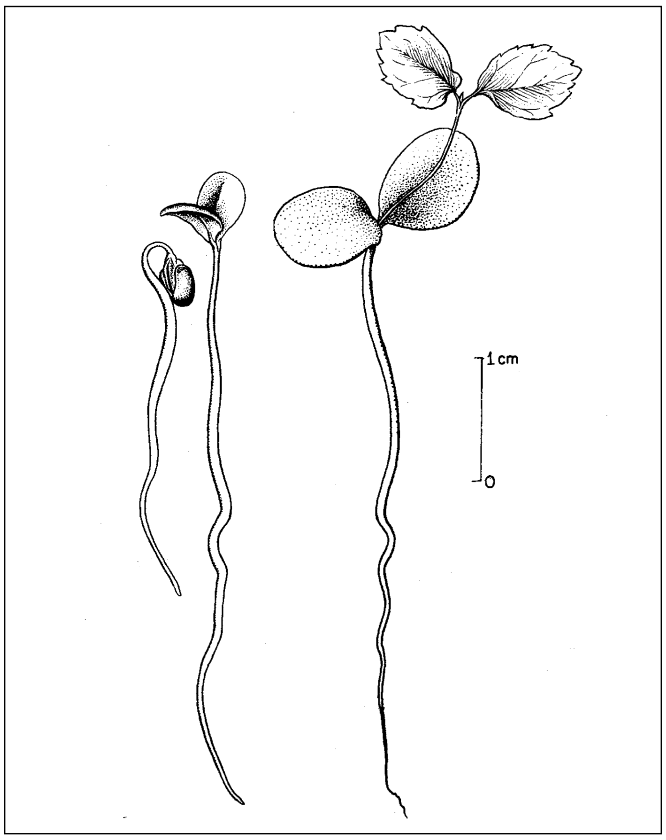
The genus includes both species that sprout vegetatively following fire (sprouters) and species that are killed by fire and reproduce only from seeds (obligate seeders). Obligate seeders appear to have overall higher germination following heat treatment and to tolerate higher temperatures and longer periods at high temperature without damage to seed viability (Barro and Poth 1987). Germination test results suggest that eastern species may not be dependent on fire to stimulate germination. For western species, however, some level of heat treatment, followed by stratification, will typically enhance germination. Although there has certainly been considerable variability in test results (table 4), a 5- to 10-minute dry heat treatment at 100 °C or a steeping treatment starting with 85 °C water, followed by several months of cold stratification, should effectively stimulate germination in most species.

Nursery practice. Seeding has been done in flats containing a medium of 5 parts loam, 4 parts peat, and 3 parts sand (Van Rensslaer and McMinn 1942). Leaf-mold may be substituted for the peat, but the peat is preferred because it is comparatively free of fungi. Sand is needed for drainage, a higher proportion being used in the seeding than in the potting medium. Seedlings are sensitive to sowing depth. In a trial by Adams (1962), deerbrush and buckbrush ceanothuses emerged best when sown at depths of 12 to 25 mm ($^1/_2$ to 1 in), and shading favored emergence of the first 2 species. However, some germination and emergence occurred at sowing depths ranging from 6 to 64 mm ($^1/_4$ to $2^1/_2$ in). Many species are sensitive to damping off, so for safety soil should be sterilized (Van Rensslaer and McMinn 1942). In California, seeding is usually done in November to January. Germination is epigeal (figure 4). Although all species of *Ceanothus* apparently fix nitrogen symbiotically, there has apparently been little or no research into the efficacy of or need for seed inoculation with *Frankia* to ensure nodulation of seedlings after outplanting. This is not likely to be a problem on soils where *Ceanothus* species are present, as nodulation appears to occur readily (Conard 1996) but may be of concern for horticultural uses of the genus.

Seedling care. When several sets of leaves have formed, the seedlings can be carefully planted into 2- or 3-inch (5- or 7.6-cm) pots. A good potting medium is 5 parts loam, 3 parts peat or leaf mold, and 1 part sand (Van Rensslaer and McMinn 1942). Care must be taken not to place the seedlings too deep in the soil, with root crowns should be just below the soil surface. Seedlings are susceptible to stem rot, and the loss will be greater if young plants are kept in moist soil that covers the root crown. The root development should be examined from time to time. When a loose root system has formed on the outside of the ball, the plant is ready for shifting to a larger pot or gallon can. It is best to discard potbound plants rather than to carry them along.

Planting stock of most common western ceanothus species is now available from commercial nurseries or botanic gardens, and numerous hybrids and cultivars have been developed for the nursery trade. Cultural notes on some of the commonly available species (table 1) and cultivars (Brickell and Zuk 1997) follow:

Figure 4—*Ceanothus americanus*, New-Jersey- tea: seedling development at 1, 5, and 15 days after germination.



- feltleaf ceanothus—*C. arboreus*—which can attain a height of 5 to 8 m and has pale blue flowers, grows best in coastal areas or with partial shade in areas with hot, dry summers.
- Fendler ceanothus—*C. fendleri*—up to 2 m tall with pale, bluish-white flowers, has been propagated from seeds sown in the spring and from cuttings in autumn. It grows best in light, well-drained soils and can tolerate cold.
- Carmel ceanothus—*C. griseus* var. *horizontalis* McMinn—a spreading, low-growing (to 1 m) variety, is used as ground cover and for slope stabilization. It performs best in mild coastal regions but will do well in partial shade in drier areas with adequate watering. Several named varieties are available.
- prostrate ceanothus—*C. prostratus*—a spreading, prostrate groundcover with small blue flower clusters, usually is propagated by layering. It is best if grown within its native range (for example, ponderosa pine zone of Sierra Nevada) and does not grow well at low elevations.

- Monterey ceanothus—*C. cuneatus* (Nutt.) Hoover var. *rigidus* cv. Snowball—a white-flowered cultivar, 1 to 1.5 m tall, recommended for coastal areas from southern California to the Pacific Northwest. Summer water should be restricted. It is a good bank and background plant.
- Sierra blue—*C. cyaneus* Eastw.—a medium to large shrub (to 6 m) with showy blue flowers, is a relatively fast grower that will tolerate hot, dry environments with some supplemental summer water.
- blueblossom—*C. thyrsiflorus*—a large shrub (2 to 7 m tall) with showy deep blue flowers, is a native of coastal forests. It grows well in its native range (Pacific coastal mountains) and needs shade from afternoon sun on dry inland sites, but requires little summer water once established.

There are many more ceanothus varieties that are excellent candidates for a range of domestic, commercial, or right-of-way landscaping situations. Although they are typically not widely available at retail nurseries, many native plant nurseries within the native range of ceanothus have wide selections. Additional information can be found in Kruckeberg (1982), Lenz and Dourley (1981), Perry (1992), Schmidt (1980), and the Sunset Western (1995) and National (1997) Garden Books, among others.

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Pinaceae—Pine family

Cedrus Trew

cedar

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Growth habit, occurrence, and use. The genus of true cedars—*Cedrus*—consists of 4 (or fewer) closely related species of tall, oleoresin-rich, monoecious, coniferous, evergreen trees, with geographically separated distributions (Arbez and others 1978; Bariteau and Ferrandes 1992; Farjon 1990; Hillier 1991; LHBH 1976; Maheshwari and Biswas 1970; Tewari 1994; Vidakovié 1991). The cedars are restricted to the montane or high montane zones of mountains situated roughly between 15°W and 80°E and 30 to 40°N (Farjon 1990). This discontinuous range is composed of 3 widely separated regions in North Africa and Asia (Farjon 1990): the Atlas Mountains of North Africa in northern Morocco and northern Algeria; Turkey, the mountains on Cyprus, and along the eastern border of the Mediterranean Sea in Syria and Lebanon; the Hindu Kush, Karakoram, and Indian Himalayas. The 4 species of cedars (table 1) are so closely related that habitual characteristics help differentiate the species (Farjon 1990). Isozyme analysis of cedar diploid tissue raises questions about the separation of Atlas cedar and cedar of Lebanon into 2 distinct species, because no distinguishing gene marker was detected (Panetsos and others 1992). There is disagreement as to the exact taxonomic status of the various cedars, with some authors suggesting that they be reduced to only 2 species: deodar cedar and cedar of Lebanon. In this writing, we will examine all 4 species.

The cedars are both valuable timber trees and quite striking specimen plants in the landscape. The wood of cedar of Lebanon is fragrant, durable, and decay resistant; and on a historical note, the ancient Egyptians employed cedar sawdust (cedar resin) in mummification (Chaney 1993; Demetci 1986; Maheshwari and Biswas 1970). Upon distillation of cedar wood, an aromatic oil is obtained that is used for a variety of purposes, from scenting soap to medicinal practices (Adams 1991; Chalchat and others 1994; Maheshwari and Biswas 1970; Tewari 1994).

Atlas cedar is a large tree that grows rapidly when young and is closely related to cedar of Lebanon. The Atlas cedar is distinguished by a taller crown, less densely arranged branchlets, bluish green leaves (needles) that vary from light green to silvery blue, smaller cones, and smaller

Table 1—*Cedrus,* cedar: nomenclature, occurrence, height at maturity, and date first cultivated

Scientific name	Common name	Occurrence	Height at maturity (m)	Year first cultivated
C. atlantica **(Endl.) G. Manetti ex Carriere**	**Atlas cedar**	In Algeria on Mts. Babor & Tababort & in Hodna Mtns; in Morocco in Rif Mtns (at 1,370–2,200 m); planted in US	9–40	Before 1840
C. breviifolia **(Hook. f.) A. Henry**	**Cyprian cedar**	Two separate locations on Mt Paphos in western Cyprus (at 900–1,525 m)	8–24	1879
C. deodara **(Roxb. ex D. Don) G. Don F.**	**deodar cedar**	E Afghanistan (Hindu Kush), NW Pakistan (Karakoram), NW India (Kashmir & Gharwal Himalaya), rare in Nepal (1,700–3,000 m in western range & 1,300–3,300 m in eastern range); planted in US	15–50	1831
C. libani **A. Rich.**	**cedar of Lebanon**	In S Turkey (Taurus Mtns), also Syria (Djebel el Ansiriya) & Lebanon (Djebel Loubnan); disjunct relict population in N Turkey near Black Sea (at 1,300–3,000 m); planted in US	15–40	Pre-1650

Sources: Dirr (1990), Farjon (1990), Hillier (1991).

seeds (table 2) (Dirr 1990; Farjon 1990; Hillier 1991; Loureiro 1990, 1994). Young trees appear stiff, with an erect leader and a pyramidal overall shape; with maturity this species assumes a flat-topped habit with horizontally spreading branches (Dirr 1990). Atlas cedar is hardy in USDA zones 6 to 9, with several beautiful cultivars that differ in color and characteristic habit (Dirr 1990; Hillier 1991; Vidakovié 1991). Of special note is '*Glauca*' (f. *glauca*), with very blue to silvery blue leaves, which is a spectacular specimen tree (Dirr 1990; Hillier 1991).

Cyprian cedar is a rare species that grows slowly but eventually develops into a medium-sized tree. This species is distinguished from cedar of Lebanon only by its habitual form and shorter leaves (table 2) and the broad and umbrella-shaped crown on older specimens (Farjon 1990; Hillier 1991; Vidakovié 1991).

Deodar cedar is an excellent specimen tree. The deodar cedar is broadly pyramidal when young, with gracefully pendulous branches (Dirr 1990; Tewari 1994). It is distinguished from the other species by its drooping leader and longer leaves (table 2) (Hillier 1991). Multi-stemmed crowns occasionally evolve from the higher branches turned erect, but the crown seldom becomes flat-topped, remaining conical or pyramidal (Farjon 1990). Deodar cedar is hardy in USDA zones 7 to 8, but young trees are prone to injury from frosts and cold wind (Dirr 1990). There are many cultivars of deodar cedar, but 2 worth mentioning are 'Kashmir' and 'Shalimar'. The former is winter-hardy—it tolerates cold winters to –30 °C—with silvery blue-green foliage (Dirr 1990; Vidakovié1991). The latter displays good blue-green leaf color and is the hardiest cultivar planted in the United States (Dirr 1990; Koller 1982).

Cedar of Lebanon is a majestic tree with innumerable historical and biblical references. It has a thick, massive trunk and wide-spreading branches; it is pyramidal when young but develops a flat-topped crown and horizontally tiered branches when mature (Chaney 1993; Dirr 1990; Farjon 1990; Hillier 1991). The dark green foliage, stiff habit, and rigidly upright cones (table 2) give this tree its splendor for landscape specimen planting. The morphological differences between cedar of Lebanon and Atlas cedar are small and not entirely constant (Farjon 1990; Maheshwari and Biswas 1970). Cedar of Lebanon is hardy in USDA Zones 5 to 7 (Dirr 1990; Dirr and others 1993). A geographical form—*C. libani* ssp. *stenocoma* (Schwarz) Davis—differs from the typical Lebanon cedar in having a broadly columnar habit and needle and cone characteristics that are intermediate between Atlas cedar and cedar of Lebanon; it is also more cold-hardy (Hillier 1991; Vidakovié 1991). There are also several dwarf cultivars of cedar of Lebanon that are of interest for use in the landscape (Hillier 1991; Vidakovié 1991).

Flowering and fruiting. The male flowers of cedar are erect catkins, up to 5 cm in length, whereas the female flowers are erect, cone-like inflorescences, 1 to 1.5 cm long, surrounded by needles at the base (Vidakovié 1991). Male and female strobili of the true cedars are borne (usually) on the same tree, but on separate branches (Farjon 1990; Maheshwari and Biswas 1970; Rudolf 1974). The male cones are solitary, grow more or less erect from the short shoots, and bear abundant yellow pollen (Farjon 1990; Maheshwari and Biswas 1970). Depending upon the altitude, locality, and weather, the pollen is shed late in the year (September through November), relating to the late development of the female strobilus (Farjon 1990; Maheshwari and Biswas 1970). The female cones are borne singly at the tips of the dwarf shoots, stand erect, and are less abundant than the male cones (Farjon 1990; Maheshwari and Biswas 1970). Although pollination takes place in the fall, the cones do not mature until the second year, requiring about 17 to 18 months for full development (Farjon 1990; Maheshwari and Biswas 1970; Rudolf 1974).

The mature, barrel-shaped cones (figure 1) are resinous and characterized by numerous closely appressed, very broad scales, each containing 2 seeds (table 2) (Rudolf 1974). The scales are attached to the persistent rachis with a narrowed, petiolate base and dismember from it by abscission at maturity, as in fir (*Abies*) (Farjon 1990; Rudolf 1974). The irregularly triangular mature seed is rather soft and oily, with resin vesicles present on each side of the seed,

Table 2—*Cedrus,* cedar: cone, seed, and leaf (needle) characteristics

	Cone characteristics			Seed size		Leaf characteristics	
Species	Ripe color	Length (cm)	Width (cm)	Length (mm)	Width (mm)	Length (cm)	No. in whorls
C. atlantica	Light brown	5–8	3–5	8–13	4–6	1–2.5	20–45
C. brevifolia	Light brown	5–10	3–6	8–14	5–6	0.5–1.6	15–20
C. deodara	Reddish brown	7–13	5–9	10–15	5–7	2–6.0	20–30
C. libani	Grayish brown	8–12	3–6	10–14	4–6	1–3.5	20–40

Sources: Farjon (1990), Rudolf (1974), Vidakovie (1991).

and it has a membranous, broad wing that is several times larger than the seed (figures 2 and 3) (Farjon 1990; Rudolf 1974). Seeding habits of the various species are given in table 3. Commercial seed bearing of deodar cedar begins from 30 to 45 years of age, and good seedcrops are borne every 3 years, with light crops in the intervening years (Doty 1982; Maheshwari and Biswas 1970; Rudolf 1974; Tewari 1994; Toth 1979).

Collection of fruits; extraction, cleaning, and storage of seeds. Cones should be collected directly from the trees, before the cones turn brown, or cone-bearing twigs may be cut from standing or felled trees just before ripening is complete (Dirr and Heuser 1987; Rudolf 1974; Singh and others 1992). One cubic meter (28.38 bu) of cones weighs from 12.2 to 15.9 kg (27 to 35 lb) and yields about 1.4 kg (3 lb) of cleaned seeds (Rudolf 1974). Cones should be allowed to dry until the scales loosen and the seeds can be removed (Dirr and Heuser 1987; Macdonald 1986; Toth 1980a). It is important to avoid any more drying than is absolutely necessary, because the seeds may be killed. Cones of cedar may be soaked in warm water for 48 hours to encourage them to disintegrate (Rudolf 1974; Macdonald 1986). Freezing moist cones (as a last resort) will also force the scales to open up (Macdonald 1986). After the cone scales are dry, they can be placed in a cone shaker to remove the seeds (Rudolf 1974), and seeds can be separated from the debris by fanning or sieving (Macdonald 1986). Seeds are de-winged by simply rubbing them in a dry cloth (Macdonald 1986), for resin from the resin pockets in the wings can make de-winging with bare hands difficult (Macdonald 1986). Purity of commercially cleaned seed has been 85 to 90% (table 4).

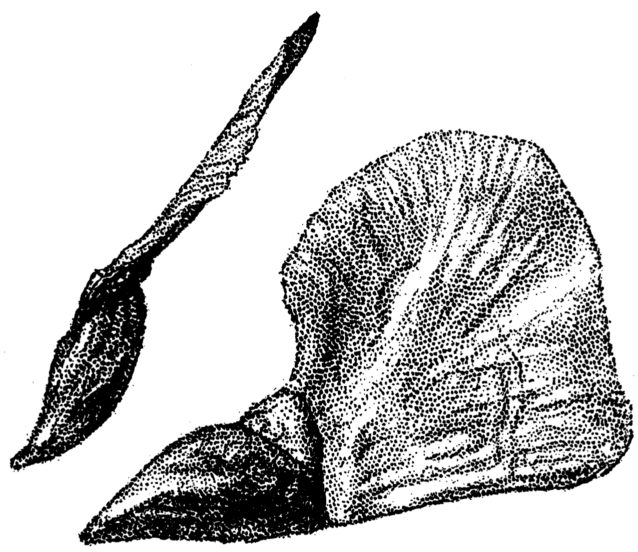
Even though cedar seeds are orthodox in storage behavior, they are very oily and do not keep well under many storage conditions (Allen 1995; Rudolf 1974). If cedar seeds are dried below a critical level, they will not imbibe water in a way that will allow the food reserves to be used by the embryo (Macdonald 1986). Cedar seeds have retained viability for 3 to 6 years when dried to a moisture content of less than 10%, placed in sealed containers, and held at temperatures of –1 to –5 °C (Erkuloglu 1995; Rudolf 1974).

Pregermination treatments. Cedar seeds exhibit little or no dormancy and will germinate without pretreatment. However, variable degrees of dormancy may be observed within a single lot of seeds (Dirr and Heuser 1987). Seeds should be stratified at 3 to 5 °C for 2 weeks (6.5 weeks for Cyprian cedar) to give more uniform germination (Allen 1995; Rudolf 1974). Thapliyal and Gupta (1980) found that 9 °C was a better temperature for stratification than 3 °C. Deodar cedar and cedar of Lebanon seeds are prone to damping-off disease and thus should be treated with an appropriate fungicide (Mittal 1983).

Germination tests. The AOSA (1993) prescribes germination tests of stratified seeds (14 days) on top of blotters for 3 weeks at 20 °C for all cedars (see also Toth 1980a). ISTA (1993) rules, however, specify diurnally alternating

Figure 1—*Cedrus libani*, cedar of Lebanon: mature cone.

Figure 2—*Cedrus libani*, cedar of Lebanon: seeds with membranous wing attached.



C

temperatures of 20 °C (night) and 30 °C (day) for a period of 4 weeks. Tests may also be made in sand flats (Rudolf 1974). Deodar cedar seeds stratified at 4 °C in moist sand for 30 days showed 45% germination versus 11% without stratification (Dirr and Heuser 1987). Thapliyal and Gupta (1980) also found that the percentage of germination without stratification to vary from 16 to 69%. Singh and others (1992) found that seeds from larger cones exhibited higher germination (66%) in Himalayan cedar. Singh and others (1997) also found that there were significant differences between tree-diameter classes in fresh and dry weight of seeds and also in germination in the laboratory and in the nursery. Germination of cedar seed is epigeal (figure 4).

Nursery practice and seedling care. Deodar cedar seeds should be sown in the fall (or in spring) at a rate of 200 to 250 seeds/m^2 (19 to 23/ft^2), in drills 10 to 15 cm (4 to 6 in) apart for lining-out stock and for root stocks (Macdonald 1986; Rudolf 1974). Chandra and Ram (1980) recommend sowing deodar seeds at a depth of 1 cm (0.4 in); further increase in depth results in decreased germination. Al-Ashoo and Al-Khaffaf (1997) reported that the best treatment for germination of cedar of Lebanon seeds was a 1.5-cm (0.6-in) sowing depth, with a covering medium of clay or alluvial soil. In northern areas, fall-sown beds should be mulched over winter, the mulch removed early in the spring, and the bed racks covered with burlap on critical spring nights to prevent freezing (Heit 1968). Cedar seeds can be sown in containers in the fall, transplanted into other

Table 3—*Cedrus,* cedar: phenology of flowering and fruiting

Species	Flowering	Cone ripening	Seed dispersal
C. atlantica	June–Sept	Sept–Oct	Fall–spring
C. deodara	Sept–Oct	Sept–Nov	Sept–Dec
C. libani	June–Sept	Aug–Oct	Fall–spring

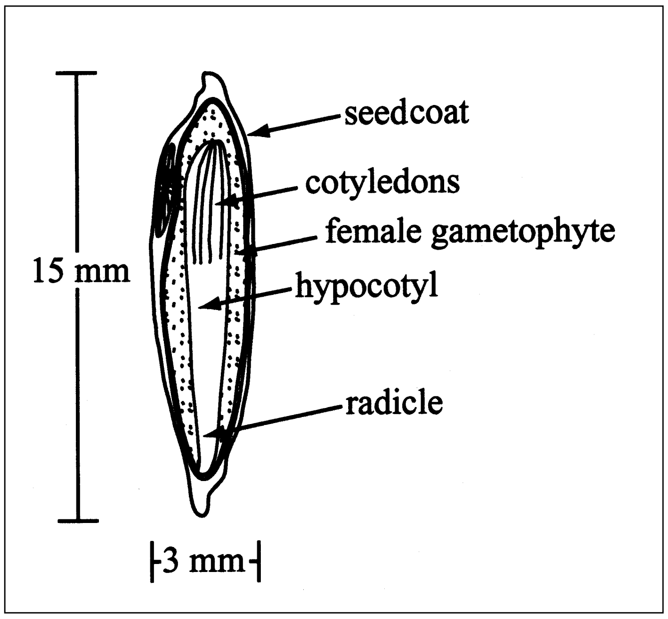
Sources: Rudolf (1974), Vidakovic (1991).

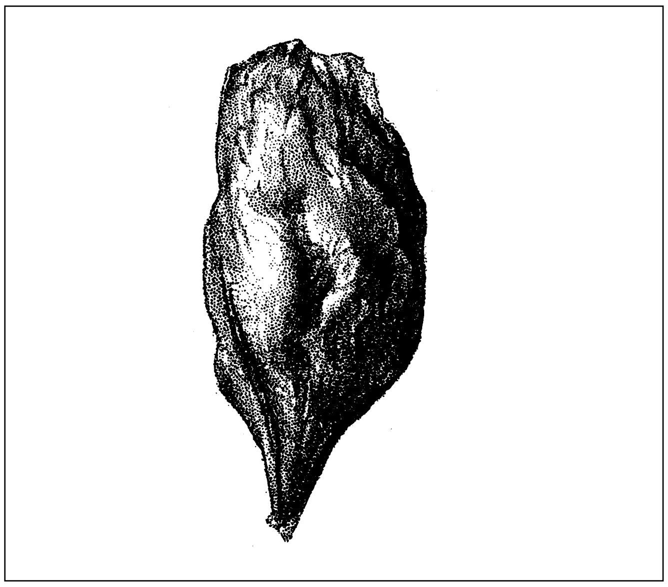
Table 4—*Cedrus,* cedar: seed data

Species	Avg no. cleaned seeds /kg	/lb	Commercial seed purity (%)
C. atlantica	13,900	6,300	89
C. brevifolia	13,000	5,890	—
C. deodara	8,150	3,700	85
C. libani	11,700	5,300	87
C. libani ssp. *stenocoma*	17,600	8,000	—

Sources: Allen (1995), Rudolf (1974).

Figure 3—*Cedrus brevifolia*, Cyprian cedar: longitudinal section through a seed (**top**) and exterior view of a de-winged seed (**bottom**).

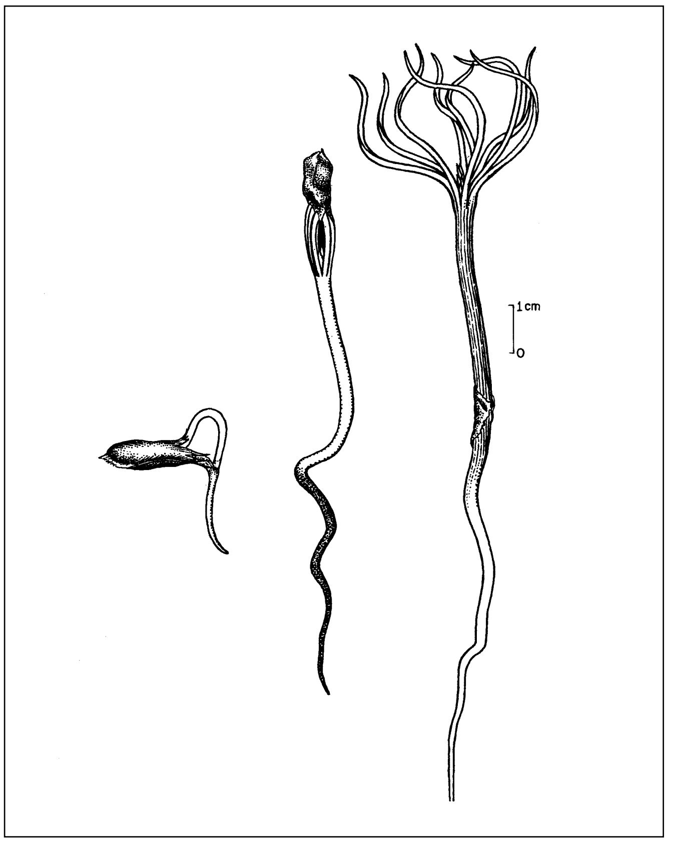




containers during the winter, and kept in shaded beds in the summer to produce 1/2- to 1 1/2-year-old planting stock (Rudolf 1974). The size of the propagation container, growth media, transplanting date, and handling of seedlings is important in container or field grown stock (Appleton and Whitcomb 1983; Burger and others 1992; Doty 1982; Guehl and others 1989; Puxeddu and Alias 1991; Toth 1980b).

Deodar cedar 'Shalimar' can be propagated by collecting cuttings in late fall to early winter; 67% of such cuttings given a quick dip in 5 g/liter (5,000 ppm) indole-3-butyric acid (IBA) solution and placed in a sand–perlite medium with bottom heat (Nicholson 1984) rooted. Shamet and Bhardwaj (1995) reported 69% rooting of deodar cedar cut-

Figure 4—*Cedrus libani*, cedar of Lebanon: seedling development at 1, 4, and 8 days after germination.



tings treated with 0.5% indole-3-acetic acid–talc or 1% naphthaleneacetic acid–activated charcoal, both supplemented with 10% captan and 10% sucrose. However, cuttings taken from Atlas cedar and cedar of Lebanon are difficult to root, although some rooting may occur on cuttings taken in late winter and treated with 8 g/liter (8,000 ppm) IBA–talc (Dirr and Heuser 1987). Cultivars of cedar species are more routinely propagated by grafting (Blomme and Vanwezer 1986; Dirr and Heuser 1987; Hartmann and others 1990; Lyon 1984; Macdonald 1986; Richards 1972). Two reports have been published on the *in vitro* culture of deodar cedar (Bhatnagar and others 1983; Liu 1990). A method for *in vitro* propagation of cedar of Lebanon through axillary bud production, a study of bed dormancy *in vitro*, and detection of genetic variation of *in vitro*–propagated clones has also been described (Piola and Rohr 1996; Piola and others 1998, 1999).

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Celastraceae—Bittersweet family

Celastrus scandens L.

American bittersweet

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Other common names. climbing bittersweet, shrubby bittersweet.

Growth habit, occurrence, and use. American bittersweet is a deciduous climbing or twining shrub of eastern North America (Brizicky 1964; Fernald 1950) that occurs in thickets, in stands of young trees, along fence rows, and along streams, usually in rich soil. It occurs naturally from southern Quebec; west to southern Manitoba; and south to Oklahoma and central Texas, Arkansas, Tennessee, northern Alabama, and western North Carolina (Brizicky 1964). Some authors (Fernald 1950; USDA FS 1948) reported it in Louisiana, New Mexico, Georgia, and Mississippi, but its occurrence has not been verified in Georgia, Louisiana, or Mississippi (Brizicky 1964).

The plant is valuable for ornamental purposes and game food and cover; the bark has been used for medicinal purposes (USDA FS 1948). Among the animals and birds feeding on American bittersweet are the bobwhite quail (*Colinus virginianus*), ruffed grouse (*Bonasa umbellus*), ringneck pheasant (*Phasianus colchicus*), eastern cottontail (*Silvilagus floridanus*), fox squirrel (*Sciurus niger*), and various songbirds (Van Dersal 1938). It was introduced into cultivation in 1736 (USDA FS 1948).

By 1970, oriental or Asiatic bittersweet—*C. orbiculatus* Thunb.—had become naturalized on at least 84 sites from Georgia to Maine and west to Iowa (McNab and Meeker 1987), occupying many of the same sites as American bittersweet. It is listed as an invasive plant by the United States Government (USDA NRCS 1999). In some locales, the species is found mainly along fence lines, resulting from the germination of seeds contained in the droppings from frugivorous birds (McNab and Meeker 1987). The stem, leaves, and berries of oriental bittersweet are reported to be toxic for human consumption (McNab and Meeker 1987).

Flowering and fruiting. The small greenish, polygamo-dioecious or dioecious flowers open from May to June and are borne in raceme-like clusters at the end of branches (Brizicky 1964; Fernald 1950). Hymenopterous insects, especially bees, seem to be the main pollinators, although wind may also be involved (Brizicky 1964). Seeds are about 6.3 mm long and are borne in bright orange to red, fleshy arils, 2 of which are usually found in each of the 2 to 4 cells composing the fruit, a dehiscent capsule (figure 1). The yellow to orange capsules ripen from late August to October. They split open soon thereafter, exposing the seeds covered with showy red arils (figures 2 and 3). Good seedcrops are borne annually and may persist on the bushes throughout much of the winter (USDA Forest Service 1948). In Pennsylvania, only 1 seedcrop failure was reported in a 14-year period (Musser 1970). Sunlight is reported necessary for abundant fruiting to occur (Musser 1970).

Collection of fruit. The ripe fruits should be collected as soon as the capsules separate and expose the arils, or from about mid-September as long as they hang on the vines (USDA FS 1948), but rarely later than December (Van Dersal 1938). In Pennsylvania, the fruits are collected from late October through November (Musser 1970).

Figure 1—*Celastrus scandens*, American bittersweet: fruiting branch.

Extraction and storage of seeds. Collected fruits should be spread out in shallow layers and allowed to dry for 2 or 3 weeks (USDA FS 1948). In Pennsylvania, the fruits are allowed to air-dry for 1 week in shallow trays (Musser 1979). The seeds are then removed from the capsules by flailing or running the fruits through a hammermill or macerator with water (Musser 1970; USDA FS 1948). Then the seeds are allowed to dry for another week and the chaff is separated by windmilling (Musser 1979). The dried arils are left on the seeds (USDA FS 1948) except when seeds are to be stored.

American bittersweet has 4 to 8 seeds/fruit. On the basis of 10 samples, the number of seeds per weight ranged from 26,000 to 88,000/kg (12,000 to 40,000/lb) with an average of 57,000/kg (26,000/lb). Average purity was 98% and average soundness 85% (USDA FS 1948).

In Pennsylvania, the seeds usually are sown in the fall soon after collection and extraction or stored in cloth bags until used (Musser 1970). For longer storage periods, viability has been retained for 4 to 8 years by cleaning the fleshy material from the seeds, air-drying the seeds at low humidity, and then storing them in sealed containers at a temperature between 1 and 3 °C (Heit 1967).

Pregermination treatments. Seeds of American bittersweet have a dormant embryo and thus require after-ripening for germination. There is also some evidence that the seedcoat may have an inhibiting effect on germination (Hart 1928; USDA FS 1948). Good germination is obtained

Figure 2—*Celastrus scandens*, American bittersweet: seeds with aril removed.

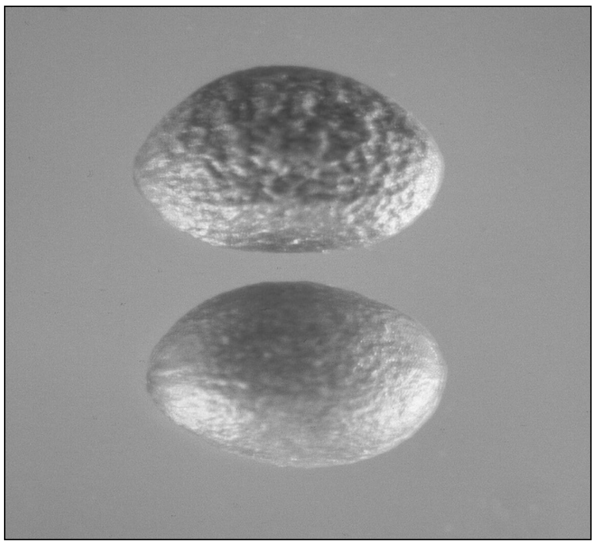
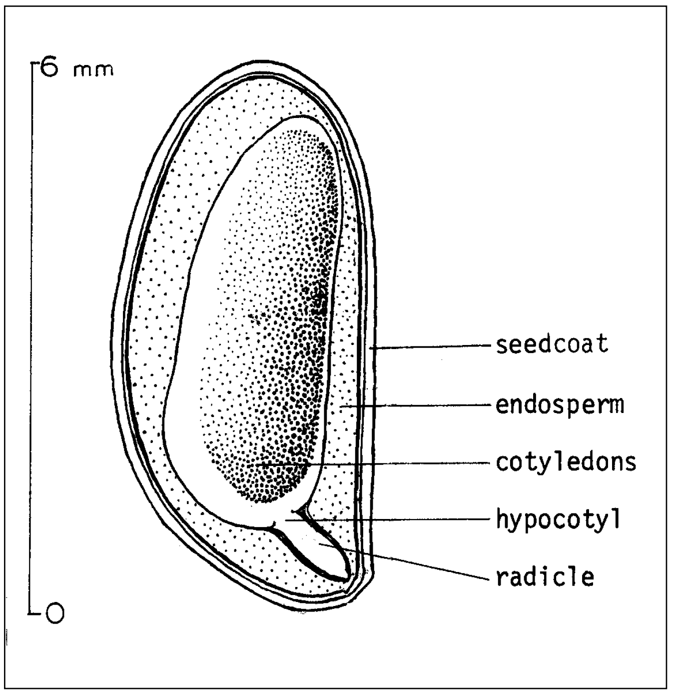


Figure 3—*Celastrus scandens*, American bittersweet: longitudinal section through a seed.

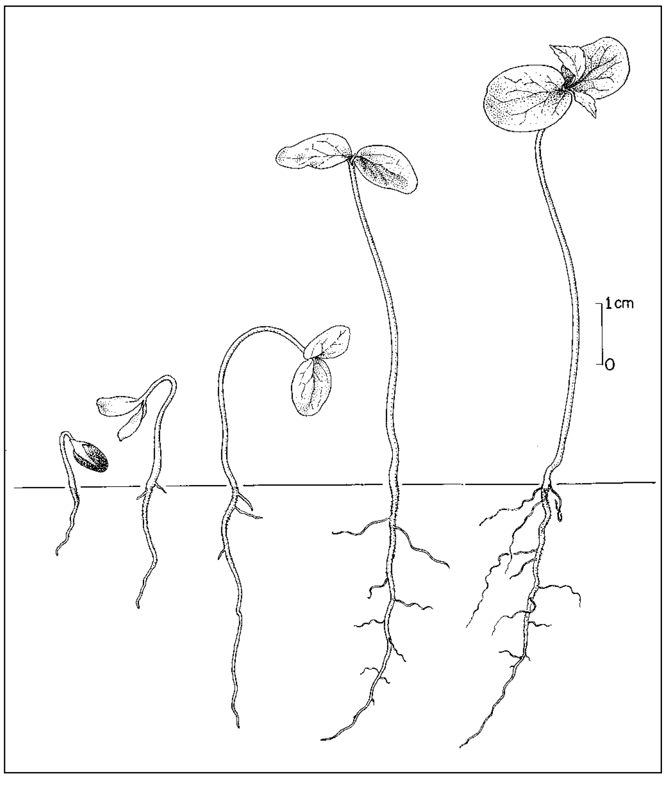


by fall-sowing or by stratification in moist sand or peat for 2 to 6 months at 5 °C (Heit 1968; Musser 1970; USDA FS 1948). Three months of cold stratification has resulted in good germination for American bittersweet (Dirr and Heuser 1987). It seems to make little difference whether cleaned seeds or dried fruits are sown; however, it appears that both cleaned seeds and fruits should be dried at room temperature for 2 to 3 weeks before they are sown (USDA FS 1948).

Germination tests. On the basis of 6 tests, using stratified seedlots in sand flats, at temperatures alternating from 10 to 25 °C, germinative capacity was found to range from a low of 9 to a high of 80% in 30 days, with an average of 47%. Potential germination varied from 9 to 93% (USDA FS 1948). Seedlots of oriental bittersweet showed 100% germination after 3 months of cold stratification (Dirr and Heuser 1987). Germination of American bittersweet is epigeal (figure 4).

A good estimate of germination can be obtained by the excised embryo method (Heit and Nelson 1941). The seeds are soaked until plump; seedcoats are removed and the embryos excised. The excised embryos are placed on moistened filter paper in covered petri dishes. A room temperature of 21 to 22 °C appears to be most satisfactory. Viable embryos will either show greening of the cotyledons, remain perfectly white in color but grow larger, or exhibit radicle elongation. Embryos exhibiting such characteristics can be counted as being from healthy seeds, capable of germinating

Figure 4—*Celastrus scandens,* American bittersweet: seedling development at 1, 2, 5, 10, and 30 days after germination.



with proper afterripening treatment. Five to 20 days are required to secure approximate germination by the excised embryo method.

Nursery practice. In Pennsylvania, good results have been obtained by sowing cleaned seeds in the first fall after collection and extraction. The seeds are broadcast on seedbeds and firmed in with a roller; then covered with a mixture of 1 part of sand to 2 parts of sawdust. The beds are covered with shade until germination occurs. Germination usually begins about 20 days after conditions become favorable (Musser 1970).

Another practice is to stratify cleaned or dried seeds in the pulp in January, and then sow them in the early spring. Young seedlings are somewhat susceptible to damping-off (USDA FS 1948). About 6,600 usable plants can be produced per kilogram of seeds (3,000/lb) (Van Dersal 1938). Propagation by root cuttings, layers, or stem cuttings is also sometimes practiced (Sheat 1948).

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C

Ulmaceae—Elm family

Celtis L.

hackberry

Franklin T. Bonner

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Growth habit, occurrence, and use. The genus *Celtis*—hackberry—is a large, widespread genus that includes about 70 species of shrubs and trees in the Northern Hemisphere. The 3 species listed in table 1 are all medium to large deciduous trees.

Flowering and fruiting. The small, greenish flowers of all 3 species appear in the spring as the new leaves emerge (table 2). These species are polygamo-monoecious (Krajicek and Williams 1990; Kennedy 1990; Vines 1960). Hackberry fruits are spherical drupes 6 to 13 mm in diameter with a thin pulp enclosing a single bony nutlet (figures 1–3). Good seedcrops are borne practically every year, and the fruits persist on the branches into winter (Bonner 1974; Krajicek and Williams 1990; Kennedy 1990). Other fruit and seed data (Little 1950; Preston 1947; Rehder 1940; Swingle 1939) are presented in tables 2 and 3.

Collection of fruits. Mature fruits can be picked by hand from trees as late as midwinter. Collection is much easier after all leaves have fallen (Bonner 1974). Limbs of sugarberry can be flailed to knock the fruits onto sheets spread under the trees. Fruits collected early in the season should be spread to dry to avoid overheating and molding (Williams and Hanks 1976). Fruits collected later in the season, unless the collection is done in wet, rainy weather, usually do not require additional drying (Bonner 1974).

Extraction and storage. Twigs and trash can be removed by screening or fanning, and the fruits can be depulped by wet or dry maceration. If wet maceration is used, the seeds will have to be dried for storage. If they are to be planted immediately, drying is not necessary. The pulp on dried fruits can be crushed and the resulting debris removed by washing on a screen under water pressure (Williams and Hanks 1976).

Table 1—*Celtis*, hackberry: nomenclature and occurrence

Scientific name & synonym	Common name(s)	Occurrence
C. laevigata **Willd.** *C. mississippiensis* Spach	**sugarberry,** sugar hackberry, hackberry, sugarberry, *palo blanco*	Maryland & Virginia to Florida & Texas; N to Kansas, Texas, Illinois, & Kentucky
C. laevigata **var. reticulata (Torr.) L. Benson** *C. reticulata* Torr.	**netleaf hackberry,** hackberry, western hackberry, *palo*	Washington & Colorado S to W Texas, S California, & central Mexico
C. occidentalis **L.** *C. crassifolia* Lam.	**common hackberry,** hackberry, sugarberry, northern hackberry	New England to North Dakota; S to NW Texas & N Georgia

Source: Little (1979).

Table 2—*Celtis*, hackberry: phenology of flowering and fruiting

Species	Flowering	Fruit ripening	Seed dispersal
C. laevigata	Apr–May	Sept–Oct	Oct–Dec
C. laevigata var. *reticulata*	Mar–Apr	Late fall	Fall–winter
C. occidentalis	Apr–May	Sept–Oct	Oct–winter

Source: Bonner (1974).

Figure 1—*Celtis*, hackberry: fruits (**left**) and seeds (**right**) of *C. laevigata*, sugarberry (**top**) and *C. laevigata* var. *reticulata*, netleaf hackberry (**bottom).**

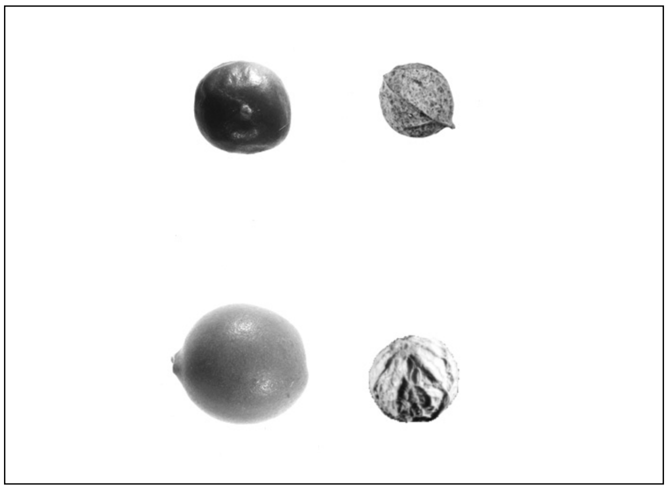
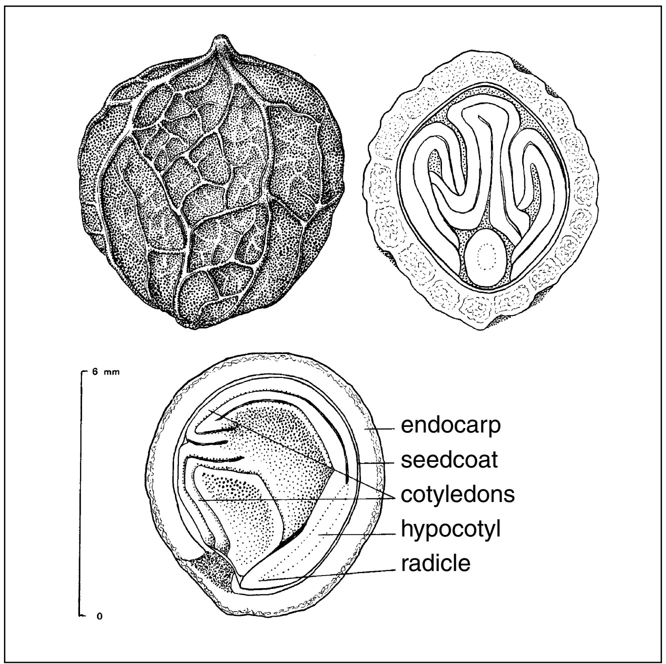
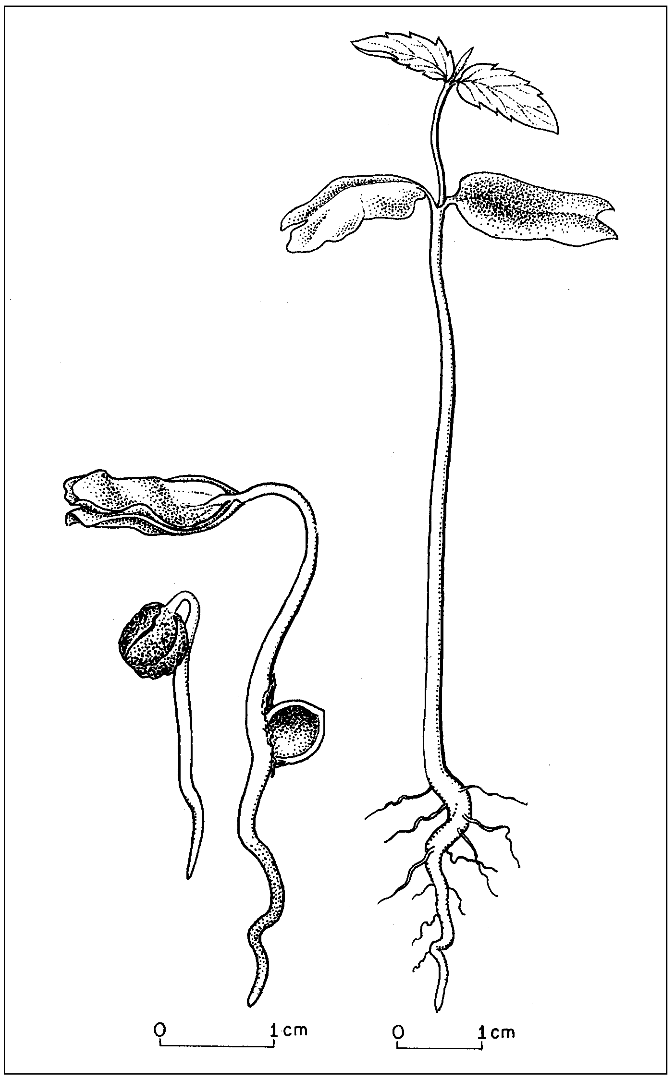


Figure 2—*Celtis occidentalis*, common hackberry: exterior view of seed (**top left**) and transverse section (**top right**) and cross section (**bottom**) of a seed.



Removal of the pulp may not be absolutely necessary, but it has been reported to aid germination of all 3 species (Bonner 1974; Vines 1960). Seed yield data are listed in table 4. Dry fruits or cleaned seeds store equally well in sealed containers at 5 °C. Dried fruits of hackberry were stored in this manner for 5 $^{1}/_{2}$ years without loss of viability (Bonner 1974), proving that they are orthodox in storage behavior.

Figure 3—*Celtis laevigata*, sugarberry: seedling development at 1, 2, and 5 days after germination.



Pregermination treatments. Hackberry seeds exhibit dormancy that can be overcome with stratification at 5 °C in moist media. Sugarberry, common hackberry, and netleaf hackberry all should be stratified for 90 to 120 days (Bonner 1974, 1984). In the southernmost part of sugarberry's range, no pretreatment was required for timely germination, but depulping prior to sowing was very beneficial (Vora 1989). Fermenting the fruits for 3 days at room temperature and then depulping before stratifying gave excellent results for common hackberry (Bonner 1974).

Germination tests. Germination test recommendations for treated seeds are the same for all 3 species (table 5). Untreated seeds should be tested for 90 days (Bonner 1974). Because of the long periods necessary for germination tests, rapid estimates of viability are very useful in this genus. Tetrazolium chloride staining works well with sugarberry. Incubation of clipped and imbibed seeds in a 1% solution for 24 hours at 26 °C has given good results (Bonner 1984).

C

Table 3—*Celtis,* hackberry: height, seed-bearing age, and fruit color

Species	Height at maturity (m)	Year first cultivated	Minimum seed-bearing age (yrs)	Fruit color: Preripe	Fruit color: Ripe
C. laevigata	18–24	1811	15	Green	Dark orange to red
C. laevigata var. *reticulata*	9–14	1890	—	—	Orange-red or yellow
C. occidentalis	9–40	1656	—	Orange-red	Dark reddish purple

Source: Bonner (1974).

Table 4—*Celtis,* hackberry: seed yield data

Species	Fruits/weight /kg	Fruits/weight /lb	Cleaned seeds/weight Range /kg	Range /lb	Average /kg	Average /lb
C. laevigata	4,850	2,200	8,150–15,600	3,700–7,080	13,200	6,000
C. laevigata var. *reticulata*	—	—	5,150–14,500	2,340–6,600	10,500	4,870
C. occidentalis	4,520	2,050	7,700–11,900	3,500–5,400	9,500	4,300

Source: Bonner (1974).

Table 5—*Celtis,* hackberry: germination test conditions and results

Species	Germinative test conditions * Temp (°C) Day	Night	Days	Germination rate Amount (%)	Days	Germination % Avg	Samples
C. laevigata	30	20	60	30–50	25–30	55	6+
C. laevigata var. *reticulata*	30	20	60	—	—	37	7
C. occidentalis	30	20	60	39	37	47	7

Source: Bonner (1974).
* Media used: sand, a sand–peat mixture, or a sandy loam soil.

Nursery practice. Both fall-sowing of untreated seeds and spring-sowing of stratified seeds are satisfactory. Seeds may be broadcast or drilled in rows 20 to 25 cm (8 to 10 in) apart and covered with 13 mm ($^1/_2$ in) of firmed soil. Beds should be mulched with straw or leaves held in place with bird screens until germination starts. Germination is epigeal (figure 4). These species can be propagated by cuttings (Bonner 1974), and grafting and budding success has been reported for common hackberry and sugarberry (Williams and Hanks 1976).

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Rubiaceae—Madder family

Cephalanthus occidentalis L.

buttonbush

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Other common names. common buttonbush, honey-balls, globe-flowers.

Growth habit, occurrence, and use. Buttonbush is a deciduous shrub or small tree that grows on wet sites from New Brunswick to Florida, west to southern Minnesota, Kansas, southern New Mexico, Arizona, and central California. It also occurs in Cuba, Mexico, and Central America (Little 1979). In the southern part of its range, buttonbush reaches heights of 4.5 to 6 m at maturity (Maisenhelder 1958), but it is shrubby in other areas. The seeds are eaten by many birds, and the tree has some value as a honey plant (Van Dersal 1938). Cultivation as early as 1735 has been reported (Vines 1960).

Flowering and fruiting. The perfect, creamy white flowers are borne in clusters of globular heads and open from June to September (Vines 1960). There is good evidence that buttonbush is largely self-incompatible (Imbert and Richards 1993). The fruiting heads (figure 1) become reddish brown as they ripen in September and October. Single fruits are 6 to 8 mm long (figure 2). Each fruit is composed of 2 or occasionally 3 or 4 single-seeded nutlets (figure 3) that separate eventually from the base (Bonner 1974b).

Figure 1—*Cephalanthus occidentalis*, common buttonbush: fruiting heads.

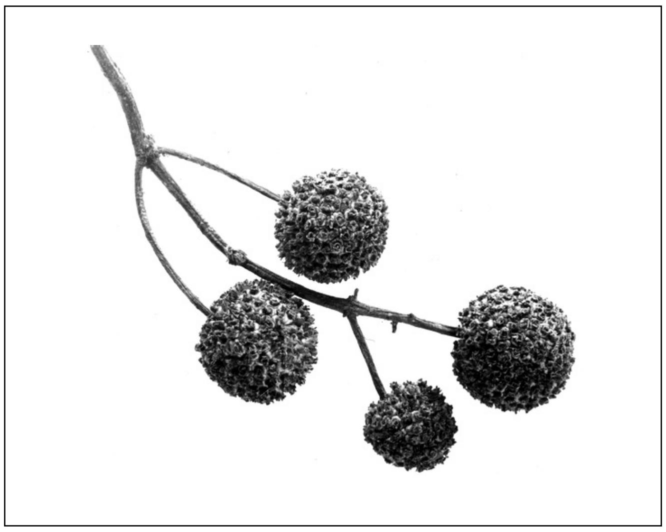
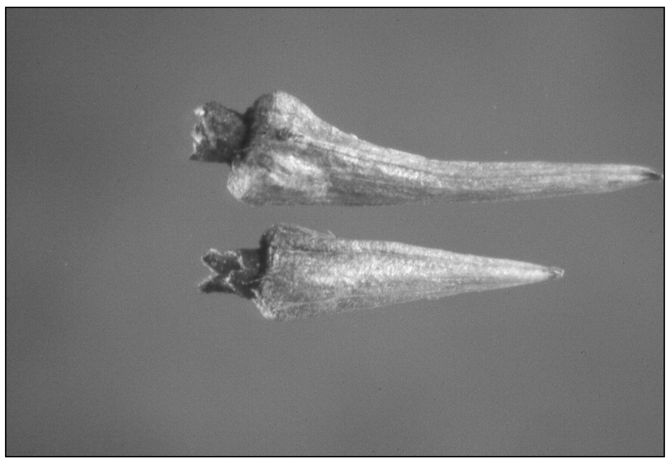


Figure 2—*Cephalanthus occidentalis*, common buttonbush: single fruits.



Collection and extraction. Collection can begin as soon as the fruiting heads turn reddish brown. Many heads disintegrate after they become ripe, but some persistent through the winter months. When the heads are dry, a light flailing will break them into separate fruits. Data from 4 samples of scattered origin showed 295,000 fruits/kg (134,000/lb), with a range of 260,000 to 353,000 (118,000 to 160,000). Purity in these seed lots was 96% (Bonner 1974b). The number of seeds per weight is about twice the number of fruits. Longevity of buttonbush seeds in storage is not known, but they appear to be orthodox in nature and thus easy to store. The principal storage food in the seeds is carbohydrate (Bonner 1974a).

Germination tests. Buttonbush seeds germinate promptly without pretreatment. Germination is epigeal (figure 4). Results with 2 test methods on seed from Louisiana (DuBarry 1963) and Mississippi (Bonner 1974b) were as follows:

	Louisiana	Mississippi
Medium	Water	Blotter paper
Temperature (°C)	24–34	30
Light	Yes	No
Test duration (days)	30	10
Germination (%)	86	78
No. of samples	4	4

Figure 3—*Cephalanthus occidentalis*, common buttonbush: longitudinal section through the 2 nutlets of a single fruit.

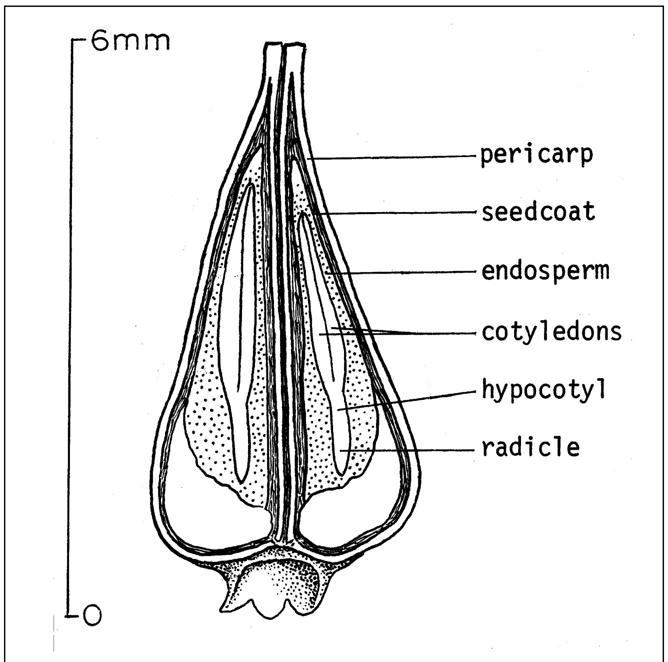
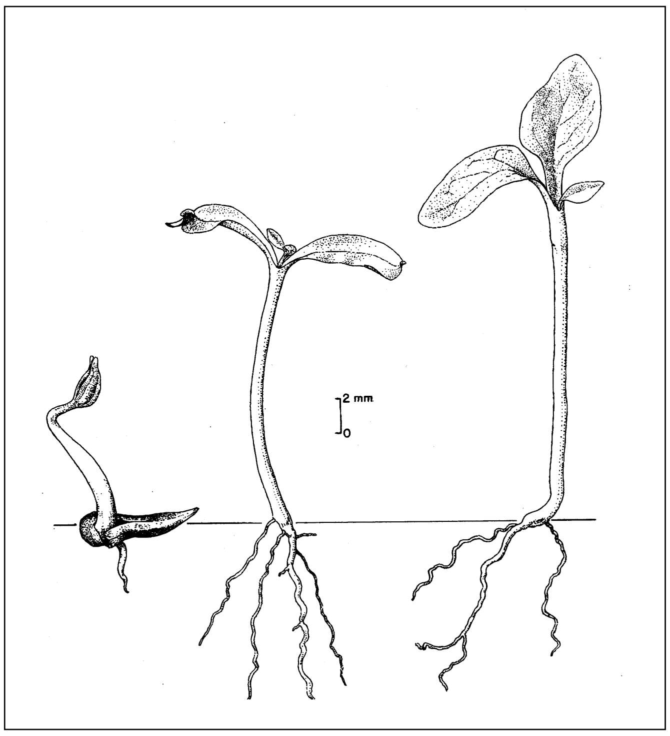


Figure 4—*Cephalanthus occidentalis,* common buttonbush: seedling development at 1, 23, and 40 days after germination.



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Fabaceae—Pea family

Ceratonia siliqua L.

carob

Wayne D. Shepperd

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Growth habit. *Ceratonia siliqua* L.—carob, St. John's bread, or locust—is a small to medium-sized broadleaf, evergreen tree that may grow to 20 m in height under ideal climatic conditions (Catarino 1993) but usually reaches heights of 8 to 15 m (Goor and Barney 1968). Carob is thought to be a tropical plant that has adapted well to Mediterranean climates by utilizing its deep rooting habit and xerophilous leaves to avoid water stress (Catarino 1993). The deep taproot's penetration into moist regions of the soil profile effectively lengthens the active growth period for carob leaves during the Mediterranean dry season (Rhizopoulou and Davies 1991).

Occurrence. Carob is native to the eastern Mediterranean from the southern coast of Asia Minor to Syria (Goor and Barney 1968; Griffiths 1952; Karschon 1960). It has been cultivated for thousands of years as a forage crop on a wide variety of soils in Asian, European, and North African countries along the coast of the Mediterranean Sea (Bailey 1947; Catarino 1993). Carob's sensitivity to low temperatures limits its area of distribution (Catarino 1993). Since its introduction to the United States in 1854, carob has done well only in the warm subtropical climates (southern Florida, the Gulf States, New Mexico, Arizona, and southern California) where annual rainfall is not below 30 to 35 cm (Bailey 1947; Coit 1951, 1962).

Use. Carob legumes (pods) are commonly used as animal feed or ground into flour and mixed with other cereals for human consumption. The legumes are rich in protein and sugar and are a highly nutritious livestock feed, comparable to barley and superior to oats (Bailey 1947; Coit 1962). However, the high sugar content (< 50%) is offset by a high tannin content (16 to 20%) that inhibits protein assimilation (Catarino 1993). Techniques are currently being developed to enzymatically separate and extract the phenolic tannin compounds to increase utilization (Catarino 1993). Legumes are also used in making health foods (as a chocolate "substitute"), carob syrup, and medicines such as laxatives and diuretics (Binder and others 1959; Coit 1951, 1962). In addition, they can be used as a cheap carbohydrate source for ethanol production, yielding 160 g of ethanol/kg of dry legumes (Roukas 1994). The annual production of carob legumes is 340,000 to 400,000 metric tons (374,800 to 441,000 tons), with Greece, Spain, Italy, and Portugal being primary producers (Roukas 1994; Catarino 1993).

Carob seeds are extremely hard, but the endosperm contains 30 to 40% by weight of galactomanane polysaccharides collectively known as carob-, or locust-bean gum (Catarino 1993). The compound is a valuable stabilizing and thickening additive used in the food processing, pharmaceutical, textile, paper, and petroleum industries.

The adaptability, ease of cultivation, and aesthetic appeal of carob also make it a desirable landscape plant (Catarino 1993). It is chiefly valuable in the United States as an ornamental evergreen but has been used to some extent in environmental plantings (Toth 1965).

Flowering and fruiting. The flowers, borne in small, lateral, red racemes, are polygamo-trioecious (Loock 1940). Nearly all cultivated species are dioecious, although flowers of both sexes may possess vestigial components of the other sex. Rarely, plants will possess both male and female flowers on the same stalk or completely hermaphroditic flowers (Catarino 1993). Flowers bloom from September to December in California, depending on the variety and the weather (Bailey 1947; Coit 1951).

The fruit is a coriaceous, indehiscent legume 10 to 30 cm long, 6 to 20 mm thick, filled with a sweet, pulpy substance bearing 5 to 15 obovate, transverse, brown, bony seeds about 6 mm wide (figures 1 and 2) (Bailey 1947; Coit 1951). Legumes ripen, turn dark brown, and begin to fall from September to November (California), depending on the variety and the weather (Bailey 1947; Coit 1951, 1962). Natural seedlings appear in Greece in November, even though temperatures do not favor shoot growth (Rhizopoulou and Davies 1991). Plants begin to bear fruit when 6 to 8 years old, and crops are abundant every second year (Bailey 1947). Average annual yield per tree at maturity

Figure 1—*Ceratonia siliqua*, carob: seed.

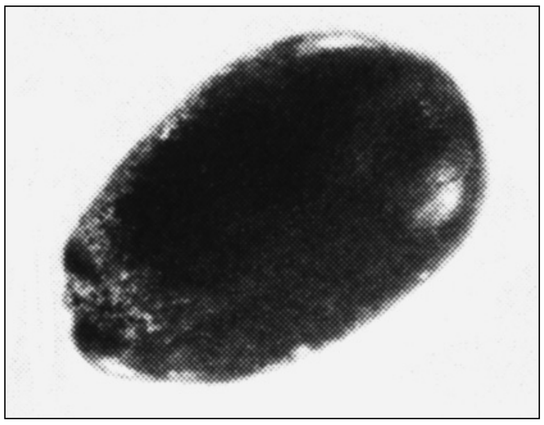
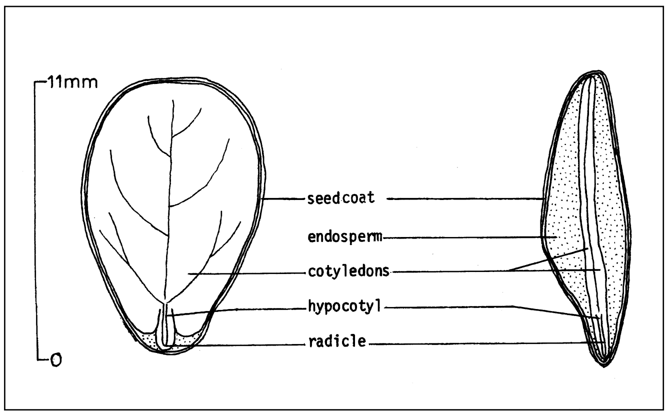


Figure 2—*Ceratonia siliqua*, carob: longitudinal sections through a seed.



is about 90 to 113 kg (200 to 250 lbs) of fruit (Coit 1951, 1962).

Collection of fruits. Fruits may be collected on the ground, or the ripe legumes may be shaken from the trees onto canvas sheets (Coit 1951). Legumes shaken from the tree should be allowed to remain on the ground for 2 to 3 days until completely dry (Coit 1962). Because of their high sugar content, legumes are likely to become moldy and quickly infested with a small scavenger worm—*Paramycelios transitella* Walker—if wet weather occurs during the harvesting season (Coit 1951, 1961, 1962). Because the worms infect the legumes while they are still attached to the tree, it is advisable to limit collections to dry years.

Extraction and storage of seed. Seeds are easily extracted after the legumes have been air-dried for a few days (Coit 1951; Goor and Barney 1968). If the legumes are to be stored for a time before extracting the seeds, they should be fumigated with an acceptable substitute for methyl bromide, which was recommended by Coit (1962) but is scheduled to be removed from use in the future. One kilogram (2.2 lb) of legumes yields about 50 to 140 g (1.8 to 4.9 oz) of cleaned seeds (Binder and others 1959). Cleaned seeds average 4,400 to 5,500 seeds/kg (2,000 to 2,500 seeds/lb) (Alexander and Shepperd 1974; Goor and Barney 1968). Soundness appears to be relatively high (<80% for 2 samples) (Alexander and Shepperd 1974). Seeds have remained viable for as long as 5 years when stored dry at low temperatures in sealed containers (Goor and Barney 1968).

Pregermination treatments. Seeds sown from recently ripened legumes germinate well without pretreatment (Rhizopoulou and Davies 1991), but if the seeds dry out they become very hard and do not readily imbibe water (Coit 1951). The best treatments to overcome seedcoat impermeability are soaking in concentrated sulfuric acid (H_2SO_4) for 1 hour and then in water for 24 hours, or alternatively, soaking for 24 hours in water that has first been brought to a boil and then allowed to cool (Goor and Barney 1968; Karschon 1960). Mechanical scarification is also effective in increasing the rate of water absorption with small lots of seeds (Coit 1951).

Germination tests. Germination tests have been run in moist vermiculite for 34 days at 21 °C. The germination rate was 66% for 16 days and the percentage germination was 80% (Alexander and Shepperd 1974).

Nursery practice. Seeds should be scarified by acid or hot water treatment and sown immediately afterwards in sterile soil or vermiculite under partial shade (Coit 1962; Karschon 1960). Seeds can be sown in either the spring or fall (Goor and Barney 1968). Seedlings have also been grown at 14 to 17 °C greenhouse temperatures with a 12-hour photoperiod of natural light supplemented with 250-W metal halide lamps (Rhizopoulou and Davies 1991). Seedlings develop a single deep taproot with a few small lateral roots less than 1.0 cm in length (Rhizopoulou and Davies 1991). Because the long taproot is easily injured, seeds should be sown in flats, pots, or containers so that seedlings can be outplanted with the original rooting medium intact (Coit 1951; Loock 1940). An alternate practice is to soak the legumes in water for 2 to 3 days and then plant without removing the seeds, but the germination rate is usually low (Coit 1951).

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C

Fabaceae—Pea family

Cercis L.

redbud

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Growth habit, occurrence, and uses. The genus *Cercis L.*—redbud—includes 8 species of trees and shrubs; 2 are indigenous to North America, 5 to China, and 1 to an area from southern Europe eastward to Afghanistan (Little 1979; Robertson and Lee 1976). Eastern redbud is widely distributed from southernmost Canada to central Mexico, spans about 24 degrees of longitude and 23 degrees of latitude, and has at least 1 well-defined variety, Texas redbud (table 1). This species shows clinal variation and substantial differences in morphological, dormancy, and hardiness characteristics associated with climatic and geographic conditions (Donselman 1976; Donselman and Flint 1982; Raulston 1990). California redbud also has variable characteristics (Smith 1986) within its much more restricted range in the southwestern United States. About 15 cultivars of eastern redbud have been developed and cultivars of other redbuds also are propagated (Raulston 1990).

Redbuds are deciduous, small- to medium-sized trees or shrubs with unarmed slender branchlets that lack terminal buds. Eastern redbud typically is a straight-trunked tree up to 12 m tall (table 2); the tallest on record reaches 13.4 m (AFA 1996). Although they also reach tree size, California and Texas redbuds are more commonly described as multiple-stemmed shrubs. California redbud grows from 2 to 6 m tall; the tallest one on record is 8.8 m and the tallest Texas redbud is about the same (AFA 1996). Eastern redbud occurs on many soils in moist open woodlands, flood plains, river thickets, and borders of small streams, whereas the variety, Texas redbud, often inhabits drier locations, primarily paleozoic limestone formations such as xeric pastures, hills, outcrops, and bluffs (Hopkins 1942). California redbud is unevenly distributed at elevations of 70 to 1,524 m along foothill streams, flats, draws, low slopes and canyons and on dry gravelly and rocky soils (Chamlee 1983; Jepson 1936; Sudworth 1908).

Redbuds are valued particularly for their showy buds and flowers that appear before the leaves (Clark and Bachtell 1992; McMinn and Maino 1937). They exhibit cauliflory—flowering directly along older branches and trunks—which is rare among temperate species (Owens and Ewers 1991) and contributes greatly to flowering showiness. Flowers typically are a deep reddish purple (magenta) but vary among localities (Coe 1993; Smith 1986) and species (table 3). Some white ones occur naturally, and cultivars have been developed for particular flower and leaf colors (Raulston 1990). Ornamental uses are extensive within each species' indigenous range and several species have proven hardy more extensively (McMinn and Maino 1937;

Table 1—*Cercis,* redbud: nomenclature and occurrence

Scientific name & synonym	Common name(s)	Occurrence
C. canadensis **L.**	**eastern redbud**, redbud, Judas-tree	Connecticut W to S Ontario, Michigan, Iowa, & E Nebraska; S to Texas & central Mexico; E to Florida
C. canadensis **var.** ***texensis*** **(S.Wats.) M. Hopkins**	**Texas redbud**	S Oklahoma to SE New Mexico & Texas
C. canadensis **var.** ***mexicana*** **(Rose) M. Hopkins**	**Mexican redbud**	E-central Mexico
C. orbiculata **Greene** *C. occidentalis* Torr. ex Gray var. *orbiculata* (Greene) Tidestrom	**California redbud**, Arizona redbud, western redbud	Utah, Nevada, California, & Arizona

Sources: Clark and Bachtell (1992), Hopkins (1942), Hosie (1969), Little (1979), Robertson and Lee (1976), Sargent (1933).

Table 2—*Cercis,* redbud: growth habit, height, legume color, and size

Species	Growth habit	Height at maturity (m)	Legume color	Legume size: Length (cm)	Legume size: Width (mm)	Seed diameter (mm)
C. canadensis	Tree or shrub	7–12	Reddish brown	5–10	8–18	4–5
C. canadensis var. *texensis*	Shrub or tree	4–10	Reddish brown	6–10	8–25	4–5
C. orbiculata	Shrub or tree	2–6	Reddish purple, dull red, to reddish brown	4–9	13–25	3–4

Sources: Fernald (1950), Hopkins (1942), Hosie (1969), Jepson (1936), McMinn (1939), Munz and Keck (1959), Sargent (1933).

Table 3—*Cercis,* redbud: phenology of flowering and fruiting

Species	Flowering	Flower color	Fruit ripening
C. canadensis	Mar–May	Magenta–purplish pink	July–early autumn
C. canadensis var. *texensis*	Mar–Apr	Magenta pink	Aug–Sept
C. orbiculata	Feb–May	Magenta pink–reddish purple	July–Sept

Sources: Abrahms (1944), Clark and Bachtell (1992), Fernald (1950), Hopkins (1942), Jepson (1936), Mirov and Kraebel (1939), Van Dersal (1938).

Robertson 1976). Where redbuds are numerous, they provide valued bee pasture in early spring (Magers 1970). The buds, flowers, and legumes (pods) of redbuds are edible and have been used in salads or batter (Coe 1993). Native Californians used the roots and bark of California redbud in basketry (Coe 1993; Jepson 1936); remedies for diarrhea and dysentery were also made from the astringent bark (Balls 1962).

Redbuds also are used for borders, erosion control, windbreaks, and wildlife plantings. Eastern redbud is browsed by white-tail deer (*Odocoileus virginiana*) and the seeds are eaten by birds, including bobwhite (*Colinus virginianus*) (Van Dersal 1938). California redbud is moderately important as fall and spring browse for deer but has been rated only fair to poor for goats and poor or useless for other livestock (Sampson and Jespersen 1963).

Two fungal diseases affect the flowering and attractiveness of eastern redbud—verticillium wilt (*Verticillium* sp.) and botryosphaeria canker (*Botryosphaeria dothidea* (Moug.:Fr.) Ces. & De Not.)—by causing die-back of branches. The canker has become more common and destructive in the eastern United States, appearing to attack trees that are under stress (Geneve 1991a; Raulston 1990; Vining 1986).

Flowering and fruiting. Flowering occurs from February to May, varying somewhat by species and location (table 3). The bisexual redbud flowers are brilliant pink to reddish purple and develop on older wood from dormant axillary buds laid down 1 to several years earlier (Owens and Ewers 1991). The flowers are borne sessile or on short, thin pedicels in umbel-like clusters densely covering the branches and trunk. Flowers of California redbud are somewhat larger than those of eastern redbud (Hopkins 1942; Robertson 1976). Eastern redbud begins flowering in 3 to 4 years from seed when trees are 1.5 to 2 m tall, and trees in open or semi-open locations flower most abundantly (Clark and Bachtell 1992; Raulston 1990). Pollination is usually done by long- and short-tongued bees (Robertson 1976). Crops of legumes are produced abundantly by both eastern and California redbud but seed set is more variable (Hopkins 1942; Jepson 1936).

Redbud fruits are pendulous, flattened legumes (figure 1) 4 to 10 cm long (table 2). The generic name *Cercis* (Greek *kerkis*, weaver's shuttle) apparently alludes to the shape of the legume (Robertson and Lee 1976). The legumes of California redbud are somewhat wider and shorter than those of eastern redbud. Legumes of eastern redbud contain 4 to 10 seeds each; those of California redbud only a few (Hopkins 1942; Robertson 1976). Legume color varies from lustrous reddish brown to dull red and turns tan or brown as the fruits mature and dry in July or later. Some legumes open and release their seeds in autumn, but many remain closed for most of the winter. Seeds are released from legumes on the tree or on the ground when the legume

Figure 1—*Cercis canadensis*, eastern redbud: 4 to 10 seeds (**left**) are in each legume (**right**).

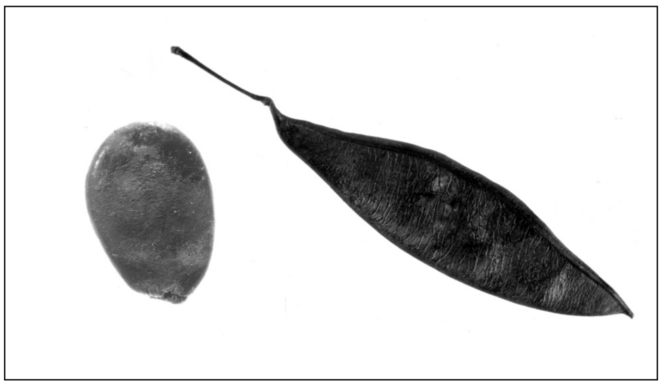
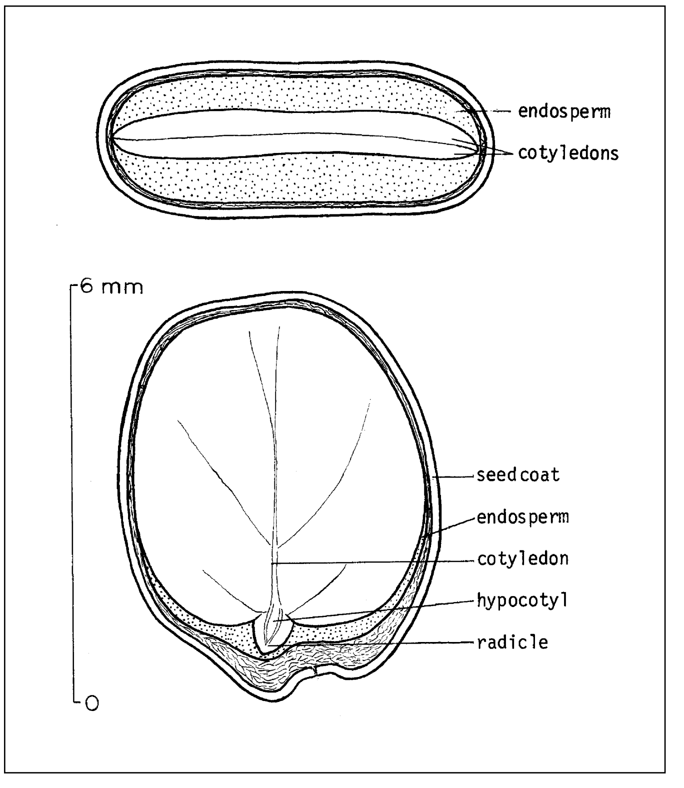


Figure 2—*Cercis canadensis*, eastern redbud: the flattened seed in transverse section (**above**) and longitudinal section (**below**).



sutures open or the walls decay (Robertson 1976). The seeds are dispersed by wind, birds, and animals, with the proportions carried by each varying by location.

Redbud seeds are somewhat flattened, oval to rounded, and hard (figures 1 and 2). Those of eastern redbud are 4 to 5 mm in diameter; those of California redbud are slightly smaller (table 2). The light tan to dark brown seedcoats are thin but made up of thick-walled cells impermeable to water (Afanasiev 1944). At maturity the embryo is straight, well-developed, and surrounded by endosperm (figure 2).

Collection, extraction, and storage. Legumes can be collected any time after they turn tan or brown. Although legumes remain closed on trees for lengthy periods, prompt collection is prudent to minimize the substantial seed losses that might occur from insects or other factors (Afanasiev 1944). Legumes can be picked by hand or loosened by flailing or shaking the branches and caught on ground cloths. Collected legumes should be temporarily stored and transported in loosely woven sacks.

If legumes are not fully dry when collected, they should be spread thinly and dried until brittle in the sun, under shelter, or in a kiln at 38 to 41 °C. The legumes can be threshed manually or in a variable speed, modified seed separator, hammermill, or grinder. Seeds are separated from the chaff by screening and fanning. Nearly 100% purity is readily obtainable in cleaning the smooth redbud seeds (Lippitt 1996).

After thorough air-drying, seeds can be stored in cloth bags or in closed glass, metal, or fiberboard containers. Because of their impermeable seedcoats, redbud seeds should store dry reasonably well at room temperature or in cool or cold storage, but little storage experience has been reported. Zins (1978) obtained substantial germination from an eastern redbud seedlot stored for 13 years in a glass jar at –25 °C. Seeds of California redbud have been stored satisfactorily for 12 years or more under the same conditions as many conifers at a moisture content of 5 to 9% and temperature of –18 °C (Lippitt 1996).

Seeds of California redbud average about half again as heavy as those of eastern redbud but seed weight varies widely among lots for both species (table 4).

Pregermination treatments and germination tests. Redbud seeds generally require pregermination treatment to overcome dormancy attributable both to a hard, impermeable seedcoat and to some demonstrated, but not fully identified, embryo dormancy (Afanasiev 1944; Geneve 1991b; Hamilton and Carpenter 1975; Heit 1967a7b; Jones and Geneve 1995; Profumo and others 1979; Rascio and others 1998; Riggio-Bevilacqua and others 1985; Tipton 1992; Zins 1978). Test results indicate that the level of dormancy varies by species, seed source, seedlot, age of seeds, and perhaps other factors. Given such variable dormancy, pretreatment might involve using the one demonstrated to be most broadly applicable, or determining sufficiently the nature of dormancy in local lots and applying a customized pretreatment.

Three pretreatments have proven satisfactory for overcoming redbud's seedcoat impermeability—mechanical scarification, immersion in sulfuric acid, or in hot water (table 5). In comparison tests, the acid treatment has generally produced more consistent or slightly better results (Afanasiev 1944; Liu and others 1981), but good imbibition of water has resulted after all 3 treatments. Acid treatment involves immersing redbud seeds in concentrated sulfuric acid for 15 to 90 minutes at room temperature followed by thorough washing in water (Afanasiev 1944; Frett and Dirr 1979; Liu and others 1981). Length of treatment required can be determined on a small sample; if immersion is too short, seedcoats remain impermeable, if too long, the seeds are damaged. Well-rinsed, acid-scarified seeds can be placed immediately in stratification or surface-dried and stored several months until sown by hand or seeder (Heit 1967a).

Abrading, clipping, or piercing the seedcoat to expose the endosperm and allow ready entry of water (Hamilton and Carpenter 1975; Riggio-Bevilacqua and others 1985; Zins 1978) can be done easily for small test lots but not as readily in quantity. Immersing small or large quantities of seeds in hot or boiling water can be done easily, but results have been more variable than for acid treatment—sometimes reasonably good (Fordham 1967; Mirov and Kraebel 1939), other times poor to mediocre (Afanasiev 1944; Liu and others 1981). Hot water treatment clearly makes redbud seedcoats permeable but may cause internal damage. Better calibration of time–temperature effects appears needed—

Table 4—*Cercis,* redbud: seed yield data

		Seed wt/ legume vol		Cleaned seeds/wt				
	Seeds/100 wt			Average		Range		
Species	of legumes	kg/hl	lb/bu	/kg	/lb	/kg	/lb	Samples
C. canadensis	20–35	—	—	39,570	17,950	30,870–55,100	14,000–25,000	18
C. orbiculata	44	2.10	1.64	27,460	12,455	20,950–40,100	9,500–18,169	24

Sources: Lippitt (1996), Roy (1974), USDA FS (1948, 1996).

Table 5—*Cercis,* redbud: scarification, stratification, germination test conditions, and test results*

	Scarification		Stratification		Germination test conditions				
Species	Treatment	Time (min)	Days	Temp (°C)	Medium	Temp (°C)	Days	Germination (%)	Samples
C. canadensis	H_2SO_4	45	Var.	5	Peat	—	48	77	2
	H_2SO_4	45	Var.	5	Peat	—	107	78	2
	H_2SO_4	30	42	5	Cotton	21	8	97	2
	H_2SO_4	25–30	35–91	3–7	Cotton	—	—	88–100	7†
	H_2SO_4	30	60	5	Sand	20–30	30	80	2
	Mech.	—	60	5	Peat–perlite	25	24	90	5
	H_2SO_4	30	60	5	Peat–perlite	25	24	88	5
	Mech	—	—	—	Peat–perlite	25	24	82	5
	H_2SO_4	30	—	—	Peat–perlite	25	24	91	5
	H_2SO_4	15–60	60	5	Vermiculite	18–21	42	87	3
	H_2SO_4	30–90	60	5	Soil	20–27	14	67–72	12
	—	—	0	1	Paper	20–30	28	43	1
	—	—	28	1	Paper	20–30	28	83	1
C. canadensis	H_2SO_4	62	35	5	Paper	21	14	95	25‡
var. *texensis*	H_2SO_4	62	35	5	Paper	28	14	100	81‡
C. orbiculata	Heat§	Overnight	—	—	Vermiculite	—	118	38	1
	Heat	9	—	—	Vermiculite	—	118	52	1
	H_2SO_4	60	90	2–4	Cotton	—	10	84	1

Sources: Afanasiev (1944), Flemion (1941), Frett and Dirr (1979), Hamilton and Carpenter (1975), Heit (1967a), Liu and others (1981), Roy (1974), Tipton (1992), USDA FS (1948, 1996), Williams (1949).

* Only the better results for each test series are listed. In several studies, only full seeds were tested.

† Best results from a set of tests on each of 7 seedlots.

‡ Test combinations used to develop a response surface.

§ Moist heat applied by immersing seeds in 82 °C water that cooled gradually.

// Dry heat applied in oven at 121 °C.

whether to dip the seeds for 15 or more seconds in boiling water or immerse them overnight in 60 to 88 °C water that cools gradually. Application of dry heat also appears to have promise (Williams 1949).

After scarification, cold stratification is generally required to overcome some degree of internal dormancy and maximize seed germination. Germination differences between unstratified and cold-stratified seeds range from none (Hamilton and Carpenter 1975), to fractional differences in the response of excised embryos (Geneve 1991b), up to major differences for intact seeds (Afanasiev 1944; Fordham 1967; Frett and Dirr 1979; Geneve 1991b). Stratification of eastern redbud for 28 to 60 days at 1 to 7 °C has proven satisfactory (table 5) and 90 days for California redbud (Heit 1967a; Van Dersal 1938). Up to a point, seed response improves with longer stratification, and extended stratification generally does no harm. Seeds should be sown promptly after stratification; drying out for more than 6 days at room temperature reduced germination of eastern redbud seeds (Afanasiev 1944).

A pretreatment and germination test protocol has not yet been specified for redbud seeds due perhaps to extensive variability in seedlot characteristics, length of time required, and low demand for a standard test. Pretreated seeds of eastern redbud will germinate at temperatures of 1 to 38 °C; Afanasiev (1944) concluded 8 days at 21 °C was most satisfactory. Texas redbud seeds germinate at 24 to 31 °C and 28 °C was optimum (Tipton 1992). Germination test methods currently used for many species—14 to 28 days at alternating temperatures of 20 and 30 °C—seem to nicely bracket conditions that yielded high germination from pretreated redbud seeds (table 5).

Viability of redbud seeds is most easily and rapidly determined by a tetrazolium (TZ) test or a growth test of excised embryos. The TZ test is the only method prescribed by the International Seed Testing Association; preparation and evaluation procedures to use are listed in a published handbook (Moore 1985). In brief, the seeds are cut at the distal end of the cotyledons either dry or after overnight soaking in water at room temperature. Soaking in a 1% TZ solution follows for 6 to 24 hours at 35 °C. The embryos are then cut longitudinally, and the staining pattern of cotyledons, hypocotyl, and radicle evaluated. A growth test of excised embryos requires making the seedcoats permeable with acid, hot water, or mechanical scarification; soaking the seeds overnight, excising the embryos, and incubating them for 4 to 6 days on moist filter paper at 20 °C (Flemion 1941; Geneve 1991b). Viability determined by tetrazolium or excised embryo test reveals that the seeds' maximum potential and generally is higher than indicated by a germination test (Flemion 1941; Hamilton and Carpenter 1975; USDA FS 1996).

Nursery practice. Redbuds are propagated most readily from seeds sown either in the fall or spring. Fall-sown seeds may or may not be scarified, and stratification occurs naturally in the seedbeds (Lippitt 1996; Raulston 1990). In one reported instance, immature seeds of eastern redbud collected, extracted, and sown before the seedcoats hardened yielded 90% germination the following spring (Titus 1940). When seeds need to be scarified for either fall- or spring-sowing—acid treatment for 15 to 60 minutes, rinsing, and a 24-hour soak in water; a boiling water dip for 15 seconds or more followed by a 24-hour soak in cooler water; or immersion overnight in 88 °C hot water that gradually cools—can be generally used to overcome seedcoat dormancy (Frett and Dirr 1979; Heit 1967b; Lippitt 1996; Raulston 1990; Robertson 1976; Smith 1986). After scarified seeds have imbibed water, they may need to be sorted to separate those not swollen and still impermeable for further treatment. When necessary, seeds are stratified at 1 to 5 °C for 30 to 90 days. Stratification requirements are uncertain for 2 reasons: variability among seedlots and the unknown stratification effect produced by low temperature storage of the seeds.

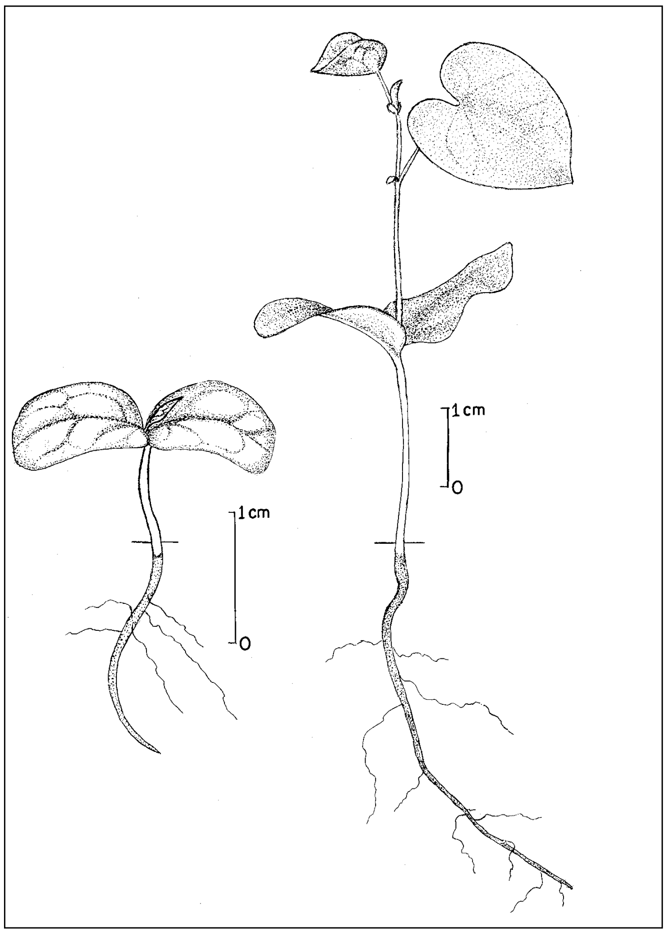
Surface-dried seeds are drill- or broadcast-sown and covered 0.6 to 2.5 cm (0.2 to 1 in) deep with soil, sand, sawdust, or bark. Some nurseries fumigate, then reinoculate before sowing seedbeds. Presence of an endomycorrhizal fungus is important; inoculation with *Glomus fasciculatum* (Thaxter) Gerdemann and Trappe has increased first-season growth of eastern redbud as much as 72% (Maronek and Hendrix 1978). Mulching of fall-sown beds can be beneficial but the mulch must be removed when germination starts. Germination is epigeal (figure 3).

Seedling return from nursery sowings is very variable. For eastern redbud, an average of 2,425 usable seedlings (range 617 to 7,055) were produced per kilogram of seeds (1,100 usable seedlings/lb, range 280 to 3,200/lb) (Roy 1974). Germination is fairly consistent year to year for California redbud, averaging 54 to 60% (Lippitt 1996). Under favorable conditions, seedling height growth of eastern redbud can be rapid: about 0.5 m (20 in) in reinoculated soil (Maronek and Hendrix 1978), 1 m (40 in) or more under an intensive nitrogen fertilizer schedule, and about 2 m (80 in) if started in January in a greenhouse under long-day conditions and transplanted outdoors after the danger from frost is over (Raulston 1990).

Redbud seedlings are also produced in pots and tube containers in both greenhouses and shadehouses where production practices and growth conditions can be closely controlled. To gain the benefits of natural stratification, containers may be sown in the fall and overwintered in shadehouses. Treatments to prevent botrytis are necessary soon after late February germination of California redbud (Lippitt 1996). Seedlings suitable for outplanting—15 to 30 cm (6 to 12 in)—can be produced readily in one season (Clark and Bachtell 1992; Lippitt 1996).

Redbuds are relatively difficult to propagate vegetatively, but that must be done to produce the desired cultivars. Redbud cultivars are generally budded or grafted. Field-grown stock is easier to bud then container-grown stock, and summer budding is much more successful then winter budding (Raulston 1990). Much effort and some progress has been reported on reproducing redbud from stem cuttings (Tipton 1990) and tissue cultures (Bennett 1987; Geneve 1991a; Mackay and others 1995).

Figure 3—*Cercis orbiulata*, California redbud: young seedlings grow rapidly: first leaf stage (**left**) and about 1 month old (**right**).



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C

Rosaceae—Rose family

Cercocarpus Kunth

mountain-mahogany

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Growth habit, occurrence, and use. The mountain-mahoganies—genus *Cercocarpus*—are 8 to 10 species of moderately to intricately branched shrubs or small trees that are endemic to dry coastal and interior mountains of the western United States and Mexico (Stutz 1990). Leaves are generally persistent and stems are unarmed. Two of the most widely distributed and utilized species are described here (table 1).

Curlleaf mountain-mahogany populations demonstrate considerable variability in height (Davis 1990; Stutz 1990). In some areas, the species occurs as a medium-statured shrub of 1 to 2 m. More commonly, it is a small tree of 4 to 10 m at maturity. Trunk diameter of mature trees measures 30 to 100 cm (Johnson 1970). Schultz and others (1990) estimated the mean age of trees in central and western Nevada stands to be 352 years. Mean plant age in Utah stands (85 years) is less than that in Nevada stands but greater than that in Oregon and Montana stands (Davis 1990).

True mountain-mahogany is a deciduous shrub of 1 to 5 m. Both species occur as components of mixed communities and as dominants in extensive stands and are important cover and browse species for wildlife, especially big game (Davis 1990). When burned, true mountain-mahogany resprouts from the crown, resulting in relatively rapid stand recovery following fire. Recovery of curlleaf mountain-mahogany stands following fire is from seed only and can be extremely slow. Because they are long-lived, produce an extensive root system, and survive well on dry steep slopes, mountain-mahogany plants play an important role in erosion control. Nitrogen fixation in root nodules has been described for both curlleaf (Lepper and Fleschner 1977) and true mountain-mahoganies (Hoeppel and Wollum 1971), suggesting a significant role by these species in improving fertility in otherwise infertile soils. The wood of curlleaf mountain-mahogany is extremely dense and heavy and has had limited use, primarily as fuel wood (Johnson 1970).

Geographic races and hybrids. Two distinct subspecies or varieties of curlleaf mountain-mahogany occur in the western United States (Stutz 1990). Although considerable overlap in distribution exists, *C. ledifolius* ssp. *ledifolius* (formerly ssp. *intercedens)* has a more northeastern distribution, whereas the distribution of ssp. *intermontanus* is centered to the west of its sister taxon. In northern Idaho, northern Wyoming, and southern Montana, ssp. *ledifolius* is the only mountain- mahogany taxon present (Stutz 1990). The leaves of ssp. *ledifolius* plants differ from those of ssp. *intermontanus* in being narrower, more strongly involute, and densely pubescent ventrally. The leaves of ssp. *intermontanus* are broadly elliptic and glabrous. Habit of ssp.

Table 1—*Cercocarpus,* mountain-mahogany: nomenclature and occurrence

Scientific name(s)	Common name(s)	Occurrence
C. ledifolius **Nutt.**	**curlleaf mountain-mahogany**, curlleaf cercocarpus, curlleaf mahogany, desert mahogany	Washington & Oregon E to Montana & Wyoming, S to Arizona, California, & Mexico (Baja)
C. montanus **Raf.** *C. betuloides* Nutt. *C. parvifolius* Nutt. *C. flabellifolius* Rydb.	**true mountain-mahogany,** mountain cercocarpus, birchleaf cercocarpus, birchleaf mountain-mahogany, alderleaf mountain-mahogany, blackbrush, deerbrush, tallowbrush	Oregon E to South Dakota S to Mexico, incl. parts of Wyoming, Colorado, Nebraska, Kansas, Texas, New Mexico, Arizona, Utah, & California

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ledifolius is more shrubby (or less tree-like) than that of ssp. *intermontanus,* especially in its northern distribution. Although it is treated as a separate species, littleleaf mountain-mahogany—*C. intricatus* Wats.—is taxonomically and phenotypically close to curlleaf mountain-mahogany spp. ledifolius. It is distinguished by its smaller leaves and stature, fewer stamens, and shorter style on mature fruits (Stutz 1990). The evolutionary processes that produced littleleaf mountain-mahogany are still proceeding and intermediates between the 2 taxa are common.

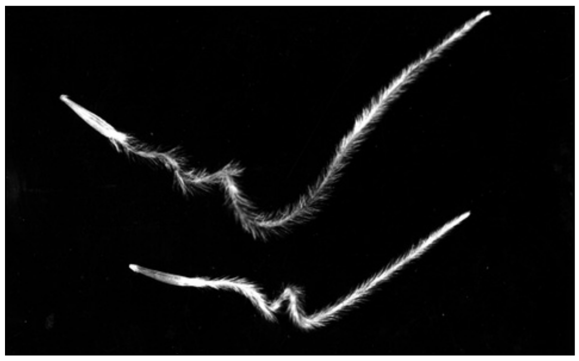
As reflected in its taxonomy, true mountain-mahogany is also quite variable across its range. *C. montanus* ssp. *montanus* has the most widespread distribution (Stutz 1990). Separate taxa have been described for parts of the Pacific Coast (ssp. *betuloide*s Nutt.) and in the Southwest (ssp. *paucidentatus* S. Wats and *argenteus* Rydb). *Cercocarpus mexicanus* Hendrickson, *C. rzedowski* Hendrickson, and *C. fothergilloides* Kunth. are closely related Mexican species.

Inter-specific hybrids are common between curlleaf and true mountain-mahoganies (Stutz 1990). Fertility in hybrids of true mountain-mahogany and curlleaf mountain-mahogany ssp. *ledifolius* is good in contrast to the low fertility encountered in hybrids of true mountain-mahogany × curlleaf mountain-mahogany spp. *intermontanus* (Stutz 1990). Hybrids between true and littleleaf mountain-mahoganies are rare.

Flowering and fruiting. Small perfect flowers bearing no petals are borne individually or in small clusters. Flowering for these wind-pollinated shrubs occurs some time between late March and early July depending on latitude, elevation, and aspect. Fruits are cylindrical achenes bearing a single seed and are distinguished by a 3- to 10-cm plumose style that facilitates wind dispersal (figure 1). Ripened fruits disperse from July through October. Abundant fruit production occurs at 1- to 10-year intervals (Plummer and others 1968); however, a high percentage of nonviable (empty) fruits is not uncommon. Plants may reach reproductive maturity in 10 to 15 years (Deitschman and others 1974).

Fruit collection. Fruit maturation within a stand is generally somewhat asynchronous. Because of this and because fruits will not dislodge before they are fully ripe, harvests are most productive when delayed until the fruits on a majority of plants ripen. Optimal timing for harvest varies between July and September. Delays may result in diminished or lost harvests due to wind dispersal. Fruits of several plants must be examined for fill and insect damage before starting collection. Ripe, dry mountain-mahogany fruits are easily shaken from branches onto tarps or hand-

Figure 1—*Cercocarpus*, mountain-mahogany: achenes with feathery style; the size of the achene varies greatly within each species.



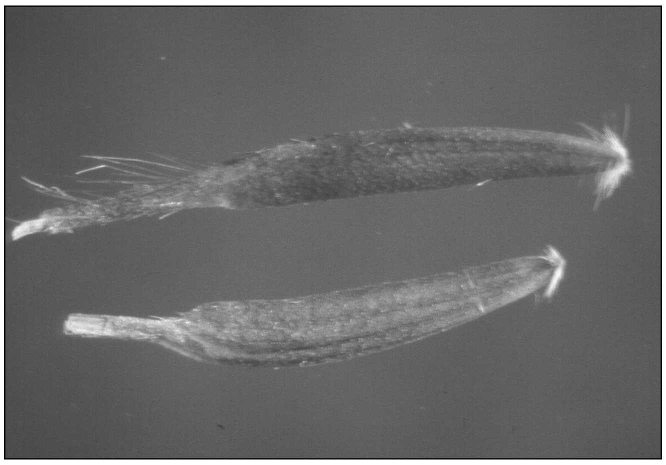
held hoppers using a beating stick. During harvest and handling, short hairs dislodge from the fruits. These hairs cause considerable discomfort to eyes and skin, thus the cowboy epithet of "hell feathers" (Plummer and others 1968). Fruits may collect in harvestable depths on the ground during years of superior production. However, collections from ground accumulations are often of poor quality due to the removal of viable seeds by rodents.

Cleaning and storage. Highest purity values are obtained by removing most broken branches from fruits during collection. For large collections, empty fruits, styles, and fine hairs are best removed using a variable-speed debearder and a 9.5-mm (#2) screen fanning mill (figure 2). Hammermilling causes excessive breakage and should not be used. Minimum standards accepted by the Utah Division of Wildlife Resources for both species are purity values of 95%, and viability values of 85% (Jorgensen 1995).

Cleaned-fruit sizes differ by species, ecotype, and year of collection. In one study, average number of fruits per weight for curlleaf (8 collections) and true mountain-mahoganies (10 collections) was 106,000 and 88,000/kg (48,000 and 40,000/lb), respectively (Kitchen and others 1989a&b). These fruit weights were either equivalent to or somewhat heavier than those previously reported (Deitschman 1974). Curlleaf and true mountain-mahogany fruits stored under warehouse conditions experienced no significant loss of viability during 15 and 7 years, respectively (Stevens and others 1981).

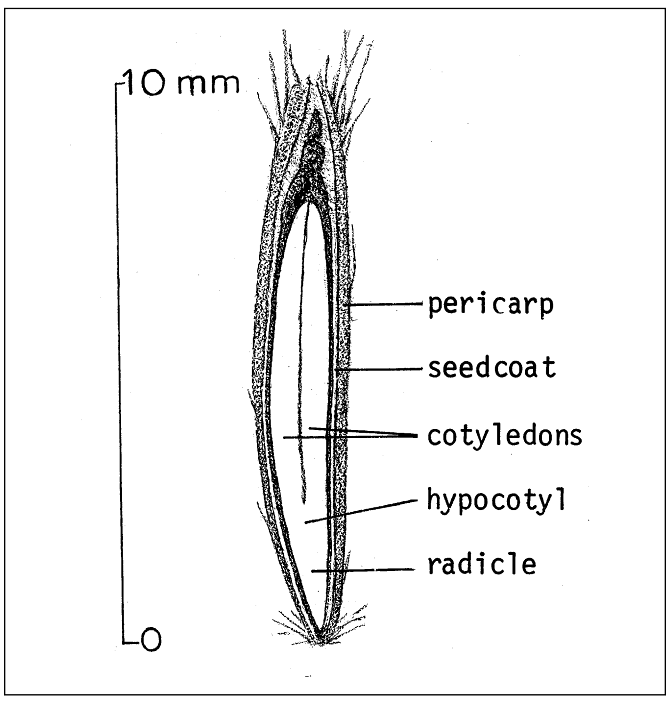
Germination. Reported germination responses to moist chilling for curlleaf mountain-mahogany range from no response after 12 weeks (Young and others 1978), to good germination with 4 weeks (Heit 1970). In most of these studies, interpretation of results is difficult because fruit fill percentage was not determined. Dealy (1975) reported 20% germination in response to 60 days of moist

Figure 2—*Cercocarpus montanus*, true mountain-mahogany: achenes with styles removed (cleaned seeds).



chilling (4 °C) followed by 30 days at 20 °C for a 2-year old Oregon source that had tested 78% viable. He also observed germination during extended chilling (75 to 270 days). Kitchen and Meyer (1990) found the length of wet chilling (1 to 2 °C) required to make 90% of viable seeds germinable at 15 °C ranged from 6 to 10 weeks for 6 fresh collections from Utah, Idaho, and Nevada. They observed that cold-temperature germination began at about 8 weeks. Chemical treatments that have provided limited success in breaking dormancy with curlleaf mountain-mahogany seeds include: gibberellins (GA_3), thiourea, hydrogen peroxide,

Figure 3—*Cercocarpus ledifolius*, curlleaf mountain-mahogany: longitudinal section through an achene.

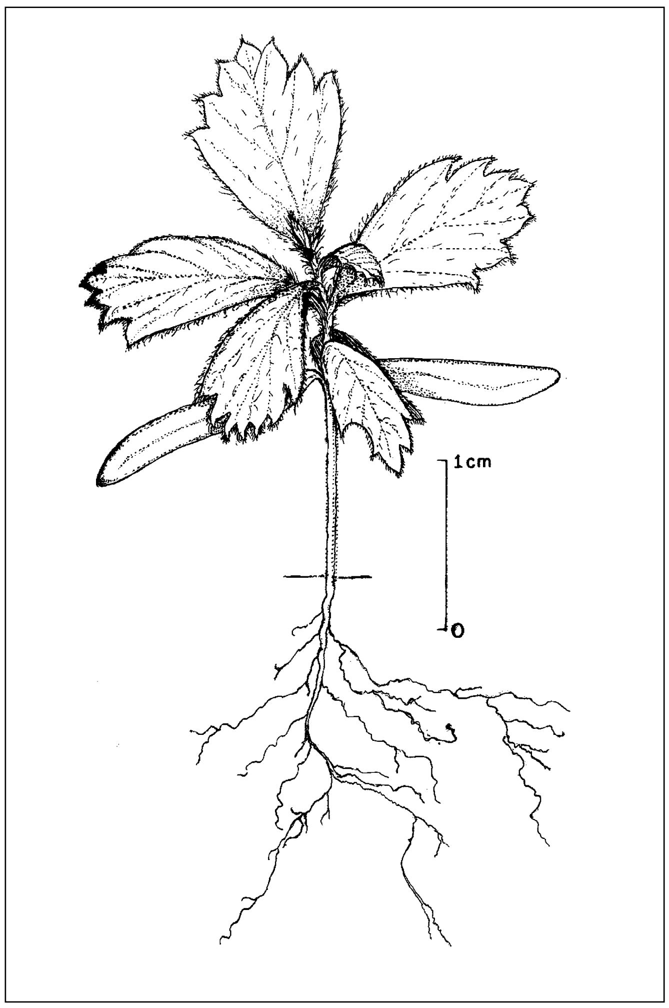


and sulfuric acid (Dealy 1975; Stidham and others 1980; Young and others 1978).

Some collections of true mountain-mahogany seeds have tested largely nondormant (Deitschman and others 1974). More typically, 2 to 12 weeks of moist chilling are required to break dormancy (Kitchen and Meyer 1990). Kitchen and Meyer (1990) found that cold-temperature germination (1 to 2 °C) for 9 Colorado and Utah collections began after 7 to 10 weeks of moist chilling.

Consistent estimations of embryo viability using standard TZ (tetrazolium) staining procedures are difficult to obtain for both species (Kitchen and others 1989a, 1989b). This is because the embryo is held tightly in the cylindrical pericarp and is difficult to extract for staining and examination (figure 3). Technical experience with mountain-mahogany TZ evaluations appears to be a major factor in accuracy of test results.

Figure 4— *Cercocarpus montanus*, true mountain-mahogany: seedling with primary leaves and well-developed secondary leaves.



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Nursery and field practice. Curlleaf and true mountain-mahoganies were first cultivated in 1879 and 1872, respectively (Deitschman and others 1974). Bareroot and container nursery stock are commercially available for both species, generally as 1- or 2-year-old stock. Unless nondormant collections are used, cleaned fruits are either prechilled or fall-sown. Seedbeds should be kept moist until seeds have germinated (Deitschman and others 1974). Deep-rooting containers filled with a minimum of 0.2 liter (13 in^3) standard potting mix is recommended for container stock production (Landis and Simonich 1984). With optimum rearing conditions a minimum of 4 to 6 months is required to develop an adequate root system. Figure 4 illustrates a seedling with well-developed secondary leaves. Direct seeding of mountain-mahogany should be carried out in fall or early winter in conjunction with seedbed preparations that minimize competition to first-year seedlings (Plummer and others 1968).

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