Verbenaceae—Verbena family

Callicarpa americana L.

American beautyberry

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station, Mississippi State, Mississippi

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

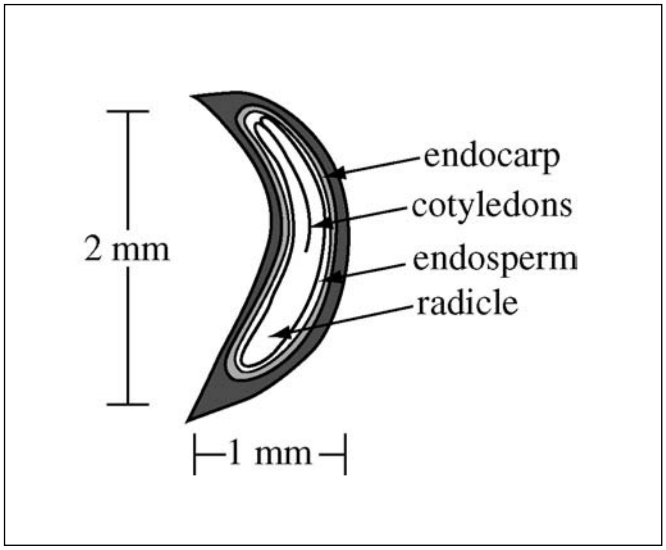
Growth habit, occurrence, and uses. American beautyberry—*Callicarpa americana* L.—is a small, woody shrub of the pine forests in the southern coastal plain. It seldom grows taller than 2 or 3 m. The shrub is common underneath the pine overstory and along roads and forest edges, where it grows best. 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.



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

William I. Stein

Dr. Stein is a forest ecologist emeritus at the USDA Forest Service's Pacific Northwest Research Station, Corvallis, Oregon

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 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).

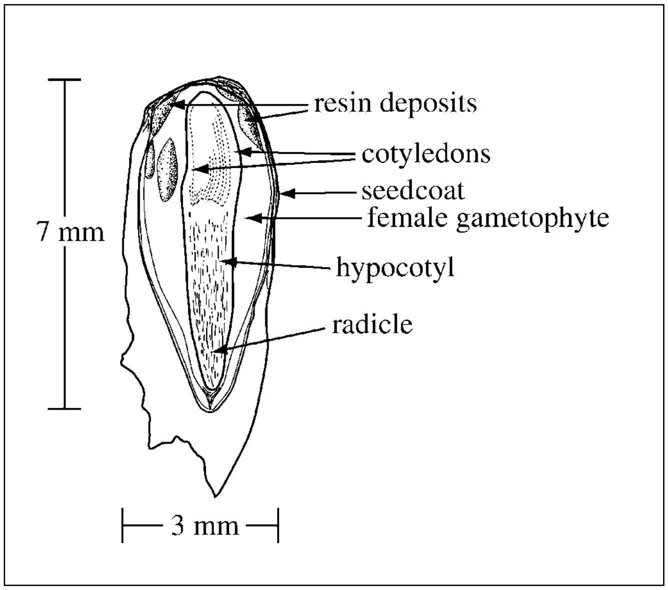
Seeds separate readily from well-opened cones; moderate tumbling or shaking is helpful. Whether done by improvised methods or in commercial machines, tumbling or shaking should be done gently, preferably at less than 27 °C, because seedcoats of incense-cedar are thin and easily broken.

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).

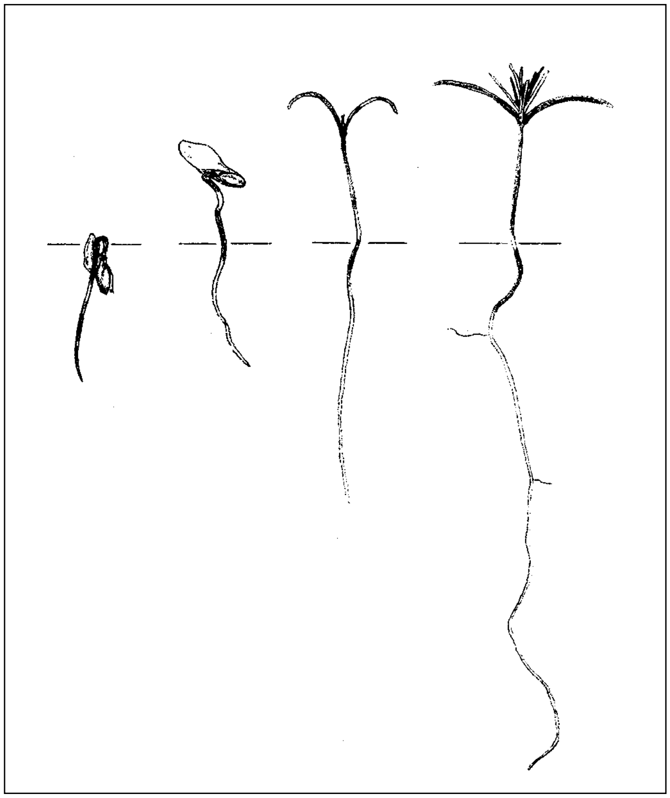
The winged seeds are usually hand-sown in rows. They should be covered about 6 to 12 mm ($^1/_4$ to $^1/_2$ in) deep (Show 1930). Burlap mulch proved satisfactory to keep seedbeds moist (Show 1930); sawdust or other mulch material and frequent sprinkler irrigation are currently used.

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

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station,
Mississippi State, Mississippi

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**).

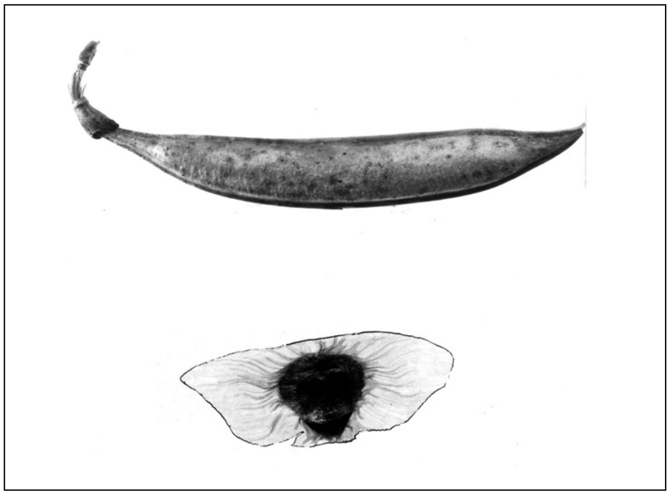
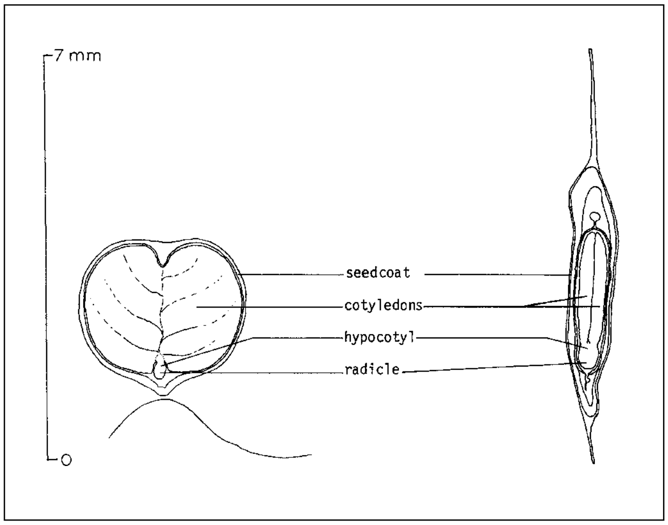


Figure 2—*Campsis radicans*, common trumpet-creeper: longitudinal section through a seed.



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).

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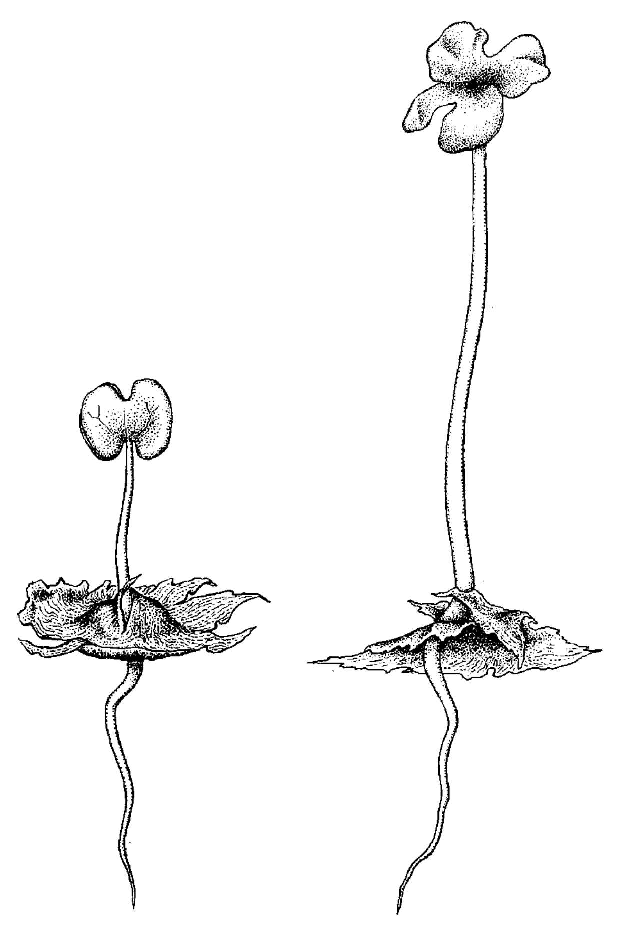
Figure 3—*Campsis radicans*, common trumpet-creeper: seedling development at 1 and 9 days after germination.



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.

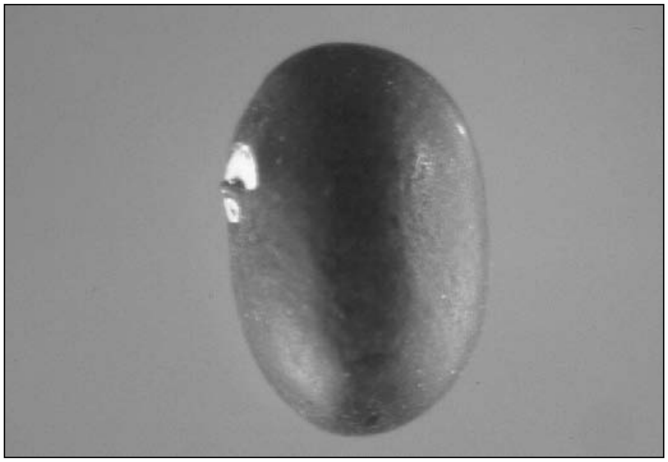
Growth habit, occurrence, and use. Siberian peashrub—*Caragana arborescens* Lam.—is one of the most hardy small deciduous trees or shrubs planted on the northern Great Plains (George 1953; Rehder 1940). Introduced into North America in 1752 (Rehder 1940), Siberian peashrub is native to Siberia and Manchuria 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).

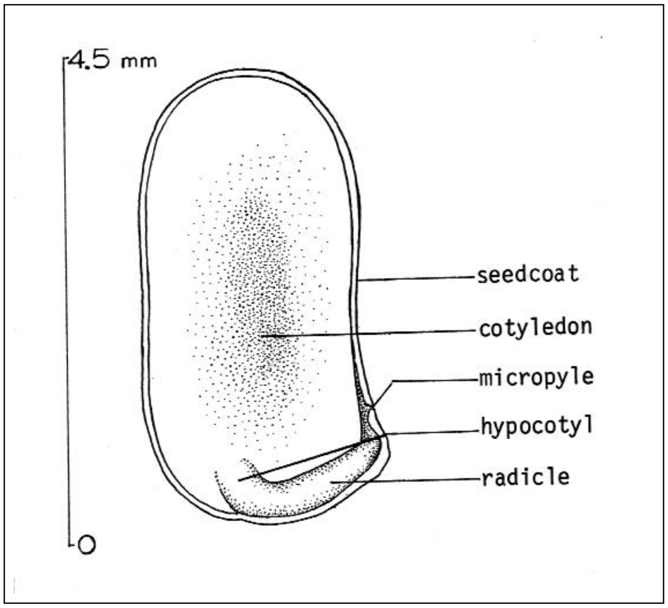
Figure 1—*Caragana arborsecens*, Siberian peashrub: legume.

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).

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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.

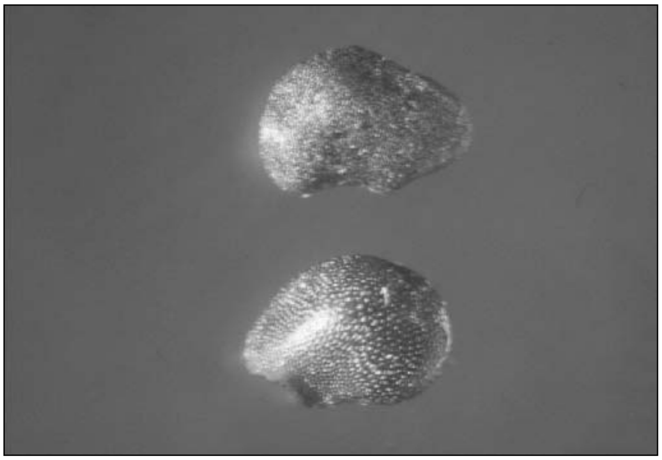
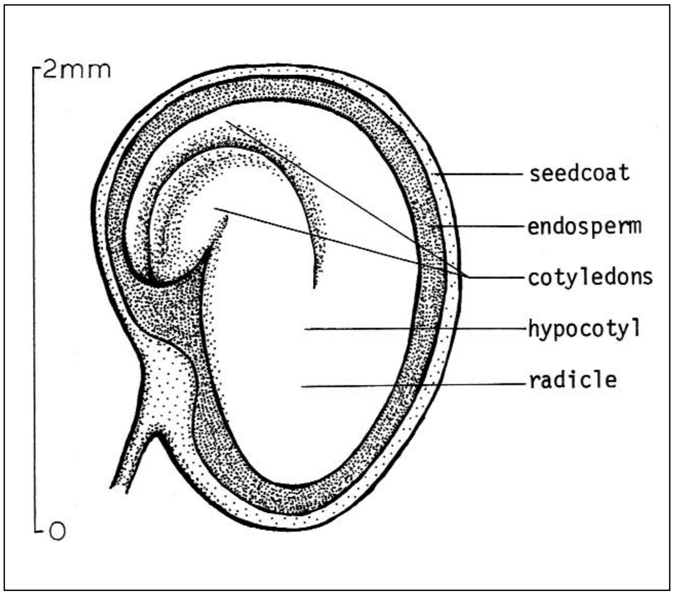


Figure 2—*Carnegiea giganteus*, saguaro: longitudinal section through a seed.



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

Dr. Neal retired from the USDA Forest Service's Pacific Southwest
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.

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**).

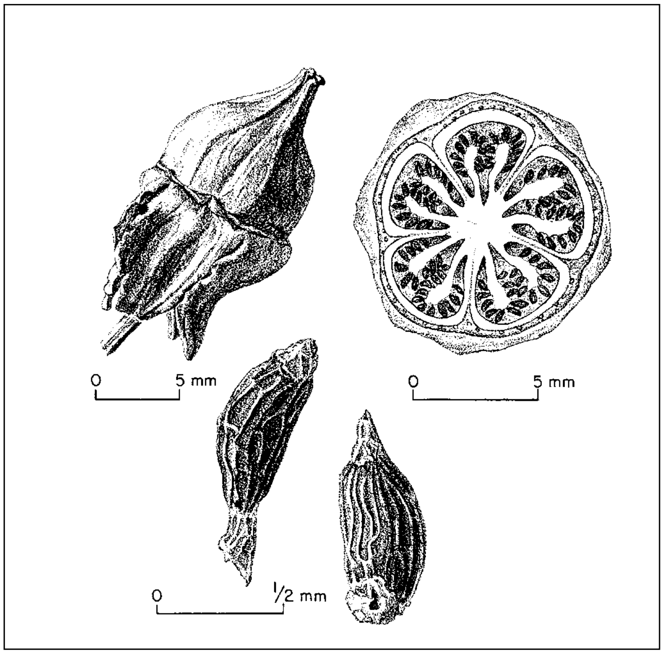
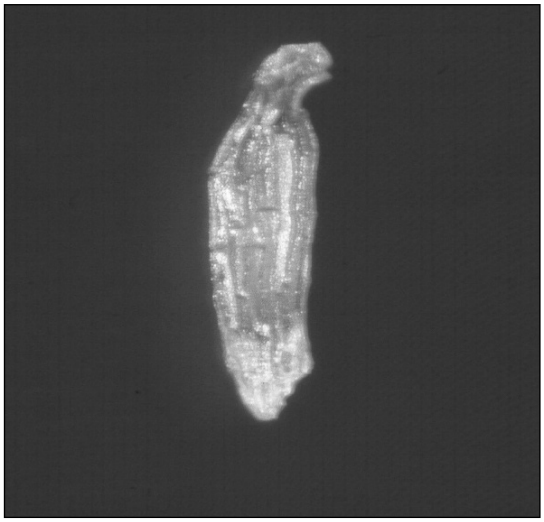


Figure 2—*Carpenteria californica*, carpenteria: seed.



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Betulaceae—Birch family

Carpinus L.

hornbeam or ironwood

Paula M. Pijut

Dr. Pijut is a plant physiologist at the USDA Forest Service's North Central Research Station, Hardwood Tree Improvement and Regeneration Center, West Lafayette, Indiana

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, "musclewood" (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; 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

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

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

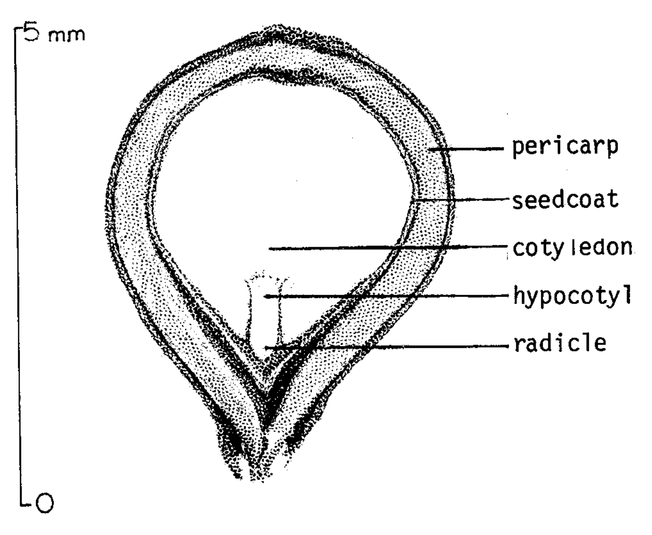

Table 2—*Carpinus,* carpinus: phenology of flowering and fruiting

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	/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

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*								
	Temp (°C)			Germination rate		% Germination		Purity	Soundness
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

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station,
Mississippi State, Mississippi

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).

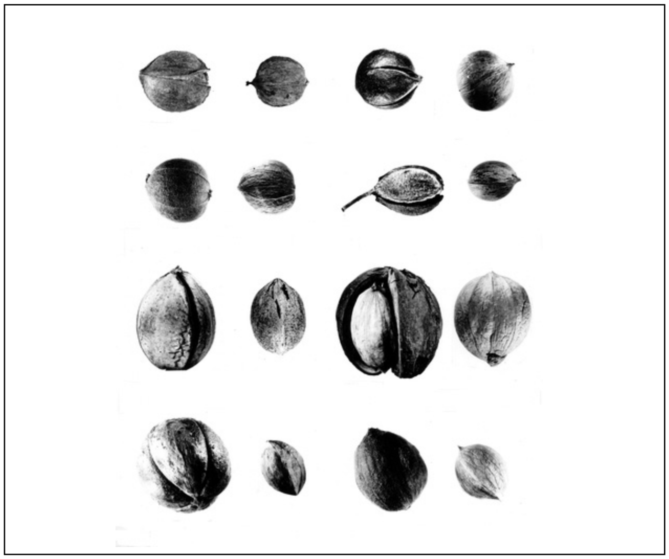
Table 2—*Carya,* hickory: phenology of flowering and fruiting

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.

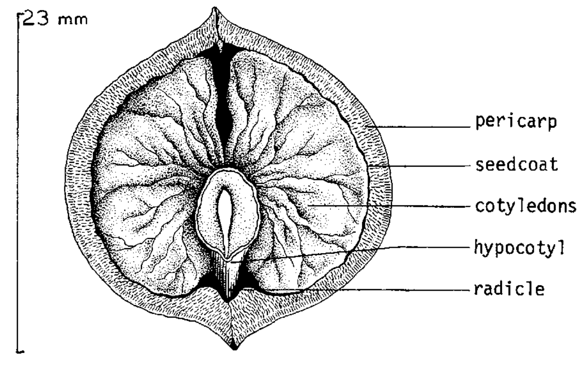


Table 3—*Carya,* hickory: height, seed-bearing age, seedcrop frequency, and year first cultivated

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

		Fruits/vol		Seed wt/fruit vol		Cleaned seeds/weight Range		Cleaned seeds/weight Average	
Species	Place 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 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

	Cold stratification	Germination test conditions				Germination		Germination %	
			Temp (°C)			Rate		Avg	
Species	(days)	Medium	Day	Night	Days	(%)	Days	(%)	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

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station,
Mississippi State, Mississippi

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	 /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.

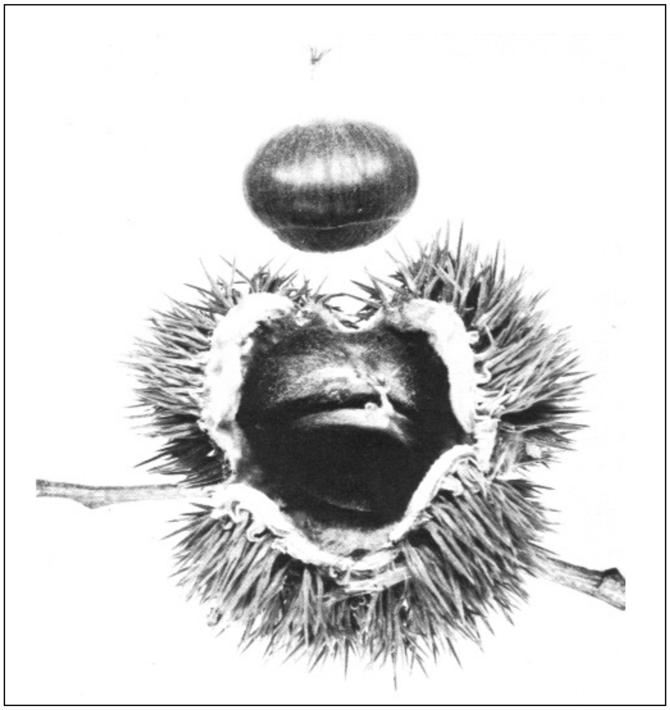

Figure 1—*Castanea dentata*, American chestnut: fruit (bur) and nut.

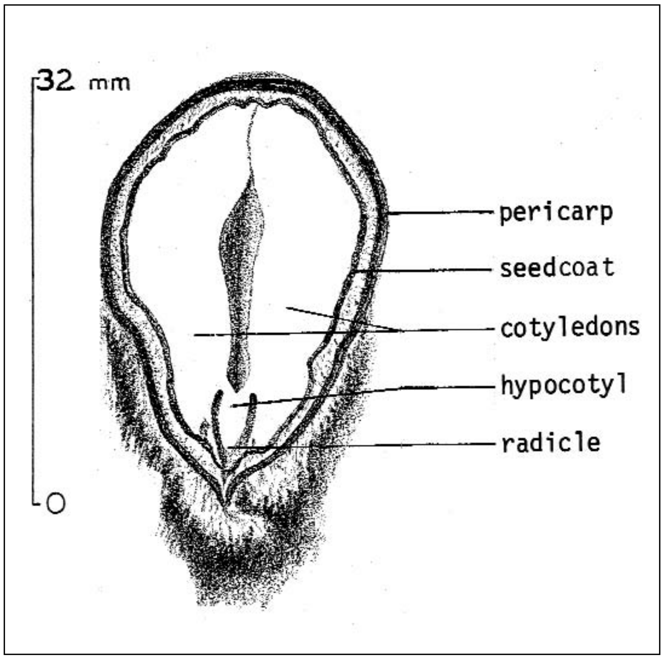

Figure 2—*Castanea dentata,* American chestnut: longitudinal section through a nut.

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 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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Casuarinaceae—Casuarina family

Casuarina Rumph. ex L.

casuarina

David F. Olson, Jr., E. Q. P. Petteys, and John A. Parrotta

Dr. Olson retired from the USDA Forest Service; Dr. Petteys is the district forester of Kauai, Hawaii Department of Lands and Natural Resources, Division of Forestry and Wildlife; Dr. Parrotta is a research program leader at the USDA Forest Service's Research and Development National Office, Arlington, Virginia

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.

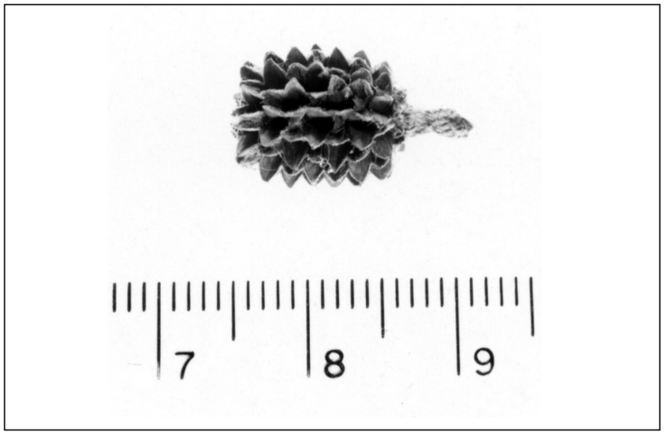


Figure 2—*Casuarina cunninghamiana*, river she-oak: longitudinal section through a seed.

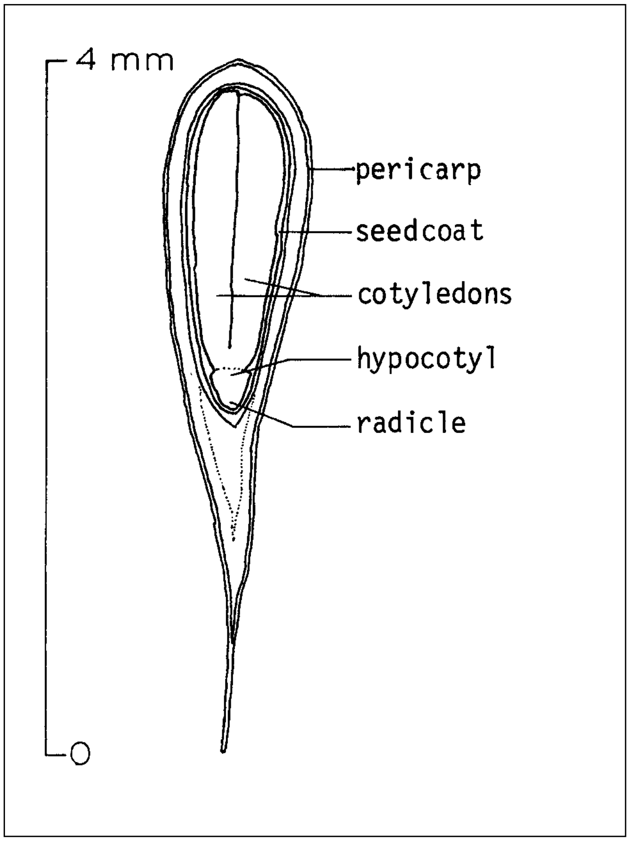
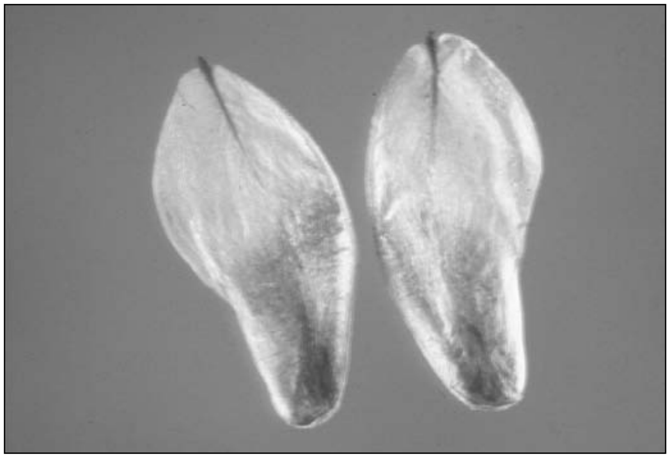


Figure 3—*Casuarina cunninghamiana*, river she-oak: winged seeds.



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.

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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Bignoniaceae—Trumpet-creeper family

Catalpa Scop.

catalpa

Franklin T. Bonner and David L. Graney

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station, Mississippi State, Mississippi; Dr. Graney is a research forester at the USDA Forest Service's Southern Research Station, Fayetteville, Arkansas

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.

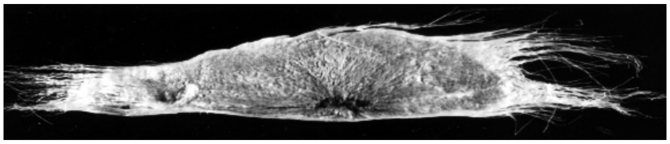
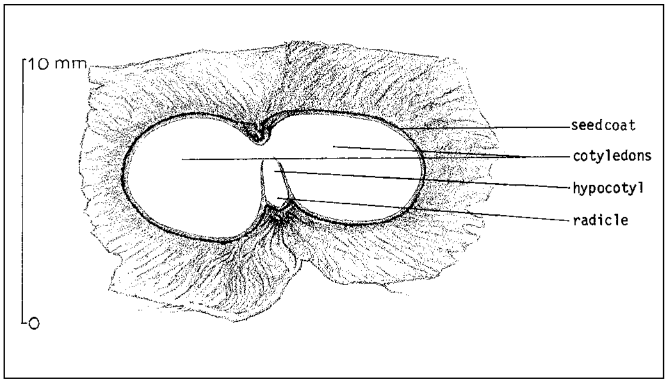


Figure 3—*Catalpa speciosa*, northern catalpa: longitudinal section through a seed.



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.

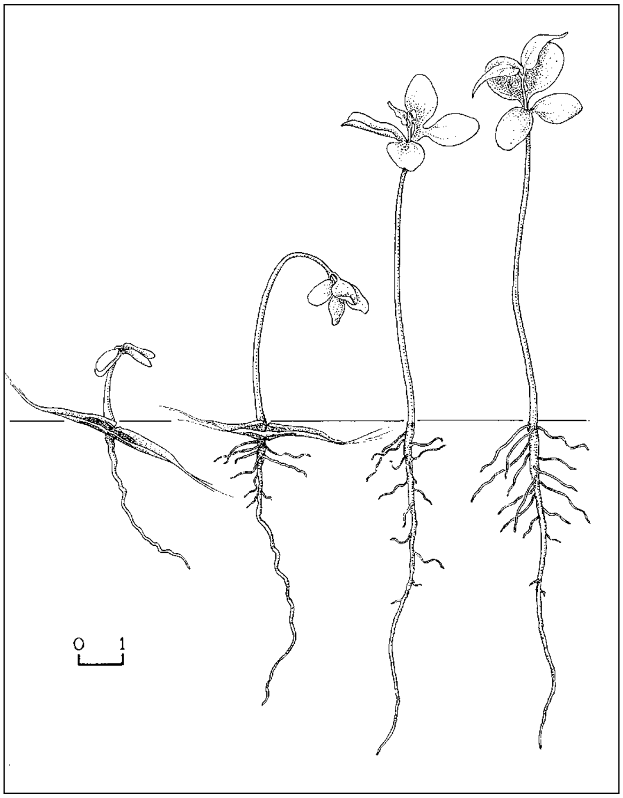


Table 2—*Catalpa,* catalpa: seed data

			Cleaned seeds/weight			
			Range		Average	
Species	**Place collected**	**Wt yield of seeds/ 100wt fruit**	**/kg**	**/lb**	**/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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Rhamnaceae—Buckthorn family

Ceanothus L.

ceanothus

Susan G. Conard and Merton J. Reed

Dr. Conard is a program leader at the USDA Forest Service's Research and Development National Office, Arlington, Virginia; Dr. Reed retired from the USDA Forest Service's Southwestern Forest and Range Experiment Station

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).

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

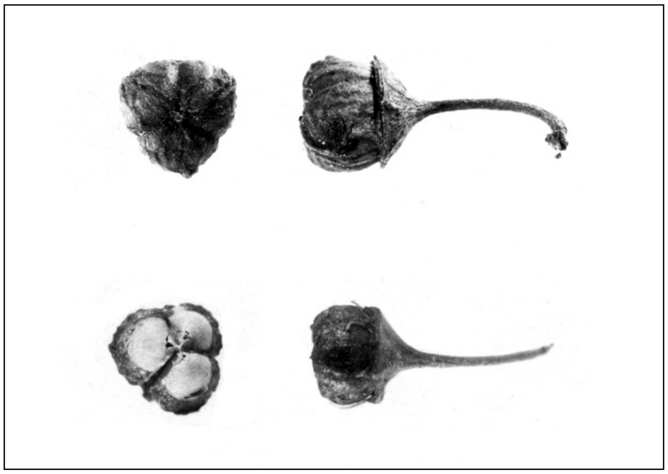


Figure 2—*Ceanothus*, ceanothus: seeds of *C. americanus*, New-Jersey-tea; *C. arboreus*, feltleaf ceanothus; *C. cordulatus*, mountain whitethorn; and *C. crassifolius* (**top, left to right**); *C. cuneatus*, buckbrush ceanothus; *C. impressus*, Santa Barbara ceanothus; *C. integerrimus*, deerbrush 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**).

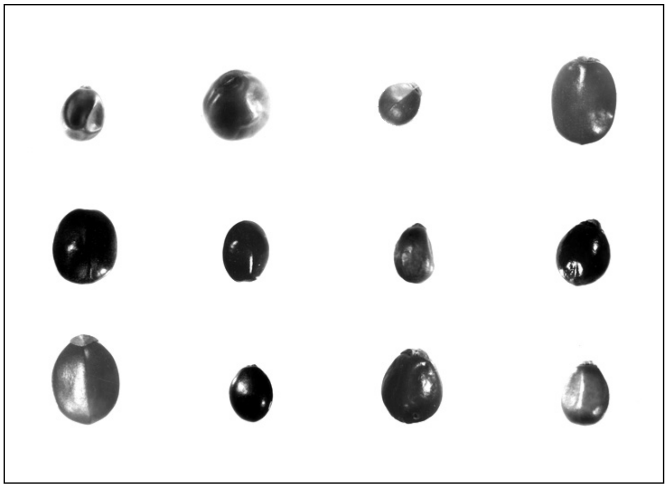
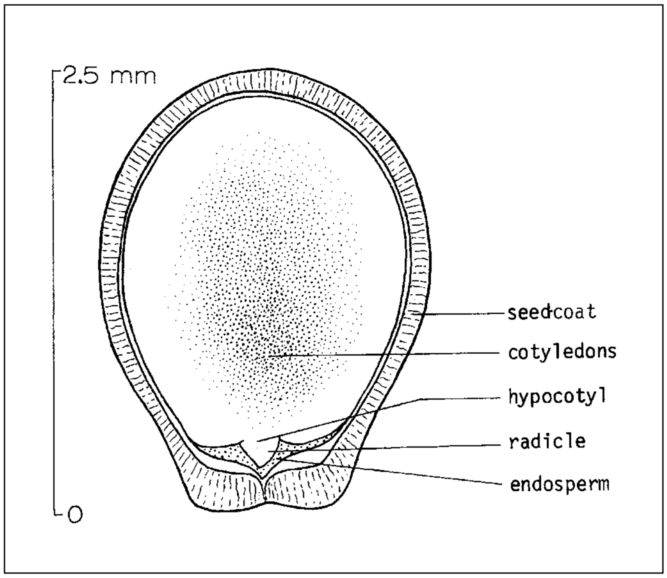


Figure 3—*Ceanothus americanus*, New-Jersey-tea: longitudinal section through a seed.



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, 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

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					
	Hot water soak				Germination rate	
Species	**Temp (°C)**	**Time* (min)**	**Cold 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.

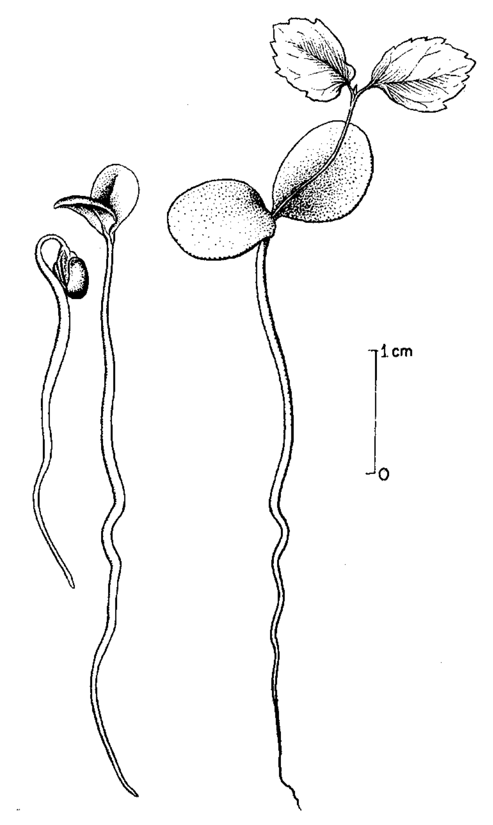
The genus includes both species that sprout vegetatively following fire (sprouters) and species that are killed by fire and reproduce only from seeds (obligate seeders). Obligate seeders appear to have overall higher germination following heat treatment and to tolerate higher temperatures and longer 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

Paula M. Pijut

Dr. Pijut is a plant physiologist at the USDA Forest Service's North Central Research Station,
Hardwood Tree Improvement and Regeneration Center, West Lafayette, Indiana

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).

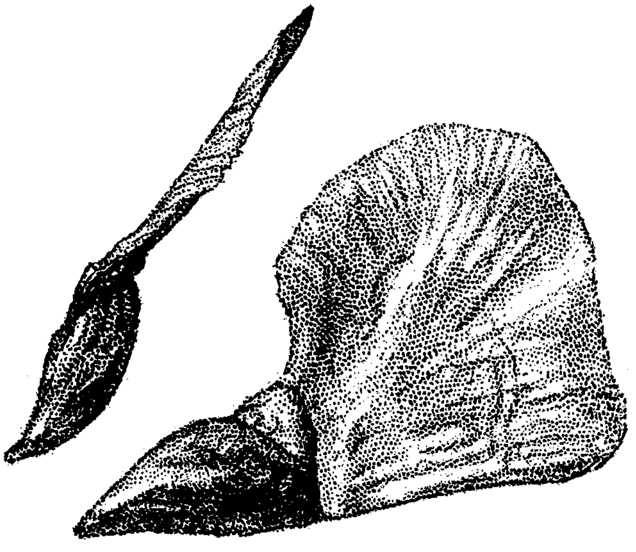
Even though cedar seeds are orthodox in storage behavior, they are very oily and do not keep well under many storage conditions (Allen 1995; Rudolf 1974). If cedar seeds are dried below a critical level, they will not imbibe water in a way that will allow the food reserves to be used by the embryo (Macdonald 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.



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 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).

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

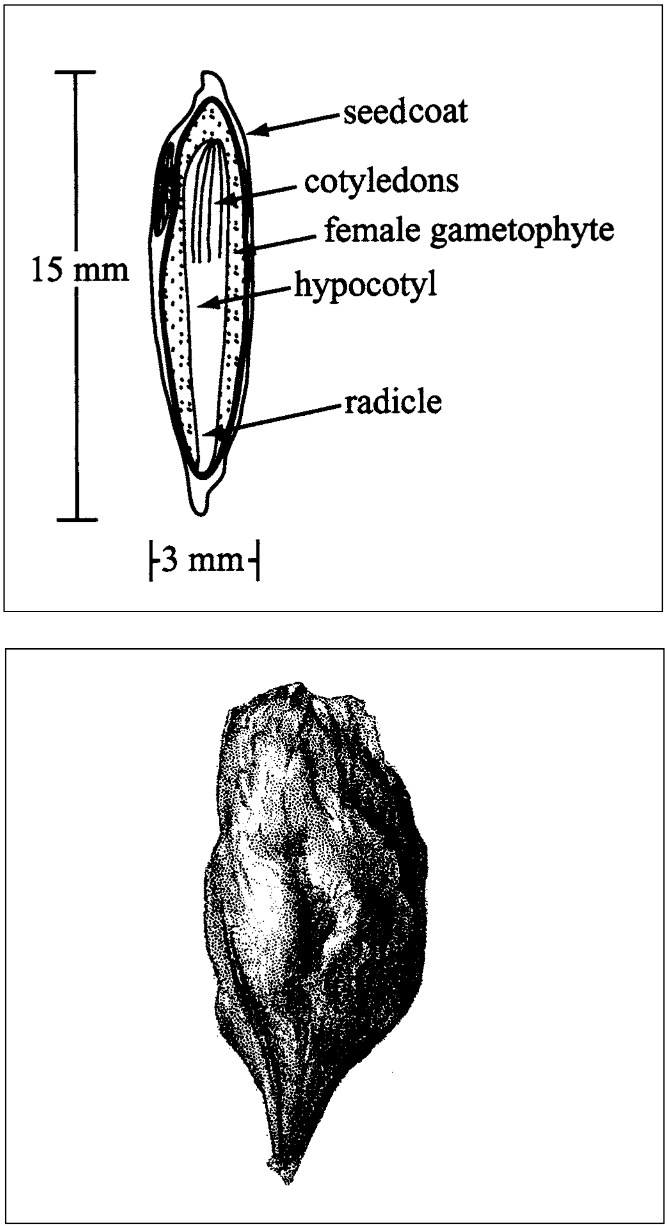
Sources: Rudolf (1974), Vidakovic (1991).

Table 4—*Cedrus,* cedar: seed data

Species	Avg no. cleaned seeds /kg	Avg no. cleaned seeds /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	—

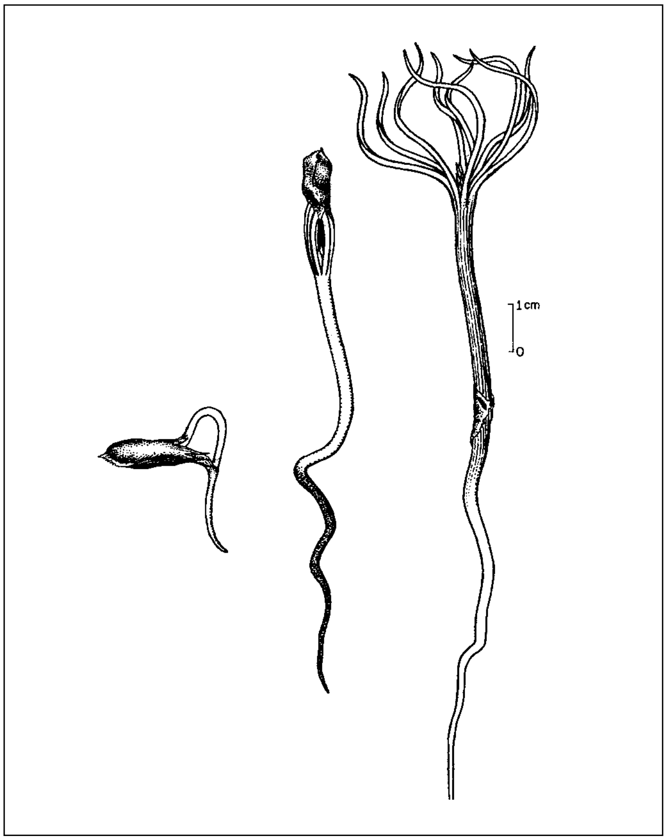
Sources: Allen (1995), Rudolf (1974).

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



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

G.W. Wendel, Jill R. Barbour, and Robert P. Karrfalt

Dr. Wendel retired from the USDA Forest Service's Northeastern Forest Experiment Station;
Ms. Barbour is a germination specialist at and Mr. Karrfalt is director of the USDA Forest Service's
National Seed Laboratory, Dry Branch, Georgia

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, polygamodioecious 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 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).

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

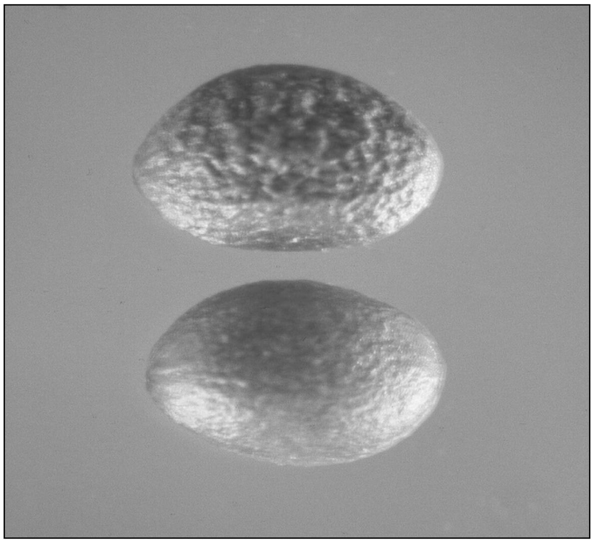
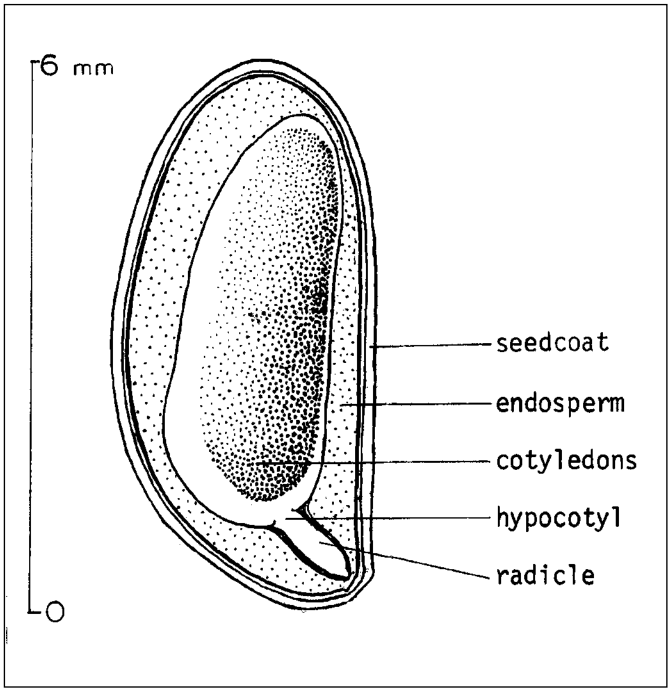


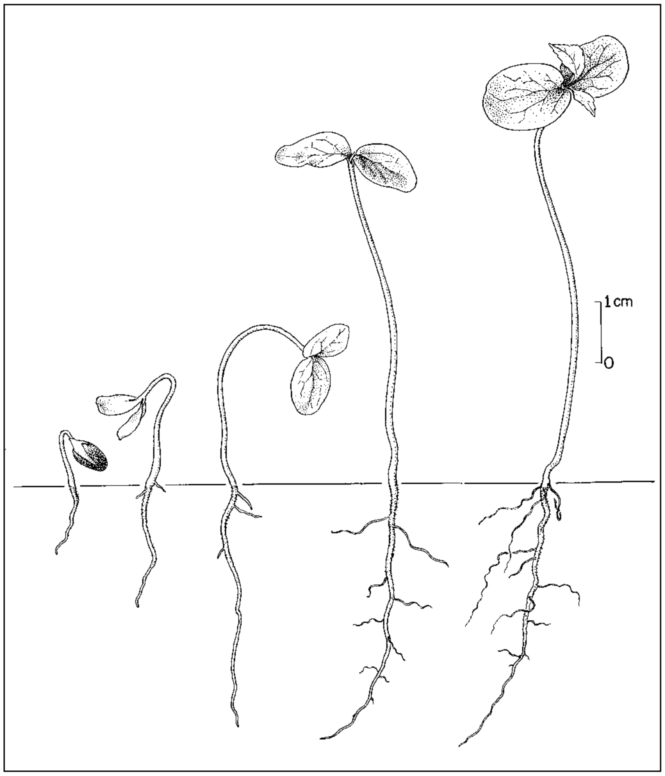
Figure 3—*Celastrus scandens,* American bittersweet: longitudinal section through a seed.



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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Ulmaceae—Elm family

Celtis L.

hackberry

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station,
Mississippi State, Mississippi

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).**

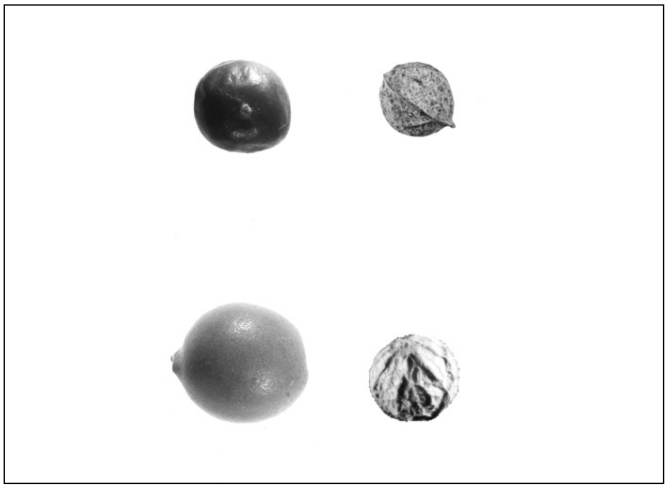
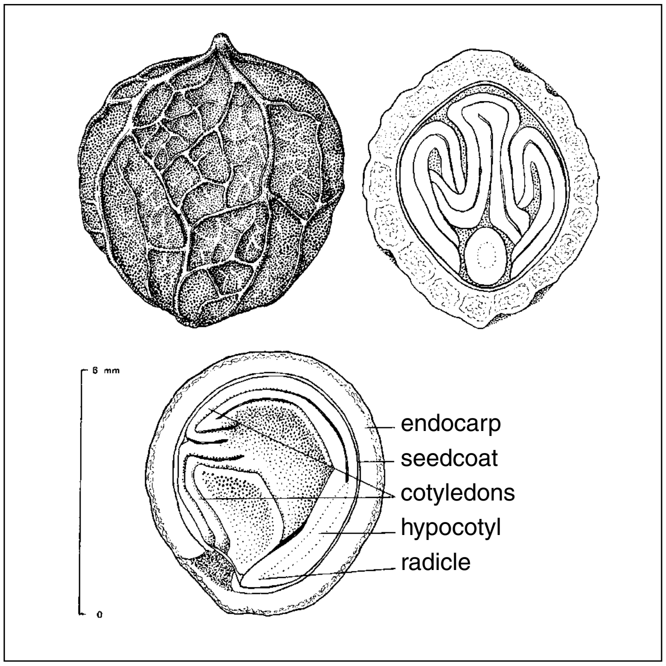
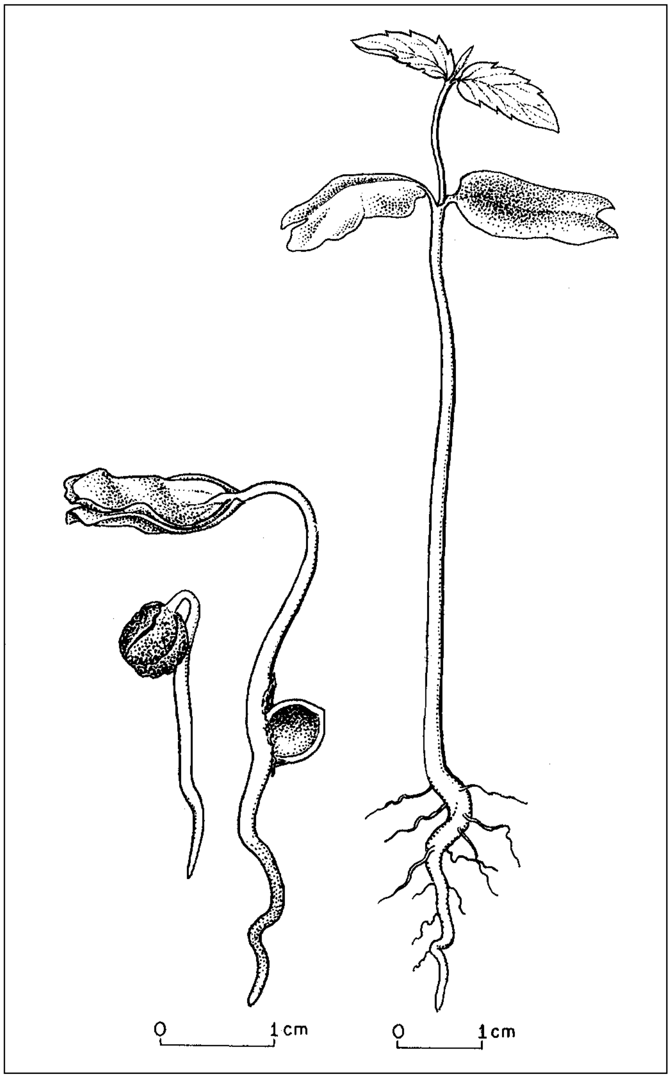


Figure 2—*Celtis occidentalis*, common hackberry: exterior view of seed (**top left**) and transverse section (**top right**) and cross section (**bottom**) of a seed.



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).

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	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

			Cleaned seeds/weight			
	Fruits/weight		Range		Average	
Species	/kg	/lb	/kg	/lb	/kg	/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

	Germinative test conditions *			Germination rate			
	Temp (°C)			Amount		Germination %	
Species	Day	Night	Days	(%)	Days	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

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station
Mississippi State, Mississippi

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).

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).

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

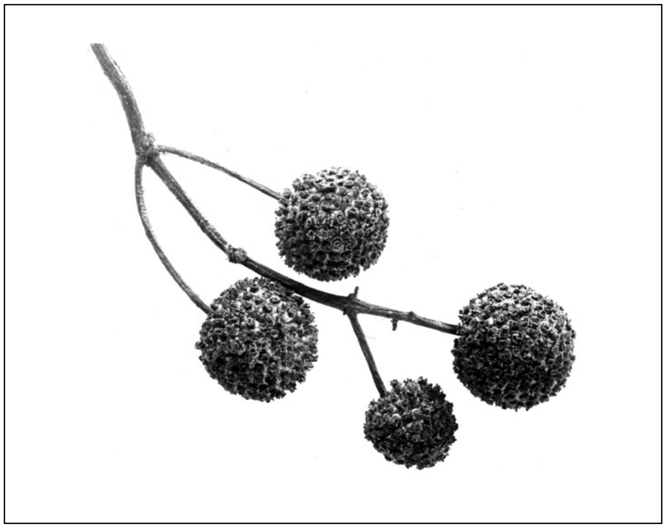
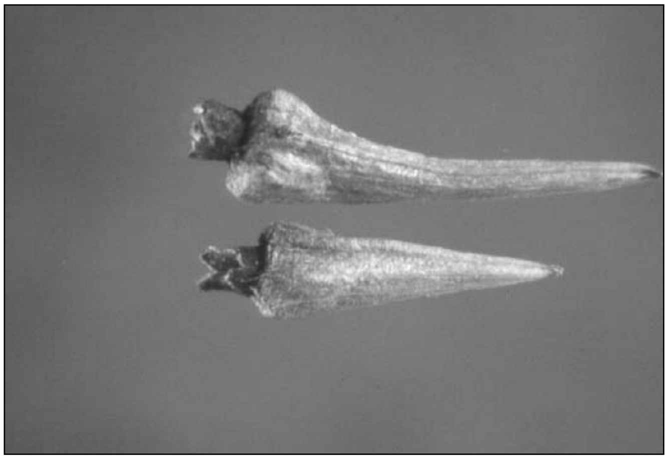


Figure 2—*Cephalanthus occidentalis*, common buttonbush: single fruits.



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.

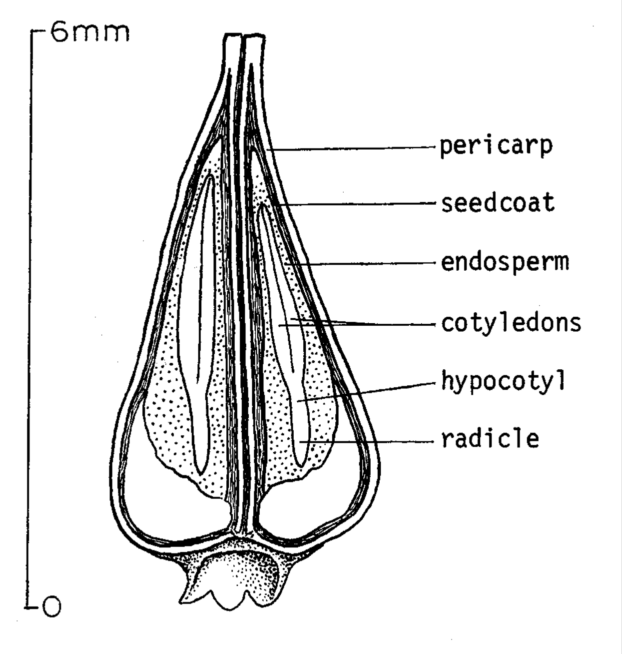
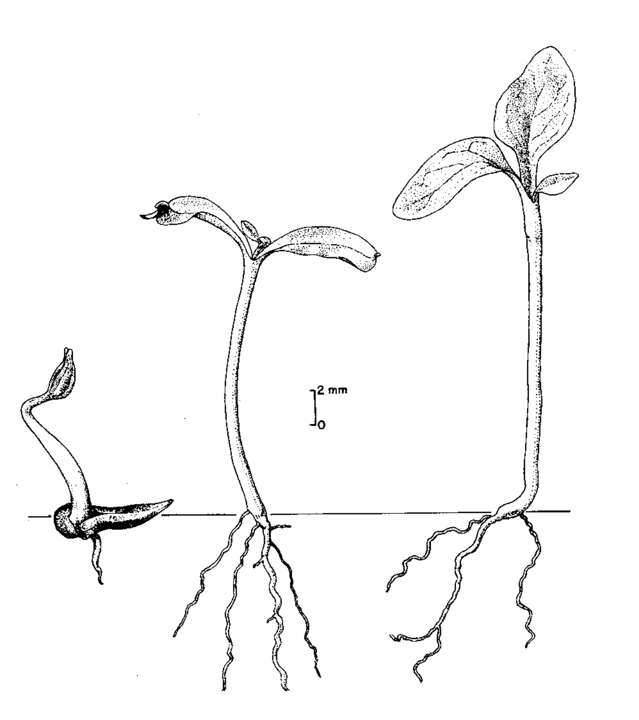


Figure 4—*Cephalanthus occidentalis,* common buttonbush: seedling development at 1, 23, and 40 days after germination.



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Fabaceae—Pea family

Ceratonia siliqua L.

carob

Wayne D. Shepperd

Dr. Shepperd is a principal silviculturist at the USDA Forest Service's Rocky Mountain Research Station, Fort Collins, Colorado

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.

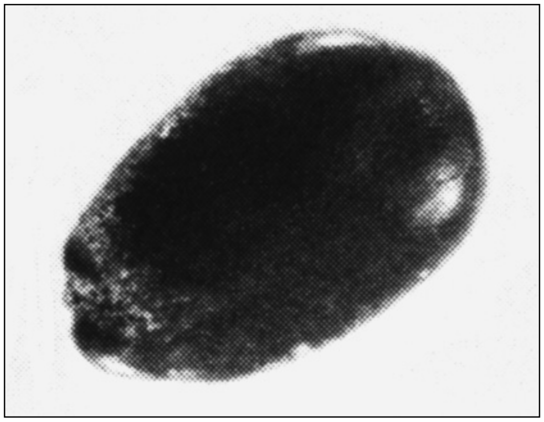
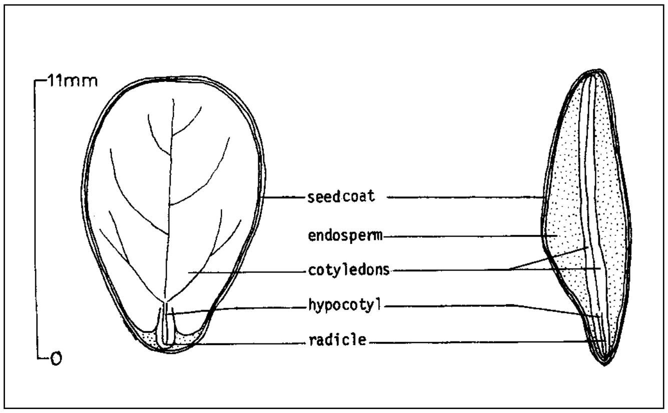


Figure 2—*Ceratonia siliqua*, carob: longitudinal sections through a seed.



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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Fabaceae—Pea family

Cercis L.

redbud

Valerie A. Banner and William I. Stein

Ms. Banner is a general biologist and Dr. Stein is a forest ecologist emeritus at the
USDA Forest Service's Pacific Northwest Research Station, Corvallis, Oregon

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**).

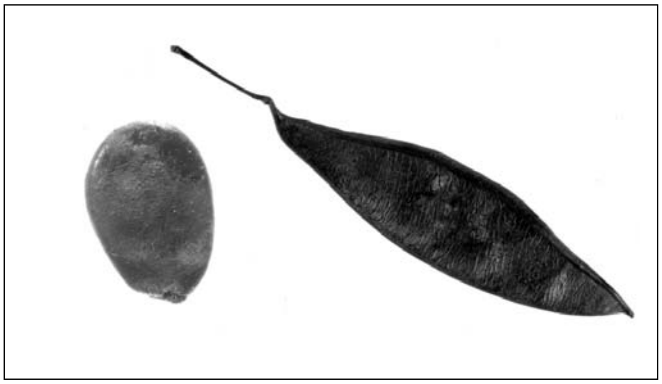
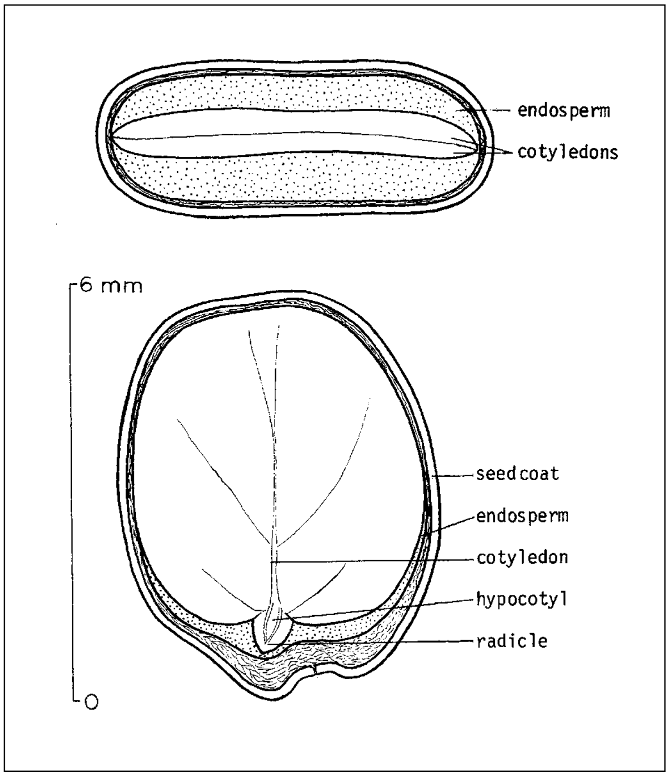


Figure 2—*Cercis canadensis*, eastern redbud: the flattened seed in transverse section (**above**) and longitudinal section (**below**).



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				
				Average		Range		
Species	Seeds/100 wt 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	Treat-ment	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.

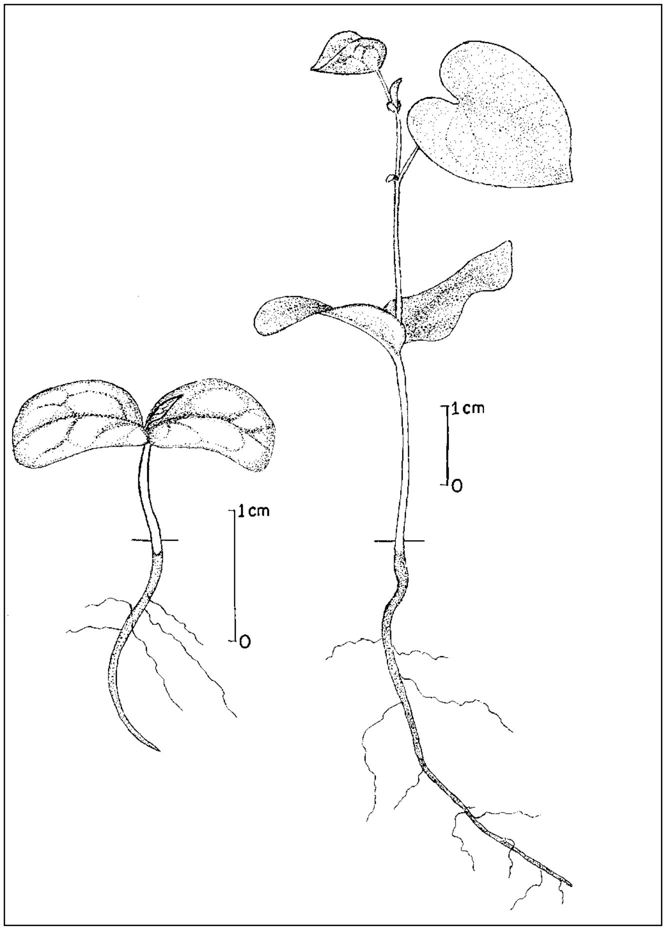
Surface-dried seeds are drill- or broadcast-sown and covered 0.6 to 2.5 cm (0.2 to 1 in) deep with soil, sand, sawdust, or bark. Some nurseries fumigate, then reinoculate before sowing seedbeds. Presence of an endomycorrhizal fungus is important; inoculation with *Glomus fasciculatum* (Thaxter) Gerdemann 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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Rosaceae—Rose family

Cercocarpus Kunth

mountain-mahogany

Stanley G. Kitchen

Mr. Kitchen is a botanist at the USDA Forest Service's Rocky Mountain Research Station,
Shrub Sciences Laboratory, Provo, Utah

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

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-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.

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

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).

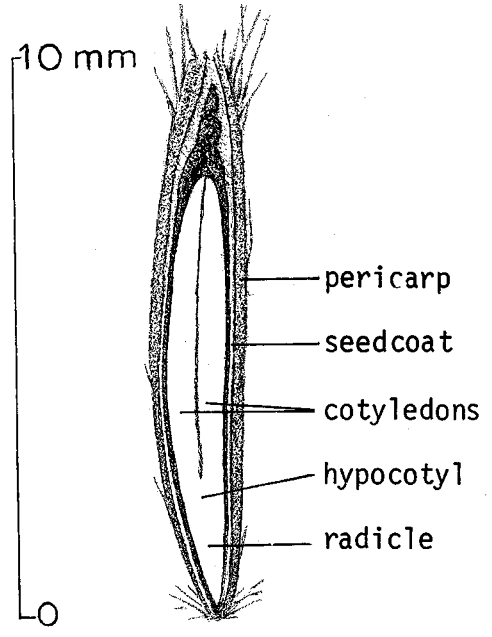
Cleaned-fruit sizes differ by species, ecotype, and year of collection. In one study, average number of fruits per weight for curlleaf (8 collections) and true mountain-mahoganies (10 collections) was 106,000 and 88,000/kg (48,000 and 40,000/lb), respectively (Kitchen and others 1989a&b). These fruit 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, and sulfuric acid (Dealy 1975; Stidham and others 1980; Young and others 1978).

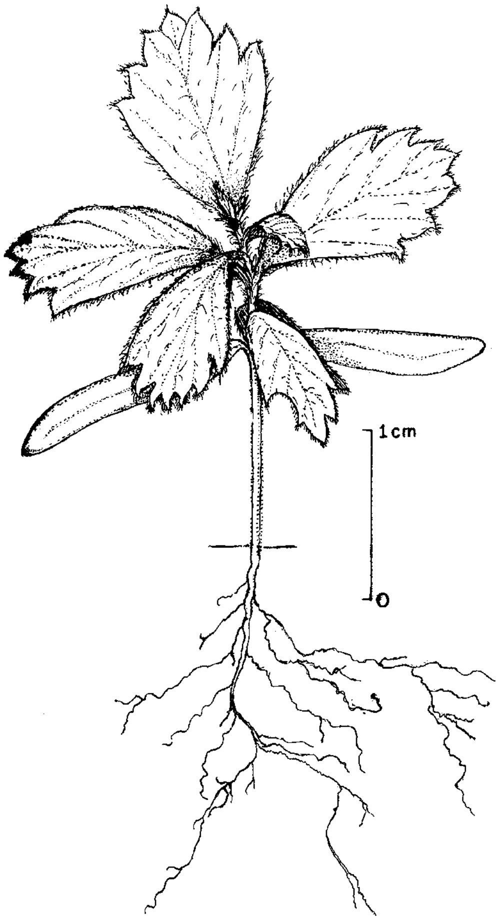
Figure 3—*Cercocarpus ledifolius*, curlleaf mountain-mahogany: longitudinal section through an achene.



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.



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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Rosaceae—Rose family

Chamaebatia foliolosa Benth.

bearmat

Arthur W. Magill and Susan E. Meyer

Dr. Magill retired from USDA Forest Service's Pacific Southwest Forest and Range Experiment Station;
Dr. Meyer is a research ecologist with the USDA Forest Service's Rocky Mountain Research Station,
Shrub Sciences Laboratory, Provo, Utah

Other common names. southern bearmat, mountain-misery, Sierra mountain-misery, San Diego mountain-misery, bearclover, tarweed, and running-oak.

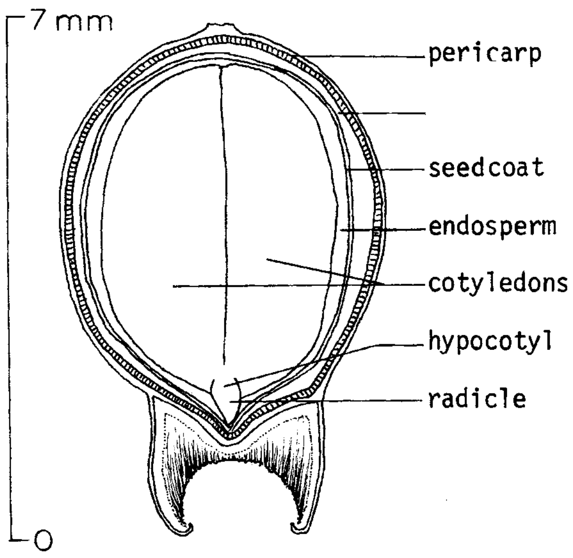
Growth habit, occurrence, and use. Two varieties of this species—*Chamaebatia foliolosa* Benth.—are recognized. The typical variety, bearmat, is an evergreen shrub, 15 to 60 cm tall, that grows between 600 and 2,100 m elevation on the western slopes of the Sierra Nevada in California. It occurs in open ponderosa pine (*Pinus ponderosa* Dougl. ex Laws.) and in California red fir *(Abies magnifica* A. Murr.) forests (Munz and Keck 1963). Southern bearmat—*C. foliolosa* var. *australis* Brandg.—grows to a height of nearly 2 m on dry slopes in the chaparral type from San Diego County to Baja California.

The typical variety is normally regarded as a pest because it inhibits the establishment and growth of trees (Adams 1969; Dayton 1931). From an aesthetic viewpoint, the plants can provide attractive ground cover, but their glutinous leaves are highly aromatic (Bailey 1928; McMinn 1959). It is useful for watershed stabilization and is a potential landscape plant (Magill 1974).

Flowering, seed production, and seed use. Bearmat produces perfect flowers throughout its range from May through July; southern bearmat flowers from November through May (McMinn 1959). The fruits are brown achenes about 5 mm in length (figures 1 and 2). Seeds require from 1 to 3 months of moist stratification at temperatures ranging from 1 to 5 °C before they will germinate (Emery 1964; Magill 1974). In the nursery, seeds should be sown in spring (Bailey 1928).

Figure 1—*Chamaebatia foliolosa*, bearmat: achene (**left**) and extracted seed (**right**).

Figure 2—*Chamaebatia foliolosa*, bearmat: longitudinal section through an achene.



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Rosaceae—Rose family

Chamaebatiaria millefolium (Torr.) Maxim.

fernbush

Nancy L. Shaw and Emerenciana G. Hurd

Dr. Shaw is a research botanist at the USDA Forest Service's Rocky Mountain Research Station, Boise, Idaho;
Dr. Hurd retired from the USDA Forest Service's Rocky Mountain Research Station

Other common names. desert sweet, fern-bush, desert-sweet.

Synonyms. *Spiraea millefolium* Torr., *Sorbaria millefolium* Focke, *Basilima millefolium* Greene, *Chamaebatiaria glutinosa* Rydb., and *Spiraea glutinosa* Fedde (Davis 1952; Hitchcock and others 1961; Peck 1961; Young and Young 1986).

Growth habit, occurence and use. Fernbush—*Chamaebatiaria millefolium* (Torr.) Maxim.—the only species in its genus, is endemic to the Great Basin, Colorado Plateau, and adjacent areas of the western United States. It is an upright, generally multistemmed, sweetly aromatic shrub 0.3 to 2 m tall. Bark of young branches is brown and becomes smooth and gray with age. Leaves are leathery, alternate, simple, bipinnatisect, stipulate, and more or less clustered near the ends of the branches. Foliage and young branches are viscid and pubescent, with simple and stellate hairs that are sharp-pointed or glandular-capitate. Southern populations are more or less evergreen (Phillips 1949), whereas northern populations are largely deciduous, retaining a few leaves near the branch tips through winter and initiating leaf development in early spring (Hitchcock and others 1961; Kirkwood 1930).

Fernbush is distributed east of the Cascade and Sierra Nevada Mountains from Deschutes Co., Oregon, to southern California and eastward across southern Oregon and Idaho, Nevada, Utah, northern Arizona, and New Mexico (Hitchcock and others 1961; Phillips 1949; Welsh and others 1987; Young and Young 1992). Fernbush is often present as an early successional species on cinder cones and basalt lava flows but is also found on soils derived from limestone and granite (Eggler 1941; Everett 1957; Merkle 1952). It occurs in cracks and fissures of rock outcrops and on well-drained soils of dry, rocky, gravelly canyons and mountain slopes at elevations ranging from 900 to 3,400 m (Albee and others 1988; Hickman 1993). Fernbush grows in isolated populations or as an associated species in sagebrush scrub (*Artemisia* spp.), sagebrush, northern juniper, mountain brush, aspen, limber pine, ponderosa pine, spruce–fir, and western bristlecone pine communities (Hickman 1993; Munz and Keck 1959; Welsh and others 1987).

Fernbush is occasionally browsed by mule deer (*Odocoileus hemionus*), sheep, and goats, but only rarely by cattle (Mozingo 1987; van Dersal 1938). Native Americans used a tea made from its leaves for treatment of stomach aches (Mozingo 1987).

Unlike its namesake genus—*Chamaebatia* Benth., bearmat or mountain misery—fernbush is not nodulated by nitrogen-fixing actinomycetes (McArthur and Sanderson 1985). Plants are cyanogenic (Fikenscher and others 1981). The species is a very rare host of juniper mistletoe—*Phoradendron juniperinum* Engelm. (Hawksworth and Mathiasen 1978).

First cultivated in 1878 (Rehder 1940), fernbush has long been recognized as an attractive ornamental because of its profuse and conspicuous inflorescences of white- to cream-colored flowers, long flowering season, and aromatic, fernlike foliage (Bailey 1902; Hitchcock and others 1961; Phillips 1949; Young and Young 1986). It is used effectively in mass plantings, xeriscapes, screens, and hedges when planted in full sun. Specimen plants provide color and texture accents (Phillips 1949).

Genetic variation, hybridization, and origin. McArthur (1984) and McArthur and others (1983) described *Chamaebatiaria* and other monotypic western North American genera of the Rosaceae as showing little variation compared to larger genera such as *Rosa* (rose) or *Cercocarpus* (mountain-mahogany). Typical of shrubby western North American members of subfamily Spiraeoideae, fernbush has x = n = 9 chromosomes (McArthur and Sanderson 1985). Hybridization of fernbush with other species has not been reported.

Chamaebatiaria (subfamily Spiraeoideae) was named for its morphologic resemblance to *Chamaebatia* (subfamily

Rosoideae). McArthur and Sanderson (1985) suggest that shrubby Spiraeoideae and Rosoideae of western North America may be rather closely related based on similarities in morphologic and other characteristics of the 2 groups. Wolfe and Schorn (1989) and Wolfe and Wehr (1988) discuss evidence from Paleogene montane floras of the Rocky Mountains indicating the possible divergence of *Chamaebatiaria* and *Chamaebatia* from a common Eocene ancestor. They suggest both lines adapted to progressively drier post-Eocene conditions than the mesic coniferous forest environment inhabited by the ancestor.

Flowering and fruiting. The showy, white, insect-pollinated flowers develop in profuse, terminal, leafy-bracteate panicles up to 20 cm in length. Flowers are complete, regular, and about 0.8 to 1.5 cm in diameter. The calyx consists of 5 persistent green sepals. A glandular disk lining the hypanthium bears 5 petals and numerous stamens. Pistils are 5 (rarely 4), ovaries superior, and styles free. The ovaries are more or less connate below in flower, but separate in fruit. The pubescent, coriaceous, few-seeded follicles dehisce along the ventral suture and upper half of the dorsal suture (figure 1). Seeds are erect, yellowish to brownish, linear to narrowly fusiform, and somewhat flattened at each end (figure 2). The outer layer of the soft thin seedcoat is ridged, giving the body of the seed a 3-angled appearance; the inner layer is thin and translucent. A fleshy endosperm layer adheres to the seedcoat. The embryo is linear-oblong with 2 flat cotyledons and occupies the central portion of the seed (figure 3). Germination is epigeal (Hickman 1993; Hitchcock and others 1961; Hurd 1995; Kirkwood 1930; Welch and others 1987).

Figure 1—*Chamaebatiaria millefolium,* fernbush: follicle.

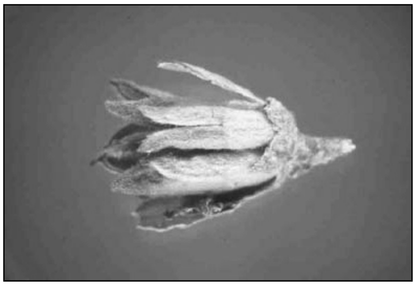


Figure 2—*Chamaebatiaria millefolium*, fernbush: seeds.

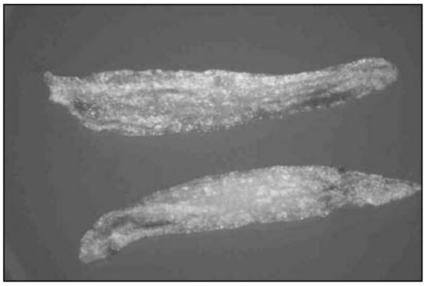
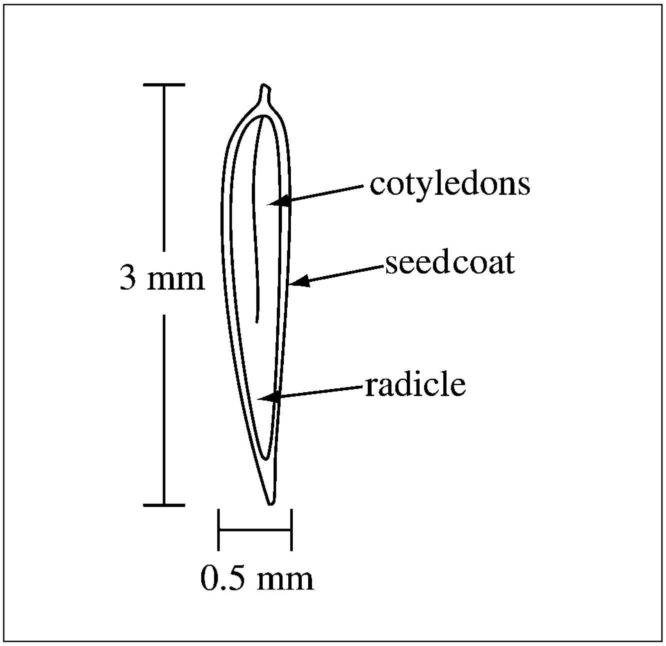


Figure 3—*Chamaebatiaria millefolium*, fernbush: longitudinal section through a seed.



Irrigated plants may begin flowering during the second growing season (Shaw 1995). Plants flower from June to September (Hitchcock and others 1961; Phillips 1949) with irrigation prolonging the flowering season (Shaw 1995). Fruits ripen from August to October.

Collection of fruits, seed extraction, cleaning, and storage. Fruits are harvested by clipping or stripping inflorescences when they are dry and brown, but before follicles open. Seeds can also be collected by briskly shaking or beating the inflorescences once the follicles begin dehiscing. Most follicles open during air-drying, releasing the seeds. Debris may then be removed with screens or a seed blower. Larger collections may be cleaned using air-screen machines. For 2 Idaho seedlots produced with irrigation, the number of seeds per seed weight averaged 3,700,000/kg (1,700,000/lb) (Hurd 1995). Storage requirements and seed longevity have not been determined, but the seeds are probably orthodox in storage behavior.

Pregermination treatments and germination and viability tests. Fresh seeds are nondormant, whereas stored seeds require 1 to 3 months of chilling to relieve dormancy (McDorman 1994; Phillips 1949; Young and Young 1986, 1992). The optimum temperature range for germination of southwestern populations is 18 to 26 °C (Phillips 1949).

Fernbush germination has received little study. Shaw (1995) examined the germination of 3 seed collections. Nampa, ID, and Sun Valley, ID, collections were harvested from irrigated plantings of seeds from a single unknown source. The third collection was from an irrigated Sante Fe, NM, planting of seeds from a western New Mexico source. All 3 collections were cleaned and held in dry storage for 4 to 5 months before testing. Total germination percentage of the Sante Fe, NM, and Sun Valley, ID, seed collections (but not the Nampa, ID, seed collection) was greater when untreated seeds were incubated at 20/10 °C (8 hours/16 hours) than at 15 °C for 28 days. A 28-day wet chilling at 3 to 5 °C (table 1) improved the total germination percentage of all seed collections when they were subsequently incubated at either 15 °C or 20/10 °C for 28 days.

Viability of fernbush seeds may be tested as follows: first, the seeds are soaked in water at room temperature for 1 hour, then the water is drained away. A horizontal slit should be made across the center of each seed without cutting it in half. Seeds are then submerged in a 1% solution of 2,3,5-triphenyl tetrazolium chloride for 6 hours at room temperature. Evaluate as described by Peters (2000) for Rosaceae III. The embryos may be read in place. The endosperm of viable seeds is living and will stain red (Hurd 1995).

Nursery practice. Nursery plantings should be made in late fall or early winter. As an alternative, artificially wet-chilled seeds may be planted in early spring. Fernbush seeds are small and must be sown on the soil surface or with a very light covering of sand or soil. Seedlings develop rapidly with irrigation and reach an adequate size for lifting after 1 growing season (Shaw 1995).

Seeds for production of container stock should be wet-chilled before planting. Survival of germinants moved from seeding flats to production containers is low (Everett 1957). Better establishment is obtained by sowing seeds directly into containers and thinning to 1 seedling per container. Developing seedlings are easily moved from small to larger containers (Phillips 1949). Seedlings should be grown in a well-drained medium.

Direct seeding. Seeds should be planted in fall or early winter. Seedlings emerge in spring from seeds naturally dispersed in late summer on rough or mulched soil surfaces (Mackie 1995; McDorman 1994; Shaw 1995). Naturally occurring seedlings generally establish where vegetative competition is limited (Shaw 1995).

Table 1—*Chamaebatiaria millefolium,* fernbush: germination test conditions and results

Source	Elevation (m)	Origin	Cold, wet chill (days)*	% Germination‡ 15 °C Incub†	% Germination‡ 20/10 °C Incub†	Seed fill (%)	Seed viability (%)
Nampa, ID	831	Unknown§	0	3	1	100	96
			28	72	65	100	96
Sun Valley, ID	1,773	Unknown§	0	12	20	100	86
			28	33	44	100	86
Santa Fe, NM	2,134	W New Mexico	0	9	22	100	85
			28	58	60	100	85

* Chilling temperature = 3 to 5 °C.

† Incub = incubation time = 28 days; seeds were exposed to 8 hours of light (PAR = 350 M m/sec) each day with temperatures of either constant 15 °C or 8 hours of 20 °C and 16 hours of 10 °C. In the alternating temperature regime, plants were exposed to light during the high-temperature period.

‡ Based on the percentage of viable seeds to germinate normally.

§ The Nampa and Sun Valley, ID, plants were grown from the same unknown seed source.

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Cupressaceae—Cypress family

Chamaecyparis Spach

white-cedar

Franklin T. Bonner

Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Research Station, Mississippi State, Mississippi

Growth habit, occurrence, and use. The genus *Chamaecyparis* occurs naturally on the Atlantic and Pacific Coasts of North America and in Japan and Taiwan. Three species are native to North America, 2 to Japan, and 1 to Taiwan (Sargent 1965). The North American species (table 1) are long-lived evergreens that attain large size. Port-Orford-cedar, the largest, has reached diameters of more than 1 m and heights of near 70 m in old-growth stands (Zobel 1990a). Branching is distinctive, with many-branched twigs and small paired scalelike leaves arranged in fernlike sprays. Another common name is "false cypress" (Little 1979); they are not true cedars (*Cedrus* spp.). Because of their somber beauty and variety of form, white-cedars are often used for ornamental plantings, hedges, and windbreaks (Rehder 1940). They produce valuable timber, the wood being sought for poles, posts, construction timbers, specialty items, and other uses where durability is desired. Atlantic white-cedar wood is especially popular for boats, outdoor furniture, posts, and utility poles (Kuser and Zimmerman 1995).

Geographic races and hybrids. Two geographic races of Atlantic white-cedar have been proposed: var. *henryae* (Li) Little in Georgia, Florida, Alabama, and Mississippi and var. *thyoides* in the area from South Carolina to Maine (Little 1966). Great variation exists within the genus, and numerous horticultural selections have been made of the 3 North America species as well as the Asian ones (Dirr and Heuser 1987; Harris 1990; Little and Garrett 1990; Zobel 1990a). Both interspecific and intergeneric crosses have been successful with certain of the cedars. Alaska-cedar and 2 of the Asian species have been crossed (Yamamoto 1981), and Alaska-cedar has also been crossed with several species of Cupressus (Harris 1990). The most well-known of these crosses is with Monterey cypress (*Cupressus macrocarpa* Hartw. ex Gord.) to produce the widely planted Leyland cypress (*Cupressocyparis* × *leylandii*).

Flowering and fruiting. White-cedars are monoecious. Their tiny inconspicuous yellow or reddish male pollen-bearing flowers and greenish female flowers are borne on the tips of branchlets (Harris 1974). Staminate flowers of Atlantic white-cedar, for example, are about 3 mm long, and the pistillate flowers are approximately 3 mm in diameter (Little and Garrett 1990). Pollination occurs generally from March to May, and cones ripen in September to October. Cones are slow to open fully, and seed dispersal occurs from fall into the following spring (table 2). Cones of Port-Orford-cedar and Atlantic white-

Table 1—*Chamaecyparis,* white-cedar: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
***C. lawsoniana* (A. Murr.) Parl.** *Cupressus lawsoniana* A. Murr.	**Port-Orford-cedar,** false cypress, Lawson cypress, Oregon-cedar, Port-Orford white-cedar	SW Oregon (Coos Bay) S to NW California (Klamath River)
***C. nootkatensis* (D. Don.) Spach** *Cupressus nootkatensis* D. Don	**Alaska-cedar,** yellow-cedar, Alaska yellow-cedar, Nootka yellow-cypress, Sitka cypress, yellow cypress	Pacific Coast region from Prince William Sound, Alaska, SW to W British Columbia & W Washington, & S in Cascade Mtns to W & NW & SW British Columbia to California; local in NE Oregon
***C. thyoides* (L.) B.S.P.** *Cupressus thyoides* L	**Atlantic white-cedar,** white-cedar, swamp-cedar, southern white-cedar	Narrow coastal belt from S Maine to N Florida, W to S Mississippi

Source: Little (1979).

Table 2—*Chamaecyparis,* white-cedar: phenology of flowering and fruiting

Species	Location	Flowering	Cone ripening	Seed dispersal
C. lawsoniana	Oregon	March	Sept–Oct	Sept–May
C. nootkatensis	Pacific Coast	Apr–May	Sept–Oct*	Oct–spring
C. thyoides	Atlantic Coast	Mar–Apr	Sept–Oct	Oct 15–Mar 1

Sources: Harris (1974), Little (1940).

* Cones require 2 years to reach maturity in the northern part of the range.

cedar mature the same year that they are pollinated, whereas cones of Alaska-cedar, in most of the species' range, take a second year to complete maturation (Harris 1974, 1990). In the extreme southern portion of the range of Alaska-cedar, cones may mature in only 1 year (Owens and Molder 1975). This condition even occurs on trees from more northern origins or from higher elevations when established in seed orchards in warm, southern, coastal sites (El-Kassaby and others 1991). The seeds from these 1-year cones are of size and germination quality equal to seeds from 2-year cones.

The white-cedars bear cones at an early age—5 to 20 years for Port-Orford-cedar (Zobel 1990a) and 3 to 5 years for Atlantic white-cedar (Little and Garrett 1990). Sprays of gibberellin (primarily GA_3) will induce flowering in even younger seedlings of Port-Orford-cedar and Alaska-cedar (Owens and Molder 1977; Pharis and Kuo 1977). The use of GA_3 and supplemental pollination on container-grown Port-Ordford-cedar trees 4 to 6 years from rooting or grafting has shown good potential to produce a large amount of seeds in a short period (Elliott and Sniezko 2000). Mature cones are 6 to 12 mm in diameter, spherical, and are borne erect on branchlets (figure 1). Cones have from 6 to 12 scales, each bearing from 1 to 5 seeds with thin marginal wings (figures 2 and 3) (Harris 1974). The average number of seeds per cone is 7 for Alaska-cedar (Harris 1990) and 8 for Atlantic white-cedar, but less than a third of these seeds may be filled. With controlled crosses in a seed orchard, Port-Orford-cedar averaged as high as 8.6 filled seeds per cone (Elliott and Sniezko 2000). Cone ripeness is normally determined by their exterior color (table 3).

Seedcrops of both western white-cedars can be damaged by larvae of the seedworm *Laspeyresia cupressana* (Kearfott) feeding on seeds in the cones. Larvae of the incense-cedar tip moth—*Argyresthia libocedrella* Busck—mine the cones and seeds of Port-Orford-cedar and can destroy practically the entire seedcrop (Hedlin and others 1980).

Collection of cones. Cones may be collected by hand or raked from the branchlets of standing or felled trees. As with many species, cone production is usually less in dense stands, although local conditions cause much variation (Zobel 1979), and open stands should be favored in collections from natural stands. In a North Carolina study,

Figure 1—*Chamaecyparis nootkatensis,* Alaska-cedar: mature cones.

Figure 2—*Chamaecyparis thyoides,* Atlantic white-cedar: seeds.

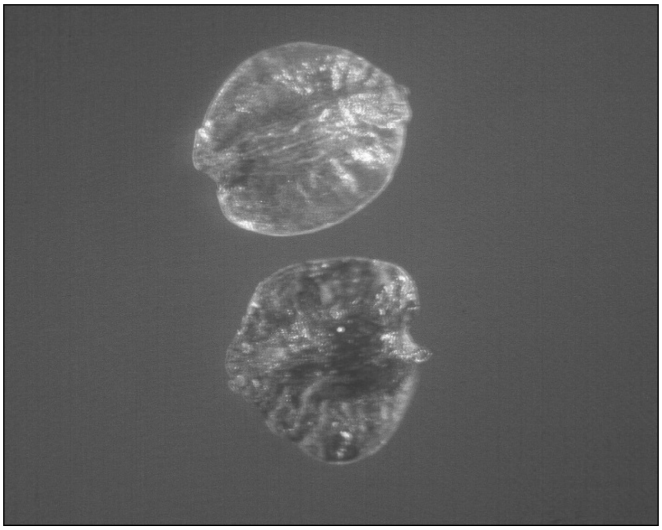
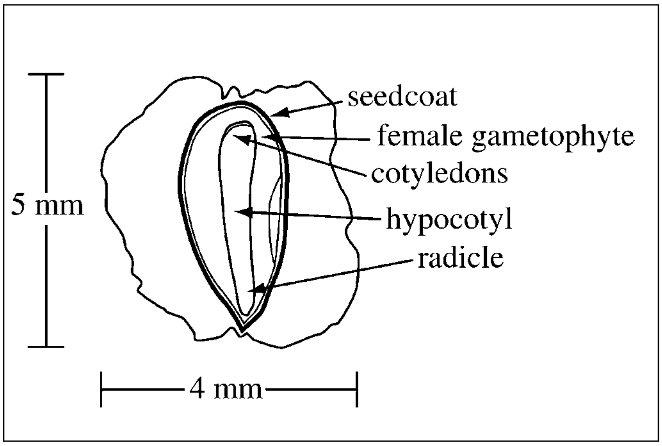


Table 3—*Chamaecyparis,* white-cedar: height, seed-bearing age, seed crop frequency, and color of ripe cones

Species	Height at maturity (m)	Year first cultivated	Minimum seed-bearing age (yrs)	Years between large seedcrops	Color of ripe cones
C. lawsoniana	to 73	1854	5–20	3–5	Greenish yellow to red brown
C. nootkatensis	to 53	1851	—	4 or more	Yellow brown to red brown
C. thyoides	12–27	1727	3–20	1 or more	Greenish with glaucous bloom to bluish-purple & glaucous, finally red brown

Sources: Little (1950), Korstian and Brush (1931), Ouden (1965), Rehder (1940), Sargent (1965).

Figure 3—*Chamaecyparis lawsoniana*, Port-Orford cedar: longitudinal section through a seed.



8- to 10-year-old plantations of Atlantic white-cedar produced good seedcrops that were easy to collect (Bonner and Summerville 1999). When collecting cones of Alaska-cedar in the northern part of the range, precautions are needed to limit the collection to mature, second-year cones. The smaller, greenish-blue, immature, first-year cones are often present on the same branches with the yellow-brown mature cones (Harris 1974).

Extraction, cleaning, and storage of seeds. Cones of white-cedars may be dried by spreading them in the sun or in a warm room, or they may be kiln-dried at temperatures below 43 °C (Harris 1974). Over 90% of the seeds can be recovered from cones of Atlantic white-cedar dried at 35 to 40 °C if 2 or 3 cycles of drying, interspersed with re-wetting of the cones, are used (Bonner and Summerville 1999). Each time the cones are redried, they open a little more. Mature cones of all white-cedars open when dried properly, and their seeds may be extracted by gentle shaking or tumbling. The thin-coated seeds of all species are easily injured and de-winging should not be attempted (Harris 1974).

Cleaning seeds of white-cedars to high purity values is difficult because the small, scalelike leaves are similar to the seeds in size and weight. For seedlots of Atlantic white-cedar, large trash can be removed with round-hole screens, and small trash can be blown off with any number of pneumatic cleaners or seed blowers. These same blowers can be used to upgrade Atlantic white-cedar seedlots by removing many of the empty seeds that occur naturally in this species. Separation is not absolute, of course, and many smaller filled seeds will be lost. With care, however, purities above 90% and filled seed percentages close to 90% can be obtained (Bonner and Summerville 1999). Similar data on the other 2 species are not available. Numbers of cleaned seeds per weight are listed in table 4.

Seeds of the white-cedars are orthodox in storage behavior. They should be stored at or below freezing at a seed moisture content of 10% or below (Allen 1957; Harris 1974). Seeds of Port-Orford-cedar from several origins stored at –15 °C lost no germination capacity over an 11-year period (Zobel 1990b). There are no comparable storage data for Alaska-cedar or Atlantic white-cedar, but the latter species is known to survive at least 2 years of similar storage without loss of viability (Bonner and Summerville 1999). Atlantic white-cedar seeds will also survive for at least 2 growing seasons in natural seedbeds (Little 1950).

Pregermination treatments and germination tests. Germination of white-cedar species is reported to be extremely variable and usually low, but this is due primarily to the naturally low percentages of filled seeds and the failure of seed managers to remove these empty seeds from the seedlots. Port-Orford-cedar germinates readily in the laboratory without pretreatment, and cold stratification does not appear to even improve germination rate (Zobel 1990b). Alaska-cedar exhibits a dormancy that can be somewhat overcome by warm incubation followed by cold stratification, but optimum schedules have not been determined (Harris 1990). In laboratory testing of germination, stratifi-

Table 4—*Chamaecyparis,* white-cedar: seed yield data

Species	Seed wt/ cone wt	Cleaned seeds/weight Range /kg	Range /lb	Average /kg	Average /lb
C. lawsoniana	20	176,400–1,323,000	80,000–600,000	463,000	210,000
C. nootkatensis	—	145,500–396,900	66,000–180,000	238,140	108,000
*C. thyoides**	20	926,100–1,102,500	420,000–500,000	1,014,300	460,000

Sources: Harris (1974), Korstian and Brush (1931), Swingle (1939).

* 1.64 kg (3.46 lb) of seeds were obtained from 1 bushel of cones (Van Dersal 1938).

cation of 21 days at 3 to 5 °C has been recommended (ISTA 1993). In nursery sowing, however, environmental conditions are seldom as favorable as those in the laboratory, so longer pretreatments are usually beneficial. One promising pretreatment schedule is moist stratification for 30 days at alternating temperatures of 20 and 30 °C, followed by 30 days at 5 °C (Harris 1974). The beneficial effect of warm incubation suggests that many of the seeds are not quite fully matured, and the incubation period enhances maturation in the same manner as warmer temperatures were shown to speed up cone ripening (Owens and Molder 1975).

Atlantic white-cedar has a variable dormancy also, although probably not as deep as that of Alaska-cedar. Some lots will germinate completely in the laboratory without any pretreatment (table 5). Recent tests with samples from North Carolina indicate that maximum germination in 28 days at good rates requires 4 weeks of moist stratification at 3 °C. Official test prescriptions (ISTA 1993) call for 90 days of stratification at 3 °C for germination within the same time period. Extremely slow germination has been reported in nursery beds in New Jersey (Little 1950), so some stratification would certainly be recommended. Germination is epigeal.

Nursery practice. For all 3 species of North American white-cedars, spring-sowing of stratified seeds is recommended. Port-Orford-cedar has the least dormancy and may only require 30 days of cold stratification. In England this species is normally not stratified at all (Aldous 1972). Alaska-cedar and Atlantic white-cedar have deeper levels of dormancy, and more extended pretreatments are necessary. Warm incubation at alternating temperatures, followed by cold stratification (as described in the previous section), has been recommended for both of these species (Dirr and Heuser 1987). Seeds from the more southern sources of Atlantic white-cedar seem to be not so dormant, and 30 to 60 days of cold stratification alone may be sufficient. Experience with Port-Orford-cedar in western nurseries suggests covering the sown seeds with 3 to 6 mm ($^{1}/_{10}$ to $^{1}/_{4}$ in) of soil and calculating sowing rates to produce 320 to 530 seedlings/m^2 (30 to 50/ft^2). One kilogram (2.2 lb) of Port-Orford-cedar seeds should produce about 284,000 plantable seedlings (Harris 1974). Shading the seedbeds until midseason of the first year may also be beneficial. For field planting, 2+0 stock is commonly used in the western United States, although 2+1 transplants are favored for Port-Orford-cedar in England (Harris 1974). For Atlantic white-cedar, 2+0 seedlings are used in New Jersey and 1+0 seedlings in North Carlina (Kuser and Zimmerman 1995).

All white-cedars can be propagated vegetatively and are commonly produced this way for the ornamental market. Port-Orford-cedar cuttings should be taken between September and April, treated with indole butyric acid (IBA) powder (3,000 to 8,000 ppm), and placed in peat or perlite with mist and bottom heat (Dirr and Heuser 1987). Zobel (1990a) suggests taking cuttings from tips of major branches from lower branches of young trees. Alaska-cedar cuttings need 8,000 or more ppm of IBA, and cuttings should be taken in late winter to early spring (Dirr and Heuser 1987). After 4 years, growth of outplanted rooted cuttings was equal to that of seedlings in British Columbia (Karlsson 1982). Atlantic white-cedar cuttings taken in mid-November and treated with auxins also root very well (Dirr and Heuser 1987). With auxins and bottom heat in mist-beds, 90% rooting can be expected. Early comparisons show that growth of seedlings and stecklings (rooted cuttings) to be about the same (Kuser and Zimmerman 1995).

Table 5—*Chamaecyparis,* white-cedar: stratification periods and germination test conditions and results

	Test conditions					Test results				
	Stratification (days)		Temp (°C)			Germ energy		Germ capacity	Soundness	
Species	Warm*	Cold†	Day	Night	Days	(%)	Days	(%)	(%)	Samples
C. lawsoniana	0	0	30	20	28	44	14	48	48	9
	0	0	30	20	60	24	34	52	—	60
C. nootkatensis	58	30	30	20	22	10	11	12	51	1
	0	30–90	30	20	41	0	—	0	57	3
	0	0	30	20	28–55	0	—	0	54	8
C. thyoides	0	0	30	20	60	—	—	84	—	11
	0	90	30	20	28	—	—	—	—	—

Source: Harris (1974).

* At alternating temperatures of 30 and 20 °C.

† At 5 °C.

‡ Seeds were exposed to light during the warm period.

§ A constant temperature of 20 °C is also suitable (ISTA 1993).

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Bignoniaceae—Trumpet-creeper family

Chilopsis linearis (Cav.) Sweet

desert-willow

Arthur W. Magill, Carol Miller, and Jane E. Rodgers

Dr. Magill retired from USDA Forest Service's Pacific Southwest Forest and Range Experimental Station; Ms. Miller was a propagationist at the USDI National Park Service Joshua Tree National Park Center for Arid Lands Restoration and Native Plant Nursery, Twentynine Palms, California; Ms. Rodgers, is a restoration ecologist at the Point Reyes National Seashore Point Reyes Station, California

Synonyms. *C. saligna* D. Don, C. *linearis* var. *originaria* Fosberg, *C. linearis* var. *glutinosa* (Engelm.) Fosberg, C. *linearis* var. *arcuata* Fosberg.

Other common names. false-willow, *jano,* flowering-willow, desert catalpa, catalpa-willow.

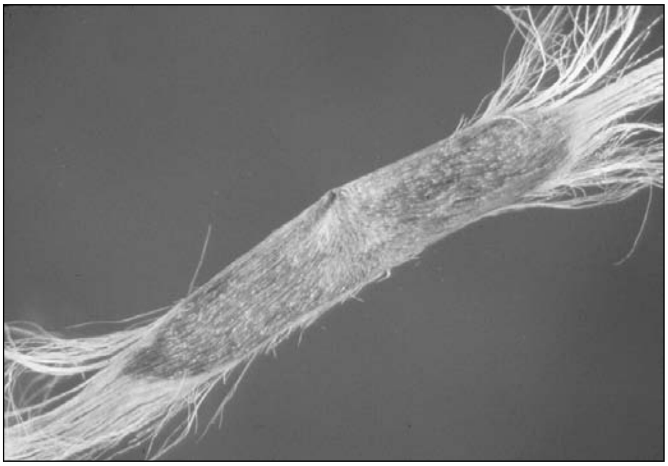
Growth habit, occurrence, and use. Desert-willow grows along dry washes and streams in the desert between 450 and 1,500 m of elevation from southern California through southern Utah to western Texas and southward into Mexico and Lower California. It is a deciduous shrub or small tree that attains heights of 3 to 7.5 m or occasionally more. Growth can be rapid, up to 1 m annually (Munz 1979). The plant is useful for wildlife cover, erosion control, restoration, stream stabilization, and ornamental plantings in arid regions (McMinn 1959; Bainbridge and Virginia 1989; Munz 1959). Seed pods and flowers are edible, but the major use for Native American people was the wood (for house frames, granaries, and bows) and the fibrous bark (for weaving nets, shirts, and breechclouts) (Bainbridge and Virginia 1989).

Flowering and fruiting. Desert-willow produces perfect flowers between April and August throughout its range (Magill 1974; McMinn 1959). The fruit is a 2-celled capsule about 6 mm in diameter and from 10 to 30 cm long. It ripens from late summer to late fall (Afanasiev 1942) and persists through winter (Little 1950). The numerous light-brown oval seeds are about 8 mm long and have a fringe of soft white hairs on each end (figures 1 and 2).

Collection, extraction, and storage. Seedpods can be hand-picked after late September and through the winter months. Care must be taken not to pick unripened fruits—the fruits on a tree may mature unevenly because of their long flowering period (Engstrom and Stoeckeler 1941). Seed extraction simply requires that the pods be spread out, dried, beaten lightly, and shaken, and then the seeds screened out. Each 45 kg (100 lb) of dried pods should produce 14 to 23 kg (30 to 50 lb) of clean seeds, which number from 88,200 to 282,240/kg (40,000 to 128,000/lb) and average 189,130/kg (86,000/lb) (Magill 1974). Commercial seedlots have averaged 92% in purity and 87% in soundness (Magill 1974). These seeds are orthodox in storage behavior, so cold, dry storage conditions are recommended for storage. Seeds have been successfully propagated after 4 years of refrigerated storage at 7 °C (CALR 1993).

Germination. Desert-willow seeds are not dormant, but storage for several days in wet sand or between wet blotter paper will speed germination. In germination tests 1,000 seeds were placed in a sand or water medium for 21 to 60 days with a night temperature of 20 °C and a day temperature of 30 °C (Engstrom and Stoeckeler 1941; Magill 1974). Germination averaged 14 to 60% in 9 to 30 days and germinative capacity ranged from 26 to 100% (Magill 1974). Average germination using blotter paper is 80% (CALR 1993).

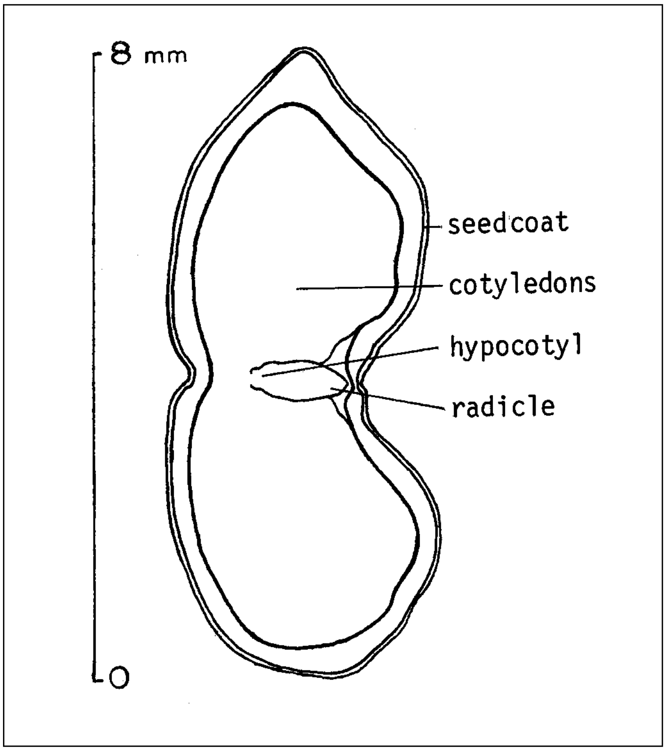
Figure 1—*Chilopsis linearis*, desert-willow: seed.



Nursery field practice and seedling care. Desert-willow seeds may decay unless sown in spring soon after the soil warms up. Sowing depth should be 6 mm ($^1/_4$ in). A ratio of seven times as many viable seeds as the desired number of usable seedlings is required to grow nursery stock (Magill 1974). Damping-off can be a problem.

Desert-willow may also be propagated from cuttings (Magill 1974); cuttings should be handled carefully and allowed to produce an extensive rootball before transplanting. Mature plants grown in ~57-liter (15-gal) pots and 0.8-m (30-in) tubes have been successfully outplanted as windbreaks and for desert restoration at Joshua Tree National Park (CALR 1993).

Figure 2—*Chilopsis linearis*, desert-willow: longitudinal section through a seed.



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Pyrolaceae—Shinleaf family

Chimaphila Pursh

chimaphila

Don Minore

Dr. Minore retired from the USDA Forest Service's Pacific Northwest Research Station

Growth habit, occurrence, and use. Taxonomists sometimes differ when classifying plants in the genus *Chimaphila* (Blake 1914; Camp 1939; Hitchcock and others 1959; Stapf 1930; Takahashi 1987; Traulau 1981; Wordsdell and Hill 1941), but there are at least 4 clearly defined species (table 1). All have a chromosome number (2n) of 28 (Haber and Cruise 1974), and all occur in the Northern Hemisphere (table 1). Pipsissewa, the most widespread species, has been divided into 5 geographically delimited subspecies: *C. umbellata* ssp. *occidentalis* (Rydb.) Hult. in western North America; ssp. *acuta* (Rydb.) Hult. in Arizona and New Mexico; ssp. *mexicana* (DC) Hult. in Mexico; ssp. *cisatlantica* (Blake) Hult. in eastern North America; and ssp. *umbellata* in Europe and Asia (Takahashi 1987).

The chimaphilas are low, usually creeping, evergreen subshrubs (Krüssmann 1984) with alternate leaves that are crowded near the summit of each year's annual growth, giving the appearance of being whorled on the short shoots ("pseudo-whorls"). Those shoots produce annual growth rings (Copeland 1947) and are connected by elongate rhizomes that are as much as 2.5 m long. The rhizomes are slender (not more than a few millimeters in diameter) and yellow or brown. They bear distant buds that are subtended by small scales and associated with single roots. The leaves may persist as long as 7 years in pipsissewa and 8 years in little pipsissewa (Copeland 1947).

Chimaphila leaves are purported to have antibacterial properties. They contain taraxerol, beta-sitosterol, ursolic acid, nonacosane, hentriacontane, isohomoarbutin, renifolin, arbutin, avicularin, hyperoside, several flavonoids, and a compound called chimaphilin (Lucia 1991; Sheth and others 1967; Trubachev and Batyuk 1968; Walewska 1971). Chimaphilin has a quinone structure similar to that of 1,4-naphthoquinone (DiModica and others 1953), and it may be responsible for the medicinal properties attributed to the chimaphilas. The boiled leaves are taken as a liver remedy (Altschul 1973). The plants also have been used as diuretics and to treat rheumatism and fever (Krüssmann 1984). Large quantities of pipsissewa are now being harvested for use as flavoring in a popular beverage.

Flowering and fruiting. Striped pipsissewa usually flowers in its third growing season, but flowering may be delayed in pipsissewa and little pipsissewa until 7 or 8 annual pseudo-whorls of leaves have been produced (Copeland 1947). Flowers are choripetalous, pentacyclic, pentamerous, actinomorphic, and protogynous (Holm 1927; Pyykko

Table 1—*Chimaphila*: nomenclature and occurrence

Scientific name	Common name(s)	Distribution
***C. japonica* Miq.**	**Japanese chimaphila**	Japan, Korea, China, & Taiwan
***C. maculata* (L.) Pursh**	**striped pipsissewa,** striped prince's-pine, spotted wintergreen	Maine & New Hampshire to Ontario, Michigan, & Illinois S to Georgia, Alabama, & Tennessee
***C. menziesii* (R. Br. ex D. Don) Spreng.**	**little pipsissewa,** little prince's-pine	British Columbia, Montana, & Washington S through Oregon & California to Mexico
***C. umbellata* (L.) W. Bart.**	**pipsissewa,** prince's-pine	North America from Alaska to Mexico & from Ontario & New Brunswick to Georgia; Europe (incl. Scandinavia), Eurasia, Japan, & West Indies

Sources: Barrett and Helenurm (1987), Blake (1914), Camp (1939), Fernald (1950), Hill (1962), Nordal and Wischmann (1989), Ohwi (1965), Prain (1960), Traulau (1981).

1968). They have 5-parted calyxes, 5 petals, 10 stamens, 5-chambered ovaries, and short thick styles with wide, 5-pointed stigmas (Krüssmann 1984). The ovary is superior, and there is a well-developed, collar-like disk at the base of the pistil that secretes nectar. Placentation is central-axile, with a massive, 2-lobed placenta intruding into each locule (Pyykko 1968). Those placentae are beset with numerous minute ovules (Copeland 1947). The 1 to 3 (little pipsissewa and Japanese chimaphila) or 2 to 6 (striped pipsissewa and pipsissewa) flowers are borne in pendulous, terminal inflorescences (Krüssmann 1984; Ohwi 1965). In pipsissewa those inflorescences are corymbs; in striped and little pipsissewas, they are cyme-like clusters (Copeland 1947). Flowers are pink or white and slightly fragrant.

Chimaphila pollen grains are packed into polyads composed of indefinite numbers of tetrads (Knudsen and Olesen 1993; Takahashi 1986). Pollination is by insects. In pipsissewa, bumble bees are the most important pollinators but the flowers also are visited by staphylinid beetles (Barrett and Helenurm 1987; Knudsen and Olesen 1993), and there is a small amount of self-pollination (Barrett and Helenurm 1987). Differences in the flower preferences of the bumble bee species involved may help to prevent interbreeding between pipsissewa and striped pipsissewa during a short overlap in flowering periods where these inter-fertile species grow together (Standley and others 1988).

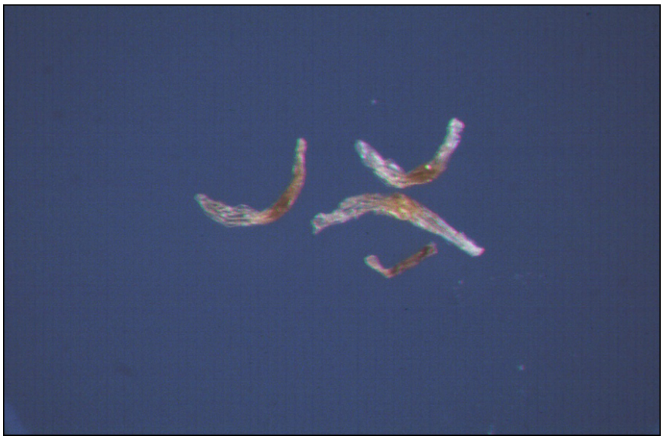
An average pipsissewa flower produces 308,800 pollen grains and 5,587 ovules—a pollen to ovule ratio of 58 (Barrett and Helenurm 1987). In central New Brunswick, Canada, anthesis occurs over a 30-day period in July (Helenurm and Barrett 1987). In the Pacific Northwest, pipsissewa flowers may be found from June until August (Hitchcock and others 1959). The fruits matured in about 70 days in New Brunswick, where an average fruit weighed 23 mg, and fruit set was 75% for both self-pollinated and cross-pollinated flowers (Barrett and Helenurm 1987).

The chimaphila fruit is a 5-celled, loculicidally dehiscent capsule that contains very large numbers of tiny seeds (Barrett and Helenurm 1987; Copeland 1947; Pyykko 1968) that sift out of the capsule openings to be borne away by the wind. The embryos of those seeds develop no distinct parts during seed development, but they eventually absorb all of the endosperm except a single layer of cells. The inner integumental cells die and remain in existence as more or less shriveled empty spaces at the ends of the seeds (Copeland 1947). The seedcoat consists only of the outer cell layer of the integument, which loses protoplasm and tannin to become transparent (Pyykko 1968). Although the inner periclinal walls of those transparent testa cells are smooth or slightly pitted in all species, intraspecific differences occur in pipsissewa (Takahashi 1993).

Chimaphila seeds are characterized by very small size, few cells, and little differentiation (figure 1). Each contains a central ellipsoidal mass consisting of an embryo covered by a single layer of endosperm cells, surrounded by a transparent seedcoat that is hollow and shriveled at each end. Ripe seeds are 0.6 to 0.9 mm long and 0.1 mm wide. There are about 1,500,000 seeds/g (42,524,250/oz) (Minore 1994).

Collection and storage of seeds. Seeds can be collected in the field by tapping recently dehisced capsules to dislodge the tiny seeds, which can then be captured in a jar or plastic bag as they drift downward. Recently dehisced capsules may be difficult to find, however, because mature capsules often are not open or have already lost their seeds. Mature closed capsules can be collected, dried, and macerated to recover the seeds. Unfortunately, this latter procedure creates debris that is difficult to separate from the seeds, and seed maturity is not assured. Optimal storage conditions and seed longevity are unknown.

Figure 1—*Chimaphila umbellata*, pipsissewa: seeds (0.1 mm wide and ~ 0.7 mm long, at center) with central embryo and elongate, transparent seedcoat.



Pregermination treatments; germination tests; and nursery practice. Chimaphila seeds have not been sown and germinated successfully. Forest soil that had been sifted to remove debris and rhizome material and then stored outdoors all winter produced pipsissewa seedlings in the spring, however, indicating that there are viable seeds in the soil seed bank and that extensive stratification may be necessary (Wilson 1994). No formal pregermination treatments or germination tests are known. Chimaphila seedlings are seldom found in nature (Holm 1927). Therefore, most natural regeneration may be accomplished by the spread of rhizomes. Cultivation attempts often fail (Kruckeberg 1982), but division of the rhizome has been recommended as a suitable method of propagation (Bailey and Bailey 1976). The chimaphilas may be partial root parasites (Kruckeberg 1982). If they are, special practices that include an unknown host may be needed to achieve successful large-scale nursery production.

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Oleaceae—Olive family

Chionanthus virginicus L.

white fringetree

John D. Gill, Franz L. Pogge, and Franklin T. Bonner

Dr. Gill and Mr. Pogge retired from the USDA Forest Service's Northeastern Forest Experiment Station; Dr. Bonner is a scientist emeritus at the USDA Forest Service's Southern Forest Experiment Station, Mississippi State, Mississippi

Other common names. old-man's-beard, flowering-ash, grandfather-graybeard.

Growth habit, occurrence, and uses. White fringetree—*Chionanthus virginicus* L.—occurs on rich, well-drained soils of streambanks, coves, and lower slopes but is most abundant in the understory of pine–hardwood forests, especially on moist, acid, sandy loam soils (Goodrum and Halls 1961). It develops best in semi-open situations but is moderately shade-tolerant, being found occasionally in dense understories. Though widely distributed, it usually is a minor part of the total vegetation. White fringetree is a relatively short-lived shrub or small tree and may attain 11 m in height (Rehder 1940). Its range is from southern Pennsylvania and Ohio south to central Florida and westward through the Gulf Coast region to the Brazos River in Texas and to northern Arkansas (Brown and Kirkman 1990).

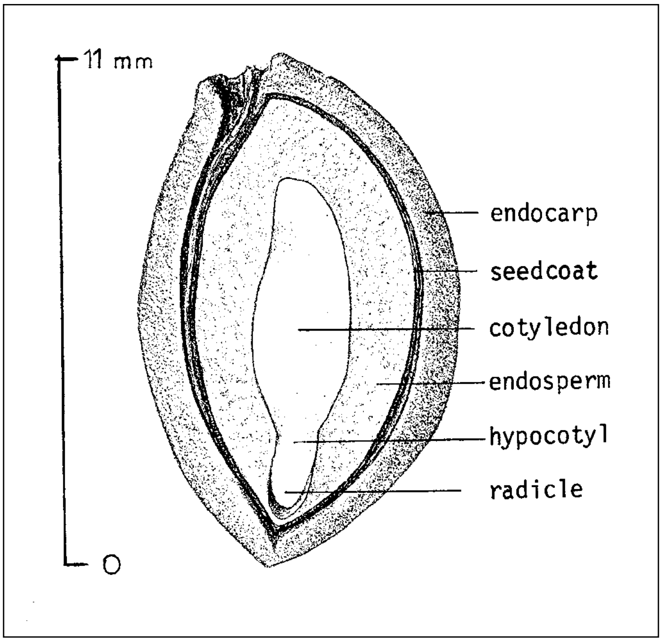
Fringetrees are planted as ornamentals throughout the South and elsewhere beyond their natural range. The bark is used as a tonic, diuretic, and astringent; it is also used to reduce fever. In Appalachia, a liquid of boiled root bark is applied to skin irritations (Krochmal and others 1969). Twigs and foliage are preferred browse for deer (*Odocoileus* spp.) in the Gulf Coastal Plain but are less preferred in the Piedmont and mountains. Browsing is least in winter. The species is only moderately resistant to browsing, and plants may die when more than a third of the annual growth is removed. The date-like fruits are eaten by many animals, including deer, turkey (*Meleagris gallopovo*), and quail (*Callipepla* spp.). Cattle may eat the foliage (Goodrum and Halls 1961). The date of earliest known cultivation is 1736 (Rehder 1940).

Flowering and fruiting. White fringetree's flowering habit is polygamo-dioecious, although it is functionally dioecious (Brown and Kirkman 1990; Gleason 1963). The white, fragrant flowers are borne in pendant axillary panicles 10 to 25 cm long that appear from March to June, depending on latitude (Brown and Kirkman 1990; Gill and Pogge 1974). The fruit is a dark blue to purple ovoid drupe about 2 cm long (figure 1). It is usually single-seeded (figures 1 and 2); rarely 2- or 3-seeded. Fruits ripen and drop from the trees in July in eastern Texas and as late as October in the northern part of the range (Lay 1961; Van Dersal 1938). Seeds are dispersed beyond the immediate vicinity of the tree by birds and rodents. Plants first produce seeds at 5 to 8 years of age. In eastern Texas, they produced some fruit each year; no seedcrop failure occurred (Lay 1961).

Collection, extraction, and storage. The fruits should be collected from the branches after they have turned purple and before birds remove them, which should be August in the South and September and October in the North (Dirr and Heuser 1987). The pulpy pericarp should be removed by maceration with either mechanical macerators for large lots or kitchen blenders for small lots, or by rubbing the fruits over hardware cloth fine enough to retain the seeds. The pulp may then be washed away. The seeds contain about 40% moisture when they are shed and must be dried to at least 22% if they are to be stored at low temperatures (Carpenter and others 1992). There have been no long-term storage tests of white fringetree seeds reported,

Figure 1—*Chionanthus virginicus*, white fringetree: fruit (drupe, **left**) and seed (**right**).

Figure 2—*Chionanthus virginicus*, white fringetree: longitudinal section through a seed.

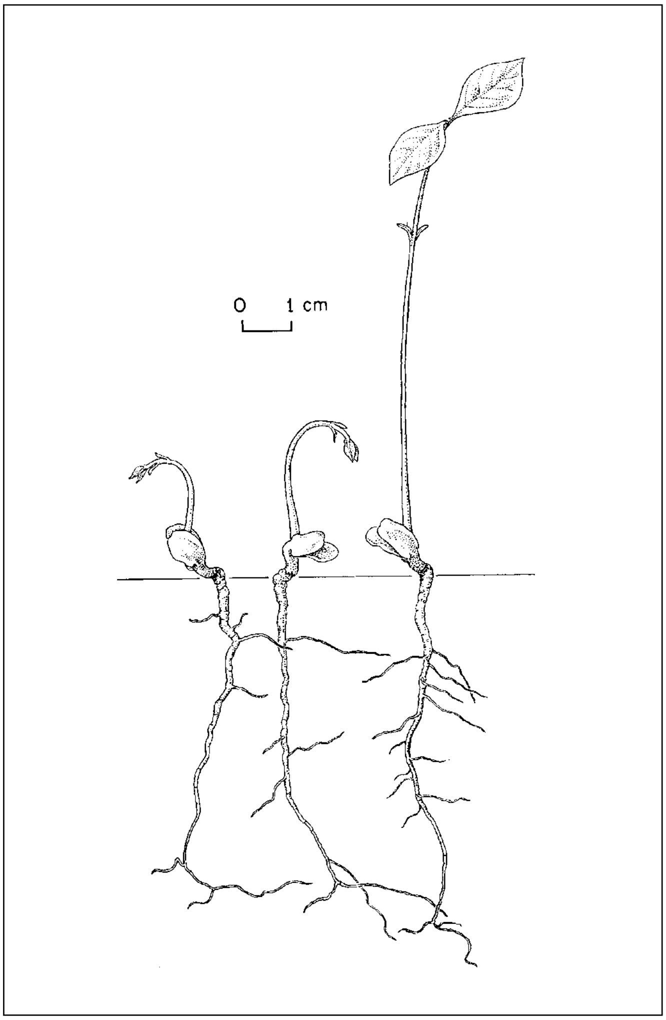


although seeds have remained viable in cold stratification for 1 to 2 years (Gill and Pogge 1974). It is not known if these seeds are orthodox or recalcitrant. There are about 1,400 fruits/kg (630/lb); 1 kg of fruits yielded 330 g of seeds and 1 lb yields 5 1/4 oz. The average number of seeds per weight is 4,000/kg (1,800/lb) with a range of 2,420/kg to 4,410 (1,100 to 2,000/lb) (Gill and Pogge 1974; Swingle 1939; VanDersal 1938).

Germination. Natural germination usually occurs in the second spring after seedfall, the results of an apparent double dormancy or combined dormancy in the seeds. Fringetree seeds first need a period of warm temperatures, commonly 3 to 5 months, during which the radicle develops while the shoot remains dormant. Subsequently, cold exposure during winter overcomes the shoot dormancy (Flemion 1941; Schumacher 1962). In the wild, these temperature exposures occur during the first summer and second winter after seedfall. In a test with 2 seedlots, seeds were held at 20 °C for 1 or more months; stratified at 5 °C for 1 month or more; then sown in flats and held at 20 to 30 °C for 1 year. Germination was about 40% (Gill and Pogge 1974). Good germination (80%) can also be obtained with removal of the hard endocarp; soaking in 1,000 ppm solution of gibberellin (GA$_3$) for 6 hours; then germinating at 25 °C (Carpenter and others 1992). No official test methods have been prescribed, and the embryo excision method has been recommended for quick viability estimates (Barton 1961; Flemion 1941; Heit 1955). Germination is hypogeal (figure 3).

Nursery practice. Seeds may be sown in either fall or spring. Seeds can be sown soon after they are cleaned, but no later than mid-October in the northern part of the range (Heit 1967). Drills should be set 20 to 30 cm (8 to 12 in) apart and the seeds covered with 6 to 12 mm ($^{1}/_{4}$ to $^{1}/_{2}$ in) of firmed soil. Beds should be covered with burlap or mulched with straw or leaf mulch until after the last frost the following spring. If spring-sowing is desired, then seeds should be given 3 months of warm storage, then 3 months of cold stratification (Dirr and Heuser 1987). As an alternative for the amateur gardener, seeds can be sown under glass, in boxes with standard compost, during February–March (Sheat 1948). Propagation by layering, grafting, or budding onto ash seedlings is sometimes practiced, but the species is almost impossible to root (Dirr and Heuser 1987).

Figure 3—*Chionanthus virginicus*, white fringetree: seedling development at 1, 4, and 7 days after germination.



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Fagaceae—Beech family

Chrysolepis Hjelmqvist

chinquapin

Philip M. McDonald

Dr. McDonald is a research silviculturist at the USDA Forest Service's Pacific Southwest Research Station, Redding, California

Synonyms. The 2 species of *Chrysolepis* found in the United States are distinct from their Asian relatives in the genus *Castanopsis* and were placed by Hjelmqvist (1948) in their current genus. This genus was accepted in Hickman's extensive and long-researched flora of California (1993). These American species, which have a floral morphology that is intermediate between *Castanopsis* and *Lithocarpus*, represent the ancient condition of the family Fagaceae (McKee 1990). The common name also has changed throughout the years. Early workers called the species "chinquapin." Later, it became "chinkapin" but more recently it was changed back to "chinquapin" (Hickman 1993; Keeler-Wolf 1988).

Occurrence and growth habit. In North America, the genus *Chrysolepis* consists of 2 species and 1 variety (Hickman 1993), all located in the Pacific Coast region. Giant chinquapin—*Chrysolepis chrysophylla* var. *chrysophylla* (Dougl. ex. Hook) Hjelmqvist —is a tree that ranges from southwestern Washington southward to San Luis Obispo County in the Cascade, Klamath, and Coastal Mountains of California. A remnant stand also exists in El Dorado County in the north central Sierra Nevada. This species achieves its best form from Marin County, California, northward (Griffin and Critchfield 1972) to Lane County, Oregon. Giant chinquapin also has a shrub form—*C. chrysophylla* var. *minor* (Benth.) Munz, often called "golden chinquapin"—that is found throughout the range of its taller brethren.

The second species—*C. sempervirens* (Kellogg) Hjelmqvist—which is always a shrub, has the common name "bush chinquapin." This species is found from the Cascade Mountains of southern Oregon westward in the Siskiyou Mountains of northern California, and southward along the east-facing slopes of the north Coast Range and the west-facing slopes of the Sierra Nevada, San Jacinto, and San Bernardino Mountains (McMinn 1939). Throughout its range, the habitat is characterized as being of low quality with shallow, rocky, and often droughty soils. In western Siskiyou County, California, and in other places where the ranges of the 2 shrub forms overlap, hybridization probably occurs (Griffin and Critchfield 1972).

Giant chinquapin is often found as a single tree or in groves; it rarely occupies extensive areas. This shade-tolerant tree is rarely found in a dominant position; it is more often found in intermediate and codominant crown positions. Pure stands are uncommon and rarely exceed 10 ha (McKee 1990). In the Klamath Mountains of northern California, giant chinquapin shows a distinct preference for mesic conditions, with highest basal areas occurring on north-facing slopes or in mesic canyon bottoms (Keeler-Wolf 1988). In general, best growth is achieved in moist environments with deep and infertile soils (Zobel and others 1976). The shrub forms occupy a plethora of topographic/edaphic sites over an elevational range that varies from 300 to 3,000 m. The shrub forms can be quite extensive and achieve greatest coverage in the extreme environments of xeric sites at higher elevations. Here they dominate, with their area corresponding to the extent of past disturbance. The amount of time that they dominate also can be lengthy, given a lack of seed source for inherently taller competitors. Over a long time span, however, disturbance is necessary for the continued presence of chinquapin. Because of its wide ecological amplitude, chinquapin is part of many associations that include most of the forest-zone conifers and hardwoods on the Pacific Coast. A general pattern for all the species and varieties is that they are at their competitive best on infertile soils (McKee 1990).

Chinquapins are vigorous sprouters and most trees originate as root crown sprouts. The sprouts grow rapidly and outstrip natural conifer seedlings for several years. Mature trees tend to have straight boles and narrow crowns. The largest trees may reach over 33 m in height and 1 to 1.2 m in girth (Sudworth 1908). For shrubs, var. *minor* tends to be stiff and upright in exposed areas and semiprostrate in shaded environments. Bush chinquapin is stiff and upright in all environments.

Use. The light, fairly hard, and strong wood of chinquapin has been used for veneer, paneling, cabinets, furniture, turned products, pallets, and fuel (EDA 1968).

Flowering and fruiting. The flowers of giant chinquapin, which bloom from June through midwinter, and the flowers of the shrubs, which bloom throughout the summer, are unisexual, with staminate and pistillate flowers being borne on the same plant. The staminate flowers are borne in groups of 3 in the axils of bracts, forming densely flowered, erect cylindrical catkins 2.5 to 7.6 cm long; 1 to 3 pistillate flowers are borne in an involucre, usually at the base of the staminate catkins or borne in short separate catkins. At the time of peak blooming in June, each tree is covered with erect creamy white blossoms, which provide a pleasing contrast to the more somber foliage (Peattie 1953).

The fruit consists of 1 to 3 nuts (figures 1 and 2) enclosed in a spiny golden brown bur. The nuts mature in fall of the second year (Hickman 1993). The minimum seed-bearing age (from root crown sprouts) is 6 years (McKee 1990). Giant chinquapin trees have been reported as producing seeds at 40 to 50 years of age but probably do so before this age (McKee 1990). Controversy exists over seed productivity. Sudworth (1908) reported that the tree form is an abundant seeder, but Peattie (1953) noted that although flowering is abundant, fruiting is "strangely shy." Insects, squirrels, and birds often consume most of a given crop. Indeed, Powell (1994) observed tree squirrels (*Sciurus* spp.) cutting burs of large chinquapins during a bumper seed year. By late fall, the ground beneath the trees was covered with burs.

Collection, extraction, and storage. Because of heavy predation by many animals, collectors should hand-pick the burs in late summer or early fall, after they ripen but before they open (Hubbard 1974). The collected burs should be spread out to dry in the sun or in a warm room. After drying, the nuts can be separated from the burs mechanically. The following number of nuts per weight have been recorded (Hubbard 1974; McMinn 1939):

	Nuts/kg	Nuts/lb
giant chinquapin (*C. chrysophylla* var. *chrysophylla*)	1,826–2,420	830–1,100
golden chinquapin (*C. chrysophylla* var. *minor*)	1,540	700
bush chinquapin (*C. sempervirens*)	—	2,640 1,200

Figure 1—*Chrysolepsis chrysophila* var. *chrysophila*, giant chinquapin: nut.

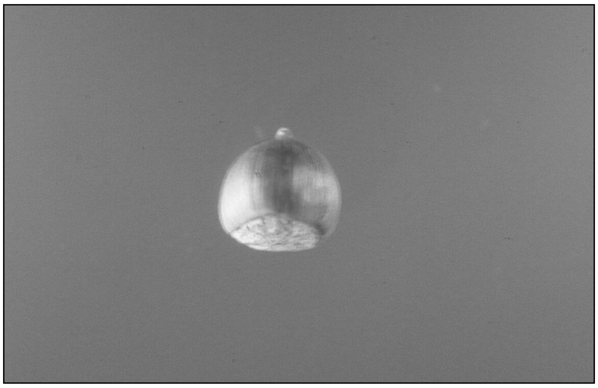
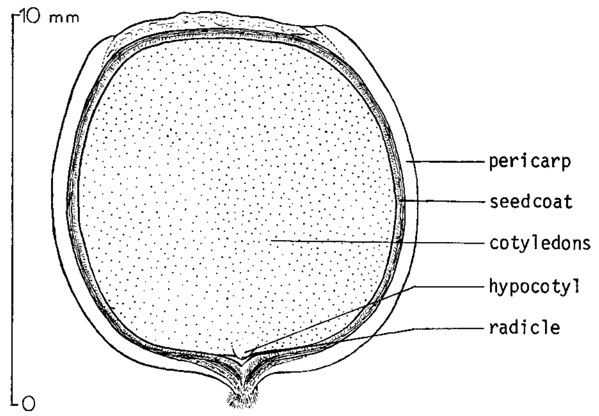


Figure 2—*Chrysolepsis*, chinquapin: longitudinal section through a seed.

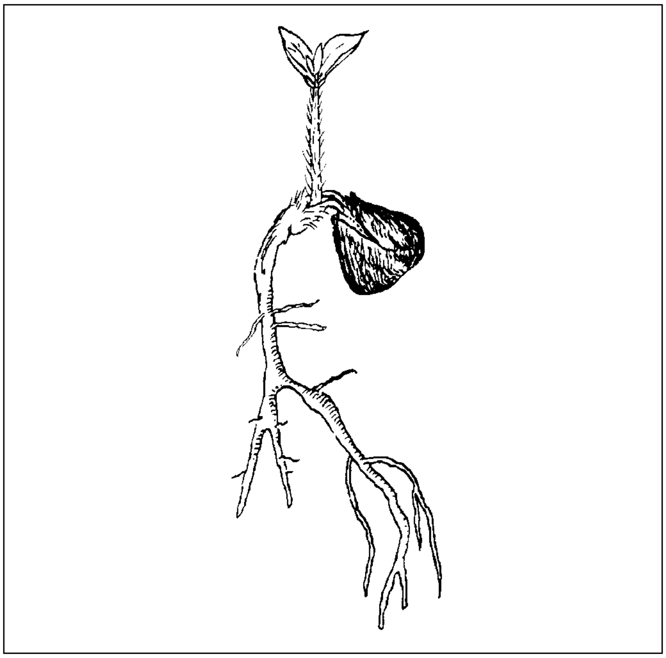


When stored in sealed containers at 6 °C, chinquapin seeds retain their viability well for at least 2 years and probably longer. Viability of 1 sample of giant chinquapin seeds stored in this manner dropped only from 50 to 44% in 5 years (Hubbard 1974).

Pregermination treatments. Mirov and Kraebel (1937) found that no stratification was needed.

Germination. Germination of untreated seeds of giant chinquapin in 3 tests ranged from 14 to 53% (Hubbard 1974)—the poorest of all hardwoods in the Klamath Mountains provenance of southwestern Oregon and northern California (McDonald and others 1983). Mirov and Kraebel (1937) found highest germination values for giant chinquapin to be 50% in 24 days and for bush chinquapin was 30% in 16 days. Germination is hypogeal (figure 3) and best in peat.

Figure 3—*Chrysolepsis,* chinquapin: seedling at 1 month after germination



Nursery practice. Little is known about raising chinquapins in nurseries. In a study at the Rancho Santa Ana Botanic Gardens in California, the 3 native species were raised in pots. Some survived through 1 or more potting stages, but none survived after outplantings (Hubbard 1974). Propagation by layering, grafting, or budding is feasible (Hubbard 1974).

Seedling care. Natural regeneration of giant chinquapin usually is sparse or totally lacking. Powell (1994) noted that not a single seedling was present on ground covered with burs beneath large seed trees. McKee (1990) also inferred that regeneration was lacking in environments of deep litter and dense understory vegetation. Sudworth (1908) noted that regeneration was best if seeds were covered, apparently by eroded soil. Keeler-Wolf (1988) found sexually reproduced seedlings and saplings to average about 19/ha (7/ac) in the Klamath Mountains but only in shaded mesic environments. In the Oregon Cascade Mountains, McKee (1990) noted that chinquapin reproduction occurred in light leaf mulch in partial shade, with plantlets that were 15 to 45 cm tall at ages 4 to 12. For bush chinquapin in the northern Sierra Nevada on 10 study areas over a 10-year period, not 1 seedling was found. Although tiny plants looked like seedlings, a gentle tug showed that they were connected to parent-plant root systems. The number of new sprouts averaged over 39,000/ha (16,000/ac) 6 years after site preparation by bulldozer bared the ground (McDonald and others 1994).

Altogether, this evidence suggests that for both natural and artificial regeneration, best seedling care will be achieved with covered seeds in partially shaded, moist conditions. Seedling growth in this environment, however, is unknown.

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Asteraceae–Aster family

Chrysothamnus Nutt.

rabbitbrush

Susan E. Meyer

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

Growth habit, occurrence, and use. Members of the rabbitbrush genus—*Chrysothamnus* spp.—are among the best-known of western shrubs (Johnson 1987; McArthur and Welch 1986; McArthur and others 2004, 1979). The genus is endemic to western North America and is made up of 16 species (Anderson 1986; McArthur and Meyer 1987). It has recently been partially subsumed under the new genus *Ericameria*, formerly an infraspecific taxon within the related genus *Haplopappus* (Anderson 1995; USDA NRCS 2001). Because the durability of this nomenclatural change has yet to be demonstrated, the decision here is to follow the traditional nomenclature (table 1).

Rabbitbrush commonly occurs on sites of natural or human disturbances such as washes, drainage-ways, and quarries; they may also occur as subdominants in later seral vegetation. Their conspicuous golden flowers are a familiar sight in autumn along roadsides throughout the West. Three of the species—rubber rabbitbrush, Parry rabbitbrush, and green rabbitbrush—are widespread, polymorphic taxa made up of multiple subspecies, whereas the remainder are taxonomically simpler and more restricted in distribution. Rubber rabbitbrush is made up of 22 subspecies, many of which are ecologically distinctive. Its more ecologically specialized subspecies are restricted to dunes, rock outcrops, shale badlands, alkaline bottomlands, or montane riparian communities. Most of the widely distributed subspecies are also broad in their ecological requirements but tend to be commonest on disturbed ground. Common garden studies have shown that marked ecotypic differentiation occurs within subspecies for such traits as growth form, growth rate, cold and drought hardiness, competitive ability, flowering time, achene weight, and germination patterns (McArthur and others 1979; Meyer 1997; Meyer and others 1989). Such ecotypic variation is to be expected in other widely distributed species as well. It is therefore important to consider ecotype as well as species and subspecies when selecting seed sources for artificial seeding projects.

Species, subspecies, and populations of rabbitbrush also vary widely in their palatability to livestock and wildlife. Certain unpalatable taxa such as threadleaf rubber rabbitbrush tend to increase on abused rangeland, and considerable energy has been invested in control methods (Whisenant 1987). A tendency to resprout after herbicide spraying, chopping, or burning, combined with an ability to reestablish from seeds, can make rabbitbrush difficult to eradicate (Young and Evans 1974).

Basin and mountain whitestem rubber rabbitbrush races are highly palatable as winter forage for deer (*Odocoileus* spp.), sheep, and cattle, and have been included in seeding mixes for the rehabilitation of big game winter range for over 30 years (Monsen and Stevens 1987). Other species and subspecies also provide winter forage for wildlife and livestock (McArthur and others 2004). Rabbitbrushes are extensively used for mined-land rehabilitation in the West (Romo and Eddleman 1988). Thousands of pounds of wildland-collected seed are bought and sold annually (McArthur and others 2004; Monsen and others 2004, 1987). Rabbitbrushes may be seeded as pioneer species on harsh mine disturbances for erosion control and site amelioration and often invade such sites on their own (Monsen and Meyer 1990). They can act as nurse plants to facilitate establishment of later seral species, as they generally offer little competition to perennial grasses or later seral shrubs (Frischknecht 1963).

Rabbitbrushes have potential uses in landscape plantings, especially with the recent emphasis on xeriscaping. Rubber rabbitbrush has also been examined as a commercial source of natural rubber and other plant secondary metabolites such as resins (Weber and others 1987).

Flowering and fruiting. The perfect yellow disk flowers of rabbitbrushes usually occur in groups of 5 in narrowly cylindrical heads subtended by elongate, often keeled bracts. The heads are numerous and are clustered in often flat-topped terminal or lateral inflorescences that can be

Table 1—*Chrysothamnus,* rabbitbrush: ecology and distribution of some common species and subspecies

Taxon & species	Common name(s)	Geographic distribution	Habitat
SECTION CHRYSOTHAMNUS			
***C. linifolius* Greene** *Ericameria linifolia* (Greene) L.C.Anders.	**spearleaf rabbitbrush,** alkali rabbitbrush	Colorado Plateau N to Montana	Deep alkaline soils; low to mid-elevation
***C. viscidiflorus* (Hook.) Nutt.** *E. viscidiflora* (Hook.) L.C.Anders.	**green rabbitbrush**	Intermountain	Wide amplitude
***C. v.* ssp. *lanceolatus* (Nutt.) Hall & Clements** *E. viscidiflora* spp. *lanceolata* (Nutt.) L.C.Anders.	**Douglas rabbitbrush**	Intermountain	Montane
***C. v.* ssp. *viscidiflorus* (Hook.) Nutt.** *E. viscidiflora* (Hook.) L.C.Anders.	**low rabbitbrush**	Intermountain	Low to mid-elevation
SECTION NAUSEOSI*			
***C. nauseosus* (Pallas ex Pursh) Britt.** *E. nauseosa* (Pallas ex Pursh) Nesom & Baird	**rubber rabbitbrush**	W North America	Wide amplitude
***C. n.* ssp. *albicaulis* (Nutt.) Hall & Clements**	**mountain whitestem rubber rabbitbrush**	Mostly Intermountain Rocky Mtn	Mostly coarse soils; mid-elevation
***C. n.* ssp. *hololeucus* (Gray) Hall & Clements**	**basin whitestem rubber rabbitbrush**	Mostly Great Basin	Mostly coarse soils; low to mid-elevation
***C. n.* ssp. *consimilis* (Greene) Hall & Clements** *E. nauseosa* ssp. *consilimus* (Greene) Nesom & Baird	**threadleaf rubber rabbitbrush**	Mostly Great Basin	Mostly fine soils; low to mid-elevation
***C. n.* ssp. *graveolens* (Nutt.) Piper**	**green rubber rabbitbrush**	W Great Plains; Colorado Plateau	Wide amplitude; low to mid-elevation
***C. n.* ssp. *salicifolius* (Rydb.) Hall & Clements**	**willowleaf rubber rabbitbrush**	N Utah	Montane
***C. parryi* (Gray) Greene** *E. parryi* (Gray) Nesom & Baird	**Parry rabbitbrush**	Scattered; W US	Mostly montane
SECTION PUNCTATI*			
***C. teretifolius* (Dur. & Hilg.) Hall** *E. teretifolia* (Dur. & Hilg.)	**Mojave rabbitbrush,** Jepson green rabbitbrush	Mojave Desert	Rocky washes; hot desert

Sources: Anderson (1995), Deitschman and others (1974), USDA NRCS (2001).

quite showy. Flowering occurs from late July through October, with higher elevation populations flowering earlier. The fruits ripen in September in the mountains but may not be ripe until December in warm desert populations. There may be considerable variation in flowering and fruiting phenology within populations and even on individual plants (Meyer 1997). Each flower has the potential of producing a single narrowly cylindrical achene that is completely filled by the elongate embryo (figures 1 and 2). The achene is topped with a ring of pappus hairs that aid in dispersal by wind. The pappus may also be involved in orienting and anchoring the achene during seedling establishment (Stevens and others 1986). Fully ripened fruits are easily detached from the plant by wind under dry conditions. Abundant flowering occurs most years, but fill is variable. Sometimes there is considerable damage by noctuid moth larvae during seed development. The damaged fruits remain attached to the plant, creating the appearance of an abundant harvest after all the sound fruits have dispersed.

Seed collection, cleaning, and storage. Dry, calm conditions are best for the harvest of rabbitbrush seeds. Fully ripe fruits are fluffy and easily detachable, and they may be stripped or beaten into shoulder hoppers, bags, or boxes. Seed fill must average 30 to 50% in order to attain purities high enough for commercially profitable harvest. On favorable upland sites, harvestable crops occur in 4 of 5 years (Monsen and Stevens 1987). The purity of the bulk-harvested material is usually near 10%. Seeds are often moist at harvest and must be dried before cleaning.

Rabbitbrush seeds are difficult to clean. The elongate achenes are brittle and easily damaged in most mechanical cleaning equipment. Using flail-type cleaners such as barley debearders results in less damage than using hammermills. After initial cleaning, the material can be fanned and screened in a fanning mill to achieve the desired purity. Cleaning removes sticks and large debris, separates the achenes from the inflorescences, detaches the pappus from the achenes, and removes unfilled fruits and other fine debris.

Figure 1—*Chrysothamnus*, rabbitbrush: achenes with pappi intact of *C. viscidiflorus*, Douglas rabbitbrush (**left**) and *C. nauseosus*, rubber rabbitbrush (**right**).

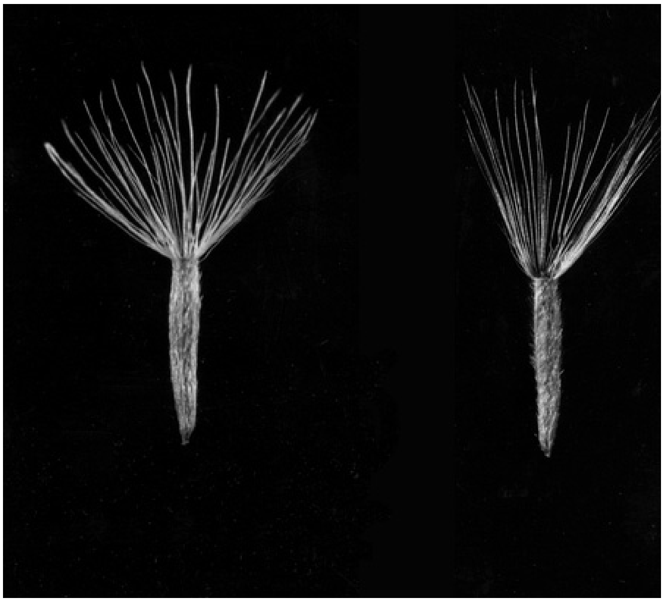
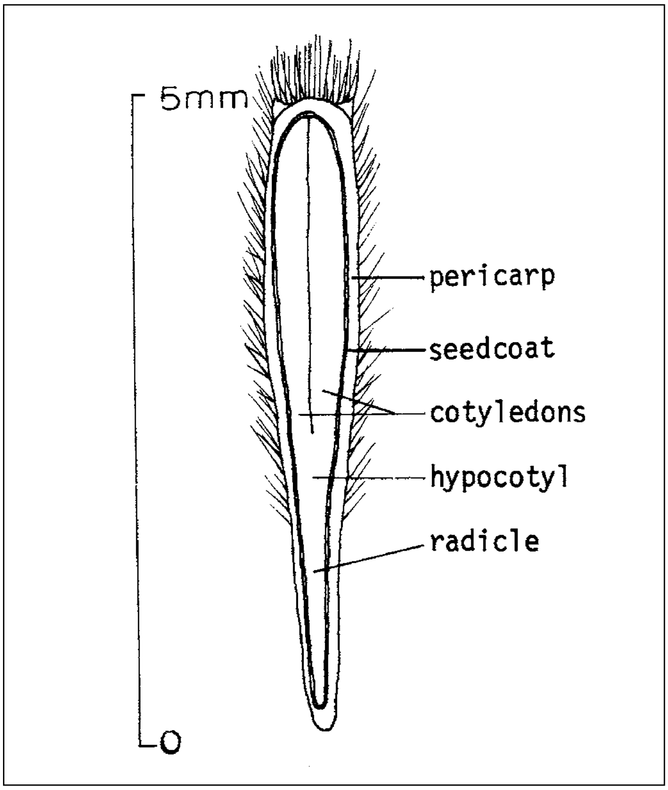


Figure 2—*Chrysothamnus viscidiflorus* spp. *lancedatus*, Douglas rabbitbrush: longitudinal section through an achene**.**



These steps are necessary to raise purities to 20% or higher and to make it possible to use conventional seeders (Monsen and Stevens 1987).

Achene weight varies substantially among species, subspecies, and populations of rabbitbrush (table 2). In rubber rabbitbrush, weight is correlated with habitat; the largest achenes come from plants that are specialized for growing on dune and badland habitats, and the smallest come from populations on temporarily open saline bottoms (Meyer 1997). There is a ninefold difference in achene weight among populations of rubber rabbitbrush, and other species also show achene weight variation (table 2). This makes it important to consider achene number per unit weight explicitly when calculating seeding rates.

Rabbitbrush seeds are not long-lived in warehouse storage. Substantial loss of viability may occur within 3 years, and storage beyond 3 years is not recommended (Monsen and Stevens 1987; Stevens and others 1981). Seeds should be retested immediately before planting so that seeding rates can be based on current values for pure live seed. Rabbitbrush seedlots with initially low vigor and viability values tend to lose their remaining viability more quickly in storage (Meyer and McArthur 1987). Because of late ripening dates, rabbitbrush seeds are usually held for a year (until the following autumn) before planting. Careful attention to moisture content (7 to 8% is probably near optimum) and storage at low temperature may prolong storage life, but data to substantiate this are lacking.

Germination. Germination requirements for rabbitbrush vary both among and within species. Rubber rabbitbrush germination is best understood (Khan and others 1987; McArthur and others 1987; Meyer and McArthur 1987; Meyer and Monsen 1990; Meyer and others 1989; Romo and Eddleman 1988). Seeds are usually nondormant at high incubation temperatures (30 °C) even when recently harvested, but display variable levels of dormancy at the intermediate temperatures characteristic of autumn seedbeds. Seeds of early-ripening high-elevation collections are most likely to be dormant or slow to germinate at autumn temperatures, whereas seeds of late-ripening warm-desert collections germinate completely and rapidly over a wide temperature range. The conditional dormancy of recently dispersed seeds is removed through moist chilling, so that all seeds are nondormant in the field by late winter. Germination rate at near-freezing temperature is even more closely tied to habitat. Collections from montane sites may

Table 2—*Chrysothamnus,* rabbitbrush: seed yield data

Species	Cleaned seeds* (x 1,000)/seed weight			
	Mean		Range	
	/kg	/lb	/kg	/lb
SECTION CHRYSOTHAMNUS				
C. linifolius	1.8	0.8	—	—
C. viscidiflorus	1.8	0.8	1.5–2.0	0.7–0.9
ssp. *lanceolatus*	1.8	0.8	1.1–2.2	0.5–1.0
	1.8	0.8	—	—
ssp. *viscidiflorus*	1.5	0.7	1.1–2.2	0.5–1.0
SECTION NAUSEOSI				
C. nauseosus	1.7	0.8	1.5–2.0	0.7–0.9
ssp. *albicaulis*	1.1	0.5	0.9–1.4	0.4–0.6
	1.5	0.7	—	—
ssp. *hololeucus*	1.3	0.6	1.1–1.5	0.5–0.7
	1.5	0.7	—	—
ssp. *consimilis*	1.5	0.7	0.9–2.4	0.4–1.1
	1.7	0.8	—	—
ssp. *graveolens*	1.3	0.6	0.9–1.1	0.4–0.5
ssp. *salicifolius*	0.9	0.4	0-9–1.1	0.4–0.5
C. parryi	0.9	0.4	—	—
SECTION PUNCTATI				
C. teretifolius	1.3	0.6	—	—

Sources: Belcher (1985), Deitschman and others (1974), Meyer (1995, 1997), McArthur and others (2004).
* 100% purity.

require more than 100 days to germinate to 50% at 3 °C, whereas warm desert collections may reach 50% germination in as few as 5 days. These germination features act in concert with seasonal patterns of temperature and precipitation in each habitat to ensure complete germination in mid to late winter. Germination is often completed just before the snow melts, with little or no carryover of seed between years. The ecotypic variation in germination phenology results in reduced emergence and survival when seed-source habitat is not matched to planting site habitat (Meyer 1990; Meyer and Monsen 1990).

Preliminary data for Intermountain and Mojave Desert populations of other species of rabbitbrush suggest that they share the same basic habitat-correlated germination patterns. Information on germination response to temperature for 6 collections of green rabbitbrush indicates that it differs from rubber rabbitbrush in having 25 °C rather than 30 °C as an optimum germination temperature and in showing some dormancy even at this optimal temperature (table 3) (Meyer 1997). Habitat-correlated germination responses at autumn and winter temperatures were similar for the 2 species and also for collections of Parry, spearleaf, and Mojave rabbitbrushes.

Evaluation of the seed quality for rabbitbrush is not without pitfalls. Reasonably repeatable purity values are obtained when only filled achenes are included as pure seed (Meyer and others 1989). Germination tests for rubber rabbitbrush should be carried out at alternating temperatures of 20 to 30 °C or a constant 25 °C for 28 days (Meyer and others 1989). This procedure is the only one listed in the official testing rules for this genus (AOSA 1993). Seedlots from low and middle elevations should complete germination within 14 days, whereas seedlots from high elevations may still show some dormancy even after 28 days, making post-test viability evaluation essential. Tests at 30 °C are not recommended, even though relative germination percentage (that is, percentage of viable seeds germinating) may be higher because there are indications that 30 °C is more stressful to marginally viable seeds.

Tetrazolium viability testing of rabbitbrush seeds is also somewhat problematical. The embryos must be removed from the achene prior to immersion in the stain because of poor penetration of the stain, even with piercing or cutting of the achene wall. The process of removal often damages the embryo, making the staining patterns hard to interpret. We frequently obtained higher viability estimates from germination tests than from tetrazolium evaluation for these species (Meyer 1997).

Nursery and field practice. Rabbitbrush species are easily propagated as container stock (Deitschman and others 1974). Seeds are sown directly into containers that provide depth for root development, sometimes after a short wet chill to ensure uniform emergence (Long 1986). The seedlings grow rapidly and are ready for outplanting in 3 to 4 months, after a hardening period. They may be outplanted in fall or spring, whenever moisture conditions are optimal. Bareroot propagation of rabbitbrush has also been successful. In spite of considerable among-lot and among-plant variation in seedling size, transplants survive quite well. Fall-seeding in nursery beds is recommended. Plants require less water than most other shrubs and should not be overwatered or fertilized excessively (Monsen 1966).

Rabbitbrushes are among the easiest shrubs to establish from direct seeding, and most plantings on wildland sites use this method. Minimal seedbed preparation is required. Surface planting onto a firm but roughened seedbed in late fall usually results in adequate stands. This planting may be accomplished through aerial seeding; hand-broadcasting; or seeding with a thimble seeder, seed dribbler, browse seeder, or a drill with the drop tubes pulled so that the seed is placed on the disturbed surface behind the disk furrow openers (McArthur and others 2004). Seeds should not be planted too deeply. One millimeter of soil coverage is sufficient. Seeding rates of about 20 to 30 live seeds/m^2 (2 to 3/ft^2) are usually adequate. This is equivalent to about 200 g/ha (ca 3 oz/ac) on a pure live seed basis for a seedlot that averages 1.5 million seeds/kg (680,400/lb). If the seedlot is cleaned to high purity, it may be necessary to dilute it with a carrier such as rice hulls in order to achieve uniform seeding rates. Seedlings emerge in early spring, and young plants grow rapidly, often producing seeds in their second growing season.

Table 3—*Chrysothamnus,* rabbitbrush: germination percentage (as percentage of total viable seeds) after 28 days at 15 °C or at 25 °C, and days to 50% of total germination during 20 weeks at 3 °C for some common species and subspecies

	Germination percentage at 15 °C			Germination percentage at 25 °C			Days to 50% germination at 3 °C		
Species	**Mean**	**Range**	**No.**	**Mean**	**Range**	**No.**	**Mean**	**Range**	**No.**
SECTION CHRYSOTHAMNUS									
C. linifolius	—	—	—	—	—	—	21	—	1
C. viscidiflorus									
ssp. *lanceolatus*	37	29–49	3	64	58–70	3	60	35–82	5
ssp. *viscidiflorus*	58	31–98	3	68	40–96	3	60	35–82	5
SECTION NAUSEOSI									
C. nauseosus									
ssp. *albicaulis*	37	29–45	2	85	78–92	2	41	9–88	2
ssp. *hololeucus*	75	17–97	8	91	74–96	8	21	7–70	12
ssp. *consimilis*	70	26–96	6	91	80–100	6	33	5–108	17
ssp. *graveolens*	76	28–100	7	91	58–100	7	33	10–105	17
ssp. *salicifolius*	25	9–55	6	65	44–92	6	89	60–100	6
C. parryi	—	—	—	—	—	—	34	12–54	4
SECTION PUNCTATI									
C. teretifolius	—	—	—	—	—	—	5	5	1

Sources: Meyer (1997), Meyer and McArthur (1987), Meyer and others (1989).

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Fabaceae—Pea family

Cladrastis kentukea (Dum.-Cours.) Rudd

yellowwood

David F. Olson, Jr., R. L. Barnes, and Jill R. Barbour

Drs. Olson and Barnes retired from the USDA Forest Service's Southeastern Forest Experiment Station;
Ms. Barbour is a germination specialist at the USDA Forest Service's National Tree Laboratory,
Dry Branch, Georgia

Synonym. *Cladrastis lutea* (Michx. f.) K. Koch

Other common names. Kentucky yellowwood, virgilia, American yellowwood.

Growth habit, occurrence, and use. Yellowwood—*Cladrastis kentukea* (Dum.-Cours.) Rudd—is a small deciduous tree that attains a height of 12 to 18 m at maturity (Sargent 1965). The native range of yellowwood is restricted; it extends from western North Carolina into eastern and central Tennessee, northern Alabama, Kentucky, southern Illinois, and Indiana; it also occurs in the glades country of southwestern Missouri and in central and northern Arkansas. Locally, it grows on limestone cliffs in rich soils, and its greatest abundance is in Missouri and in the vicinity of Nashville, Tennessee. The wood is hard, close-grained, and bright yellow, turning to light brown on exposure to light; commercially, it is a substitute for walnut in gunstocks and a source of clear yellow dye. Yellowwood is hardy as far north as New England and is often planted for its ornamental value. It was introduced into cultivation in 1812 (Olson and Barnes 1974).

Flowering and fruiting. The fragrant, perfect, white, showy flowers bloom in June, usually in alternate years, and the fruit ripens in August or September of the same year (Bailey 1949; Radford and others 1964; Sargent 1965). The fruit is a legume (pod), 7.5 to 10 cm long (figure 1) (Fernald 1950), that falls and splits open soon after maturing. The seeds are dispersed by birds and rodents. Each legume contains 4 to 6 short, oblong, compressed seeds with thin, dark brown seedcoats and without endosperm (figure 2). Weights of seeds in legumes containing 2 to 4 seeds decreased from the base of the legume to the style (Harris 1917). Good seedcrops are produced generally in alternate years.

Collection of fruits. The legumes may be collected soon after maturity by handpicking them from trees or by shaking or whipping them onto outspread canvas or plastic sheets. Legumes turn brown and split open easily at maturity.

Extraction and storage of seeds. After the legumes are allowed to dry, they can be opened by beating them in sacks or running them through a macerator. The seeds may be separated from the legume remnants with screens or air separators.

Cleaned seeds average about 24,900 to 32,200/kg (11,300 to 14,600/lb). Average purity and soundness of seeds from commercial sources have been, respectively, 82 and 67% (Olson and Barnes 1974). Seeds of yellowwood are orthodox in storage behavior and may be stored dry in sealed containers at 5 °C (Olson and Barnes 1974). For overwinter storage, seeds may be stratified in sand or a mixture of sand and peat (Olson and Barnes 1974), or they may be dried and sown the following spring (Wyman 1953).

Pregermination treatments. Natural germination of yellowwood is epigeal (figure 3) and takes place in the spring following seedfall. Dormancy is chiefly caused by an impermeable seedcoat and to a lesser degree by conditions in the embryo (Burton 1947). Burton (1947) found that shaking yellowwood seeds for 20 minutes at 400 strokes per minute made 82% of the seed permeable to water. A successful dormancy-breaking treatment is sulfuric acid scarification for 30 to 60 minutes (Heit 1967). Dormancy may be overcome also by stratification in sand or sand and peat for 90 days at 5 °C or by scarification and storage for 30 days (Olson and Barnes 1974).

An early method of overcoming dormancy includes soaking the seeds in water that is nearly at the boiling point (Jenkins 1936). The water should be preheated to 71 to 100 °C at the time the seeds are immersed; the heating element is then removed and the seeds are allowed to soak and cool for 12 to 24 hours in water (Heit 1967).

Germination tests. There are no official test prescriptions for yellowwood. Germination has been tested on pretreated seeds in sand or sand and peat flats in 30 to 42 days at 20 to 30 °C (Olson and Barnes 1974) and on moist filter paper medium for 24 days at 0, 25, and 50 °C (Rivera and others 1937). Acid-treated seeds germinated from 51 to 67% in 11 days; final germination ranged from 56 to 67% (Olson and Barnes 1974). Acid treatment for 0, 30, 60, and 120 minutes produced 5, 41, 92, and 96% germination, respectively (Frett and Dirr 1979).

Figure 1—*Cladrastis kentukea*, yellowwood: legumes.

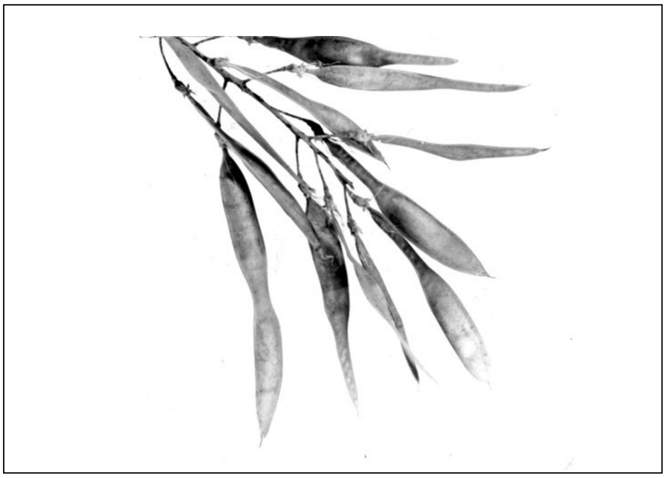


Figure 2—*Cladrastis kentukea*, yellowwood: longitudinal section (**left**) and exterior view of a seed (**right**).

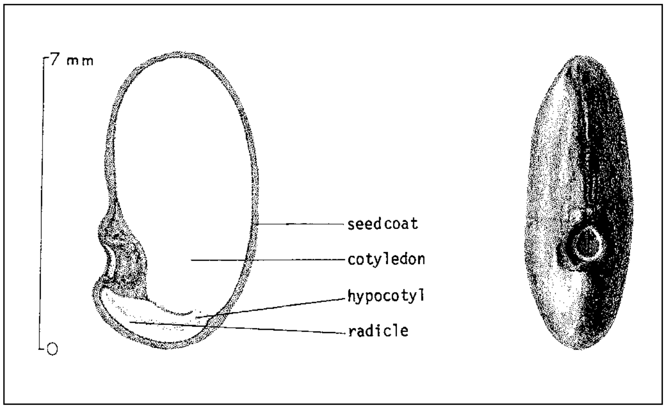
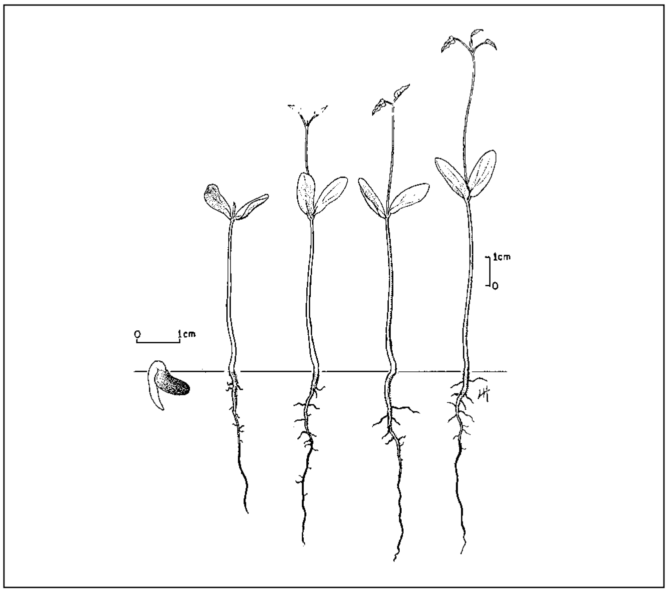


Figure 3—*Cladrastis kentukea*, yellowwood: seedling development at 1, 6, 10, 16, and 20 days after germination.



Applying hydrostatic pressure to yellowwood seeds increased their permeability in the region of the hilum and greatly increased the speed of germination (Rivera and others 1937). Pressures of 68,950 kN/m^2 (10,000 lb/in^2) applied for 10 minutes at 0 °C, 1 minute at 25 °C, and 1 minute at 50 °C resulted in 100% germination within 24 days (Rivera and others 1937). At 206,850 kN/m^2 (30,000 lb/in^2) of pressure for 1 minute or 5 minutes at 25 °C, 100% of the seeds germinated by the 12th day. However, a 20-minute exposure to a pressure of 68,950 kN/m^2 (10,000 lb/in^2) at 50 °C proved injurious to the seeds, with 15.5% of the seeds appearing soft and dead (Rivera and others 1937).

Nursery practice. Seeds may be sown in autumn or spring. Beds should be well prepared and drilled with rows 20 to 30 cm (8 to 12 in) apart, and the seeds covered with about 6 mm ($^1/_4$ in) of firmed soil. Untreated seeds may be sown in autumn and the seedbeds should be mulched and protected with bird or shade screens until after late frosts in spring. Side boards simplify mulching and screening. Stratified seeds or dry-stored seeds that have been treated to break dormancy are used for spring-sowing. If seeds were soaked in hot water at 49 °C for 24 to 36 hours until swollen and then surface-dried and planted in the nursery, they germinated readily in the spring (Dirr and Heuser 1987). Shading of seedlings is unnecessary.

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Ranunculaceae—Buttercup family

Clematis L.

clematis

John C. Zasada and Paul O. Rudolf

Dr. Zasada retired from the USDA Forest Service's North Central Forest Research Station;
Dr. Rudolf (deceased) retired from the USDA Forest Service's North Central Forest Experiment Station

Growth habit, occurrence, and use. The genus *Clematis* includes more than 200 species of climbing vines, and erect or ascending perennial herbs (sometimes woody) widely that are distributed through the temperate regions, chiefly in the Northern Hemisphere (Rehder 1940). *Clematis* is subdivided into 3 sections—Flammula (western and eastern virgin's-bowers), Atragene (western blue clematis and *C. occidentalis* (C.L. Hitchc.) Pringle), and Viorna (traveler's-joy). The taxonomy and distribution of section Atragene are described by Pringle (1971). Many horticultural varieties are grown for ornamental purposes (Dirr 1990; Lloyd 1977; Markham 1935). The 8 species included here (table 1) are also useful for erosion control, ground cover, and wildlife food (Bailey 1939; Dirr 1990; Fernald 1950; Rehder 1940; Van Dersal 1938).

Species occupy different site types within their range. In Wisconsin, for example, eastern virgin's-bower was found in 13 community types but was most abundant in the wet alder thicket community. Rock clematis is present in 2 communities and most abundant in northern dry mesic forests (Curtis 1959). Western species seem to be more common on drier well-drained sites than species native east of the Mississippi (table 1).

Geographic races. Two varieties of western virgin's-bower—*C. ligusticifolia* var. *californica* Wats. and var. *brevifolia* Nutt.—are separated geographically within the species' range (Vines 1960). These and a variety of eastern virgin's-bower—*C. virginiana* var. *missouriensis* (Rydb.) Palmer & Steyrm.—may be geographic races. Wild plants intermediate between Drummond clematis and western virgin's-bower may be of hybrid origin (Vines 1960). Several hybrids of Italian clematis are known (Rehder 1940).

Flowering and fruiting. There are both monoecious and dioecious species. Eastern virgin's-bower and western virgin's-bower (section Flammula) are dioecious, but their female flowers have non-functional stamens. Species in the sections Atragene and Viorna are monoecious (Fernald 1950). Flower size differs significantly among species, for example, eastern virgin's-bower flowers occur in clusters (panicles) containing several flowers, and their sepals are about 0.5 cm in diameter, whereas rock clematis flowers are borne singly, and their sepals are about 4 cm. Fruits are borne in heads of 1-seeded achenes with persistent feathery styles. Achenes (figures 1 and 2) are produced annually (Rudolf 1974) and are dispersed by wind in late summer or fall. Some species have been shown to produce viable seeds the first year after sowing (neoteny) (Beskaravainya 1977).

Table 1—*Clematis,* clematis: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
***C. columbiana* (Nutt.) Torr. & Gray** *C. verticillaris* var. *columbiana* (Nutt.) Gray	**rock clematis,** mountain clematis, purple clematis	Quebec to Manitoba, S to New England, West Virginia, Ohio, Wisconsin, &
***C. drummondii* Torr. & Gray**	**Drummond clematis,** Texas virgin's-bower, graybeard	NW Iowa Central & E Texas, Arizona & in Mexico on dry, well-drained soils
***C. flammula* L.** *C. pallasii* J. F. Gmel.	**plume clematis**	Mediterranean region to Iran
***C. ligusticifolia* Nutt.** *C. brevifolia* Howell	**western virgin's-bower,** western clematis, traveler's-joy	British Columbia & North Dakota S to New Mexico & California
***C. pauciflora* Nutt.**	**rope-vine**	California on dry, well-drained sites
***C. virginiana* L.** *C. catesbyana* Pursh	**eastern virgin's-bower,** Virginia virgin's-bower, eastern clematis	Maine to Georgia to Louisiana to Kansas in low woods & along streambanks
***C. vitalba* L.**	**traveler's-joy,** old-man's-beard	S Europe, N Africa, & the Caucasus Mtns.
***C. viticella* L.**	**Italian clematis,** vine-bower	S Europe & W Asia

Dates of flowering and fruiting are listed in table 2. Effects of day length and temperature on flowering and flowerbud development were reported by Goi and others (1975). Other characteristics of 8 common species are presented in table 3.

Collection of fruits and extraction and storage of seeds. Fruits are brown when ripe and may be gathered from the plants by hand, dried, and shaken to remove the seeds from the heads. Other characteristics of ripeness are when the styles have become feathery (figure 1) and the achene appears shrunken and separates easily from the head (Stribling 1986). Large quantities of fruits may be collected by means of a vacuum seed harvester, run dry through a hammermill to break up the heads, and fanned to remove debris (Plummer and others 1968).

Numbers of cleaned seeds per unit weight are listed for 7 species in table 4. Limited data for eastern virgin's-bower, traveler's-joy, and Italian clematis indicate that, in seeds not freed from the styles, purity runs from 90 to 95% and soundness about 85% (Rafn and Son 1928; Rudolf 1974). For hammermilled seeds of western virgin's-bower, a purity of 20% is acceptable in Utah (Plummer and others 1968) because separation of the broken styles from the seed is difficult and expensive. Viability of dry seeds of this species has been maintained for 2 years without refrigeration (Plummer and others 1968).

Germination. Clematis seeds have dormant or immature embryos (Dirr 1990; Dirr and Heuser 1987). Some species and hybrids may germinate over a period of from months to years (Lloyd 1977). Dirr (1990) and Dirr and Heuser (1987) also indicate that requirements for germination vary among the taxa.

Prechilling at 1 to 5 °C in moist sand, peat, or a mixture of the two for 60 to 180 days has been used to promote germination in some species (Dirr and Heuser 1987; Fordham 1960; Hartmann and others 1990; Heit 1968). Field-sowing responses of traveler's-joy and Italian clematis (Blair 1959) indicate that warm plus cold stratification may be needed. The presence of an immature embryo in Italian clematis suggests that the warm stratification allows the embryo to mature, which allows germination to occur (Clark and others 1989). Germination of seeds of *Clematis microphylla* F. Muell. ex Benth. was improved by removing the pericarp or by exposing them to a cycle of wetting and drying (Lush and others 1984). Germination of seeds of traveler's-joy collected and sown in November was lower and germination rate lower than that of seeds collected and sown in February (Czekalski 1987).

Germination tests can be run on pretreated seed in sand flats or germinators for 40 to 60 days at 20 °C (night) to 30 °C (day) (Rudolf 1974). Test results available for 4 species are shown in table 5.

Nursery practice. Only a few species are propagated from seeds because of unacceptable variation in form and flowering that detracts from their value as ornamentals (Evison 1977; Lloyd 1977). The most appropriate sowing schedule is based on species and winter conditions and will vary with geographic location. General recommendations

Figure 1—*Clematis virginiana*, eastern virgin's-bower: achenes with styles still attached.

Figure 2—*Clematis virginiana*, eastern virgin's-bower: longitudinal section through an achene.

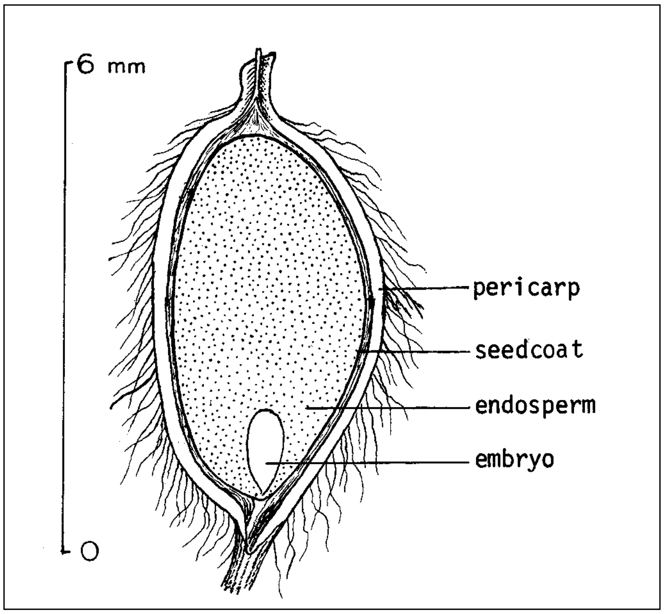

Table 2—*Clematis,* clematis: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening
C. columbiana	—	May–June	July–Aug
C. drummondii	SW US	Mar–Sept	Aug–Oct
C. flammula	—	Aug–Oct	Aug–Oct
C. ligusticifolia	California	Mar–Apr	May–Aug
	Texas	Mar–Sept	Aug–Nov
	Colorado & Utah	May–Aug	Oct–Dec
C. pauciflora	California	Mar–Apr	May–July
C. virginiana	—	July–Sept	July–Sept
	Minnesota	June–July	Aug–Sept
C. vitalba	NE US	July–Sept	July–Sept
	France	June–July	Sept–Oct
C. viticella	NE US	June–Aug	June–Aug

Sources: Fernald (1950), Loiseau (1945), McMinn (1951), Mirov and Kraebel (1939), Radford and others (1964), Rehder (1940), Rosendahl (1955), Rydberg (1922), Van Dersal (1938), Vines (1960).

Table 3—*Clematis,* clematis: size, year first cultivated, and flower color

Species	Length at maturity (m)	Year first cultivated	Flower color
C. columbiana	2.8	1797	Purple
C. drummondii	—	—	White
C. flammula	3.1–4.6	1509	White
C. ligusticifolia	0.9–12.3	1880	White
C. pauciflora	—	Before 1935	White
C. virginiana	3.7–6.2	1726	Creamy white
C. vitalba	10.2	Long cultivated	White
C. viticella	4.6	1597	Purplish

Sources: Fernald (1950), McMinn (1951), Rehder (1940), Rosendahl (1955), Vines (1960).

Table 4–*Clematis,* clematis: seed yield data

		Cleaned seeds/weight			
		Range		Average	
Species	Place collected	/kg	/lb	/kg	/lb
C. columbiana	Minnesota	—	—	141,440	64,000
C. flammula	Europe	—	—	55,250	25,000
C. ligusticifolia	California	—	—	205,530	93,000
	Utah	663,000–724,880*	300,000–328,000*	696,150*	315,000*
C. pauciflora	California	—	—	187,850	85,000
C. virginiana	Baraga Co., Michigan	402,220–446,420	182,000–202,000	424,320	192,000
C. vitalba	Europe	—	—	707,200†	320,000
C. viticella	Europe	48,620-103-870	22,000-47,000	59,670	27,000

Sources: Mirov and Kraebel (1939), Rafn & Son (1928), Rudolf (1974).

* Styles removed.

† Styles presumably removed.

are to sow untreated seeds in the fall soon after collection or to sow in the spring using seeds stratified over winter (Bailey 1939). Untreated fall-sown seeds of traveler's-joy and Italian clematis have germinated the following fall (Blair 1959). Stribling (1986) recommends the following schedule for propagating Armand clematis—*C. armandii* Franch—in central California: store seeds collected in late May in a refrigerator until September; soak in cold water for

Table 5—*Clematis*, clematis: germination test results for stratified seeds

Species	Test duration (days)	Germination capacity (%)	# Tests
C. drummondii	40	76	1
C. ligusticifolia	200	11–84	8
C. pauciflora	—	36	1
C. virginiana	60	32	1

Sources: Mirov and Kraebel (1939), Plummer and others (1968), Rudolf (1974),

24 hours and treat with a fungicide; stratify for up to 180 days at 1 to 5 °C in sealed plastic trays; and sow in March or April.

Vegetative propagation is a common practice and used exclusively to propagate most of the popular species and varieties. Procedures for vegetative propagation from cuttings, grafting, and division are discussed by Dirr and Heuser (1987), Evison (1977), Lloyd (1977), and Markham (1935).

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Clethraceae—White alder family

Clethra L.

sweet pepperbush, summersweet

Jason J. Griffin and Frank A. Blazich

Dr. Griffin is assistant professor at Kansas State University's Department of Horticulture, Forestry, and Recreation Resources, Manhattan, Kansas; Dr. Blazich is alumni: distinquished graduate professor of plant propagation and tissue culture at North Carolina State University's Department of Horticultural Science, Raleigh, North Carolina

Growth habit, occurrence, and uses. The genus *Clethra* L. comprises about 30 species native to eastern Asia, eastern North America, and Madeira (Huxley 1992; LHBH 1976). Of those, cinnamon-bark clethra and sweet pepperbush are native to eastern North America, occurring from southern Maine to Florida and west to Texas (LHBH 1976). Some taxonomists consider woolly summersweet to be a separate species in this same range, but others consider it to be a variety of sweet pepperbush (Huxley 1992; Kartesz 1994; Radford and others 1968). Japanese clethra, a native of Japan, is commonly cultivated in North America (Koller 1974). Specific geographic regions of occurrence differ among these species (table 1).

North American species of *Clethra* are deciduous shrubs or small trees with heights ranging from 3 to 10 m in their natural settings (Krüssmann 1984). Species generally grow as rounded, multi-stemmed plants that can be shaped easily into attractive small trees (Bir 1992b).

Valued for fragrant, late summer blooms and exfoliating, cinnamon-colored bark, cinnamon-bark clethra can be useful in the landscape as a specimen plant (Bir 1992b; Koller 1974) or as a hedge (Huxley 1992). Plants also fit nicely into shrub borders and are effective particularly along the edge of water (Dirr 1994). Adaptability to unfavorable environments make summersweets ideal selections for adverse planting sites. The species discussed herein perform well in both full sun and dense shade, while tolerating soil conditions ranging from drought-prone (once established) to saturated (Bir 1992b). Sweet pepperbush also has been cultivated successfully in coastal regions where it tolerates salt mist (but not salt spray), which frequently damages other plants (Bir 1993).

Geographic races and hybrids. Naturally occurring summersweets are quite variable. Although the exfoliating bark of cinnamon-bark clethra is typically cinnamon-red in color, variations of pink, chartreuse, gold, and mahogany

Table 1—*Clethra,* sweet pepperbush: nomenclature and occurrence of species cultivated in North America

Scientific name & synonym(s)	Common name (s)	Occurrence
***C. acuminata* Michx.**	**cinnamon-bark clethra,** mountain sweetpepperbush	Cliffs & mountain woods of SE Appalachian Plateau & inner Piedmont
***C. alnifolia* L.** *C. alnifolia* var. *paniculata* (Ait.) Rehd. *C. paniculata* Ait. *C. tomentosa* Lam. *C. alnifolia* var. *pubescens* Ait. *C. alnifolia* var. *tomentosa* (Lam.) Michx.	**sweet pepperbush,** summersweet, coastal sweetpepperbush	North American coastal plain, Maine to Texas, with extensions into the Carolina Piedmont; acid swamps & low moist woods
***C. barbinervis* Sieb. & Zucc.** *C. canescens* Forbes & Hemsl. *C. kawadana* Yanagita *C. barbinervis* var. *kawadana* (Yanagita) Hara *C. repens* Nakai	**Japanese clethra,** Asiatic sweet pepperbush	Hills & mountains of Japan & Korea
***C. tomentosa* Lam.** *C. alnifolia* var. *pubescens* Ait. *C. alnifolia* var. *tomentosa* (Lam.) Michx.	**woolly summersweet**	Swamps & coastal plain of North Carolina to N Florida & Alabama

Sources: Huxley (1992), Ohwi (1984), Sleumer (1967), Small (1933).

have been observed (Bir 1992b). The majority of named selections have originated from sweet pepperbush. Inflorescences of this species normally form erect racemes (LHBH 1976). However, inflorescences occur occasionally as branched panicles (Everett 1981), in which case the plants are often classified as C. *alnifolia* var. *paniculata* (Ait.) Rehd. or *C. paniculata* Ait. (Everett 1981; Huxley 1992; LHBH 1976). Dirr (1994) cited many cultivars in detail. Some of the more outstanding selections include *C. alnifolia* 'Compacta', a compact, 1.0- to 1.2-m-tall selection with lustrous, dark green foliage; 'Creel's Calico', the only variegated selection; 'Fern Valley Late Sweet', a late-flowering, almost columnar selection; and 'Hummingbird', unquestionably the most popular selection, which grows to a height of 0.8 to 1.0 m, with lustrous, dark green foliage that is covered by fragrant white flowers in mid to late summer. Of the pink-flowering forms, 'Pink Spires' and 'Rosea' are most common (Dirr 1994). These cultivars frequently are indistinguishable and may in fact be the same clone. 'Ruby Spice' occurred as a bud sport on 'Rosea' and is distinguished by deeper pink flowers that do not fade in late season. Another pink selection, 'Fern Valley Pink', produces inflorescences that can reach lengths of 20 to 25 cm for a spectacular floral display.

Flowering and fruiting. Fragrant white flowers, about 1 cm in diameter, are borne on upright or horizontally held terminal racemes or panicles, to 15 cm long (Huxley 1992), arising from the axils of leaves (Everett 1981). Flowering begins in July (with the exception of woolly summersweet, which flowers in August) and lasts to September (Krüssmann 1984), making summersweets excellent selections for late summer color. Pollination of perfect flowers most likely occurs by bees, which can be a nuisance if plants are located near walks or sitting areas (Bir 1992b; Dirr 1994; Koller 1974). Fruits are subglobose, 3-valved capsules, ranging from 2.5 to 5.0 mm in length with a persisting style and calyx (figure 1) (Huxley 1992). Upon maturation, capsules split to release many seeds. Seeds are quite small and irregularly angled, and dispersal is presumably by wind (figures 2 and 3) (Sleumer 1967). Sweet pepperbush in New Jersey averaged 6 to 17 seeds per capsule from 3 collection sites, with total seed production per plant ranging from 1,348 to 7,920 (Jordan and Hartman 1995).

Collection of fruits, seed extraction, cleaning, and storage. It appears that seeds do not mature until long after leaf abscission, November in North Carolina (Bir 1992a). Thus, collecting and sowing seeds before maturation may result in poor or no germination. Once seeds have matured, capsules can be collected before they open and

Figure 1—*Clethra*, sweet pepperbush: fruits (capsules) of *C. acuminata*, cinnamon-bark clethra (**top**); *C. alnifolia*, sweet pepperbush (**bottom**).

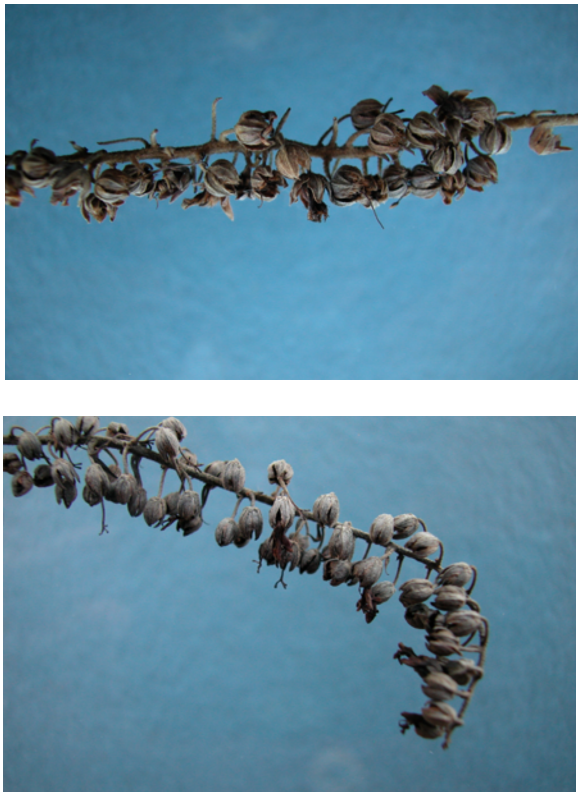
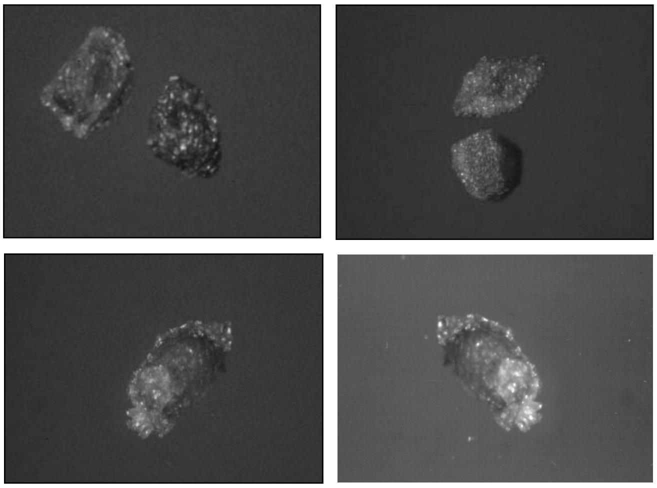


Figure 2—*Clethra*, sweet pepperbush: seeds of *C. acuminata*, cinnamon-bark clethra (**top left**); *C. alnifolia*, sweet pepperbush (**top right**); *C. barbinervis*, Japanese clethra (**bottom left**); and *C. tomentosa*, woolly summersweet (**bottom right**).



allowed to dry until they split. Seeds can then be shaken from the capsules and cleaned (Dirr 1994; Dirr and Heuser 1987). There are no reports of long-term storage, but seeds can be stored successfully for short periods at low temperatures (5 °C) and moisture contents (Bir 1992a; Dirr and Heuser 1987). The seeds are therefore apparently orthodox in storage behavior.

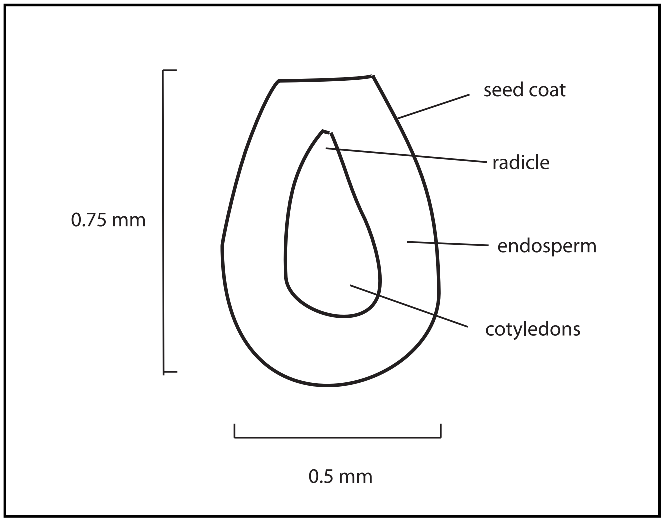
Pregermination treatments and germination testing. Jordan and Hartman (1995) reported up to 58% germination with New Jersey sources of sweet pepperbush after 5 months of stratification at 0 to 2 °C in the dark. Their germination regime was 16 hours of light at 30 °C and 8 hours in the dark at 15 °C. Other reports, however, suggest that no pretreatments are required and that germination occurs readily if seeds are sown immediately following collection (Bir 1992a; Dirr and Heuser 1987).

For successful germination seeds should be treated similar to those of azalea (*Rhododendron* L.) (Bir 1992a&b; Dirr and Heuser 1987). In general, when mature seeds are sown on the surface of a germinating medium and placed under mist at 24 °C, germination occurs within 2 weeks and is complete within 1 month (Bir 1992b).

Nursery practice. When seedlings are grown in a typical azalea growing medium of 3 parts pine bark to 1 part peat (vol/vol)—amended with 4.2 kg/m^3 (7.0 lb/yd^3) dolomitic limestone and fertilized following recommendations for azaleas—seedlings grow well, filling a 3.8-liter (1-gal) pot by the end of a growing season (Bir 1992b). Although naturally occurring as an understory species, seedlings of cinnamon-bark clethra show no symptoms of stress when grown in full sun and are visually no different than seedlings maintained under 50% shade (Bir 1992b). The species is well adapted to dry soils once established. However, if seedlings are exposed to drought conditions before a sufficient root system has developed, high mortality can be expected (Bir 1992b).

Asexual propagation of species of summer-sweet is very easy and is widely used for propagation of particular cultivars. Species listed in table 1 are propagated readily by stem cuttings taken during the summer, as well as by root cuttings taken during December and January (Dirr and Heuser 1987).

Figure 3—*Clethra alnifolia*, sweet pepperbush: longitudinal section of a seed.



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Rosaceae—Rose family

Coleogyne ramosissima Torr.

blackbrush

Burton K. Pendleton

Dr. Pendleton is a research plant ecologist at the USDA Forest Service's Rocky Mountain Research Station, Forestry Sciences Laboratory, Albuquerque, New Mexico

Growth habit, occurrence, and use. Blackbrush—*Coleogyne ramosissima* Torr.—grows in the transition zone between warm and cold deserts of southern California, southern Nevada, southern Utah, northern Arizona, and southwestern Colorado. It is found at elevations of 760 to 1,980 m. Ranging from 0.3 to 1.2 m in height, blackbrush forms almost monotypic stands in the lower Mojave–Great Basin ecotone, bounded by creosote bush (*Larrea tridentata* (Sesse & Moc.) ex DC. Coville.) communities at low elevations and by juniper–big sagebrush (*Artemesia tridentata* Nutt.) communities at higher elevations. In the eastern part of its range, blackbrush is bordered by Sonoran communities on the south and by big sagebrush, juniper, and mixed shrub communities of the Colorado Plateau in the north. Distribution of blackbrush is limited by soil depth, temperature extremes, and moisture availability.

Blackbrush occurs as a landscape dominant over much of its range and forms a major vegetational component of national and state parks in Utah, Nevada, and California. Blackbrush provides significant year-round forage for desert bighorn sheep (*Ovis canadensis*) and is eaten by mule deer (*Odocoileus hemionus*) in winter. Foliage may be grazed by domestic goats and sheep in spring, but receives minimal use by cattle. Blackbrush provides habitat to many small mammals, and its seeds are eaten by both rodents and birds.

Blackbrush, although a monotypic genus, occurs over a wide geographic and elevational range. Differences in plant size and germination characteristics suggest ecotypic variability. Use of locally adapted seed collection sites should improve chances for successful propagation and establishment.

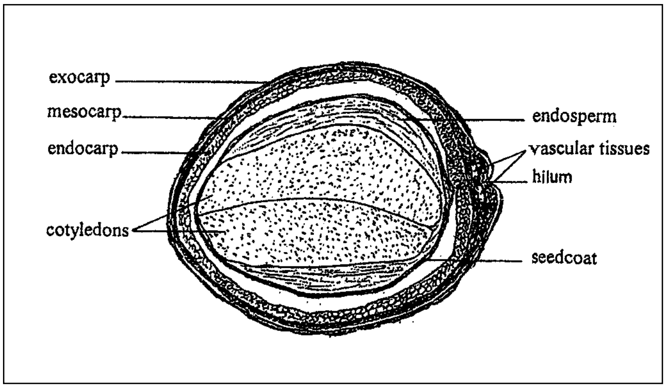
Flowering and fruiting. Flowers are perfect, apetalous, with yellow sepals 4.5 to 6.5 mm in length; however, rare individuals with 1 to 4 yellow petals can be found in most populations (Welsh and others 1987). Flowering occurs from late March through early May, each population flowering for 2 to 3 weeks (Bowns and West 1976), with individual plants lasting some 7 to 10 days. Flowering is induced by fall and winter rains, and the timing and degree of flowering varies significantly from year to year (Beatley 1974). Ripe achenes are reddish brown in color (figure 1). The fruit is an ovate glabrous achene, somewhat curved in shape, and 4 to 8 mm long (figure 2). Blackbrush is wind-pollinated (Pendleton and Pendleton 1998).

Blackbrush is mast-fruiting: the size of the seedcrop is related to plant resource reserves. The mast crop comprises almost all of the seed production at low-elevation sites, whereas some seeds are produced in the more mesic higher-elevation sites in all but the driest intermast years. Periods between mast seedcrops often exceed 5 years. Late frosts can reduce or eliminate flowering and fruit production.

Seed collection, cleaning, and storage. The ripe seeds readily separate from the floral cup. Natural seed-fall is correlated with rain showers, which dislodge the achenes from the floral cup. The fruit ripens between late May and the third week of July, depending upon elevation and year-to-year variation. Harvesting is accomplished by beating the branches with a stick or board. Fruits can be collected onto a tarp spread under the shrub or into a basket or hopper (Nord

Figure 1—*Coleogyne ramosissima*, blackbrush: fruits with and without pubescence.

Figure 2—*Coleogyne ramosissima*, blackbrush: longitudinal section of achene with seed (drawing courtesy of Dr. Emerencia Hurd, retired, USDA Forest Service, Boise, ID).



1962). Seed collections contain a significant amount of debris and cleaning is required. Seeds can be cleaned in a fanning mill or with a gravity separator (Monsen 2004). A portion of the achenes are retained within the floral cup. These can be removed with a barley de-bearder or through use of a rubbing board.

Viability of cleaned seeds is generally high, and the incidence of insect damage is extremely low. Twenty-four collections from Utah and Nevada populations ranged in viability from 74 to 98%. Nineteen collections had viability percentages greater than 85%. The number of cleaned seeds per weight averages 60,000/kg (27,000/lb), with a range of 47,500 to 68,000/kg (21,500 to 31,000/lb).

Seeds of blackbrush are long-lived and orthodox in storage behavior; they can be stored in a cool dry location for long periods without loss of viability (table 1). Germination tests on seeds collected in Washington County, Utah, showed no loss in viability after 10 and 15 years. Plants were produced from this seedlot 12 years after collection. However, the vigor of older seeds (10+ years) in field plantings has not been determined.

Germination. Fresh blackbrush seedlots are 68 to 95% dormant and remain dormant in laboratory storage for the first year after collection (table 1). Seed dormancy increases with increasing elevation of the seed source. Five-year-old seeds are essentially nondormant and will readily germinate at cool temperatures (–15 °C) (Pendleton and others 1995). Stratification of fresh seeds for 4 to 6 weeks at 1 °C will produce rapid maximum germination of all collections when seeds are removed from chill and placed at temperatures between 5 and 25 °C. Under field conditions, seeds are nondormant by October, and germination can occur at this time given proper moisture conditions and cool soil temperatures. Field germination typically occurs during the winter and early spring (figure 3) (Meyer and Pendleton 2002).

Nursery and field practice. Container stock can readily be produced from seeds. However, both stratified and unstratified seeds will germinate and emerge over a long period of time (up to 1 year). The most efficient method to synchronize germination and produce uniform-aged plants is to plant germinated seeds. Fresh seeds should be stratified between moist blotter paper for 6 weeks at 1 °C. When seedlots are removed from 1 °C and kept at room temperature, more than 75% will germinate in 24 to 48 hours. Seed collections 3 years old or older, stratified for 3 to 4 weeks, produce similar results. Germinated seeds, with radicals 2 to 10 mm (0.1 to 0.4 in) long, should be planted about 2 cm (0.8 in) deep in a well-draining soil mix. However, using a soil medium that retains moisture often leads to problems with damping-off diseases. Under experimental greenhouse conditions, blackbrush responds positively to inoculation with arbuscular mycorrhizal fungi (Pendleton and Warren

Table 1—*Coleogyne*, blackbrush: germination of nonstratified and stratified* seeds

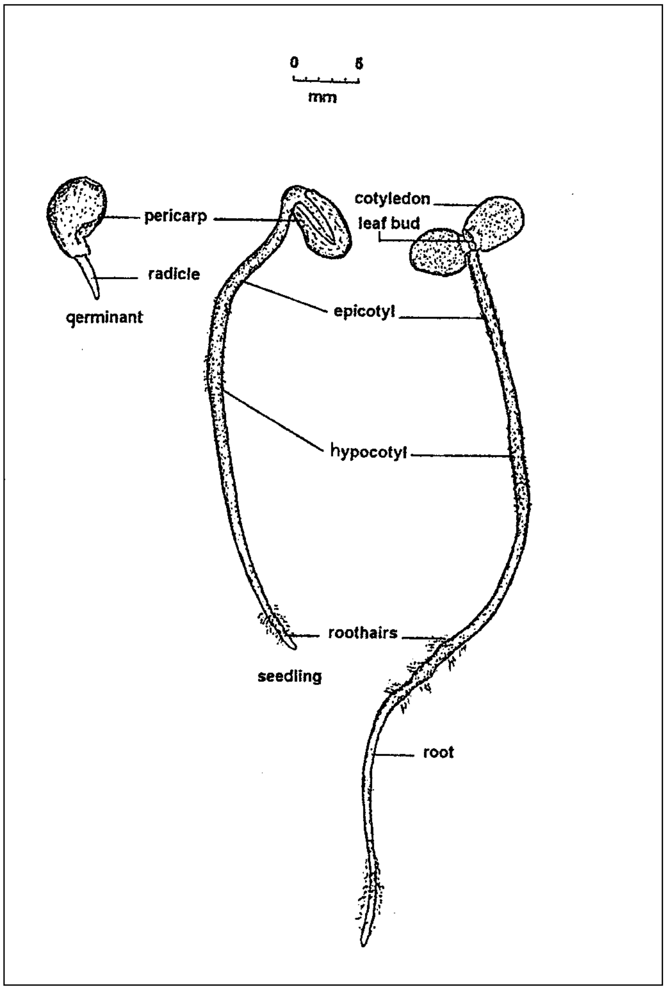
	Percent germination†		
Seed age	**Range**	**Mean**	**Collections**
Fresh			
Stratified	5–32	17.6	10
Nonstratified	84–98	91.8	10
1 year			
Stratified	0–13	6.5	10
Nonstratified	85–95	92.0	10
5 years			
Nonstratified	82–96	90	6

Source: Pendleton and Meyer (2002).

* Stratification was a moist chilling for 4 weeks at 1 °C.

† Germination tests done on wet blotter paper with a 12-hour alternating temperature regime of 5 and 15 °C; mean percent viability of these collections > 90%.

Figure 3—*Coleogyne ramosissima*, blackbrush: germination and seedling development (courtesy of Dr. Emerencia Hurd, retired, USDA Forest Service, Boise, ID).



1996). Addition of mycorrhizal inoculum to the planting medium should be considered. Optimal growing temperature for seedling growth is about 20 °C (Wallace and others 1970; Wallace and Romney 1972). Warmer conditions result in slow growth and plants that enter dormancy. Greenhouse-grown plants are susceptible to aphid infestation, but blackbrush is tolerant of standard control methods.

When seeds are not available, plants can be produced from stem cuttings. About half of the cuttings taken from current-year growth (June or September) and treated with 0.8% indole butyric acid (IBA) or commercial rooting hormone produced roots (Hughes and Weglinski 1991).

Outplanted container stock, whether produced from seeds or cuttings, should be protected from herbivory by tree tubes or diamond netting. Container stock has successfully been outplanted at Joshua Tree National Monument in California (Holden 1994) and on a natural-gas pipeline right-of-way in Arches National Park in Utah.

A limited number of attempts to establish blackbrush through seeding have been reported. In general, these attempts have had poor success (Monsen 2004). Factors that may have been responsible include low moisture levels during the germination season, lack of sufficient seeds, seeding at a less than optimal time, weed competition, herbivory, and seed theft by rodents. Seed availability is a major limiting factor in blackbrush reestablishment. Mast crops of seeds occur infrequently (5- to 10-year periods) and should be collected and stored for future use. Blackbrush seeds maintain good viability when stored for these time periods.

Insufficient work has been done to firmly determine the seeding rates and seedbed conditions necessary to establish blackbrush stands from seeds, but experimental work and reported successful seedings do offer some guidelines. Rodent-cached seeds are found at depths of 1 to 3 cm (0.4 to 1.2 in), suggesting that conventional seeders should be set to this planting depth. Because blackbrush does exhibit ecotypic variation, locally collected seeds should be used. Seeding should be done in the late summer or fall. Germination and emergence occurs as early as November (Graham 1991) and, more typically, January to March (Bowns and West 1976; Meyer and Pendleton 2002).

Spring seedings have also been attempted. Blackbrush was included in seed mixes used in experimental restoration plantings at the Nevada Test Site in southern Nevada. The seedings were conducted during March 1993. Although no blackbrush seedlings emerged that spring, some seedlings were observed during subsequent springs (USDoE 1994). An experimental spring-seeding conducted in southeastern Utah in March 1992 included stratified and unstratified seeds from 4 populations. Of the stratified seedlots, 12.5% emerged during spring 1992; of the unstratified seedlots, <1%. During spring 1993, an additional 16% of the stratified and 62% of the unstratified seedlots emerged. Lots of stratified seeds produced half as many seedlings as unstratified seeds from March 1992 through May 1993 (Pendleton and Meyer 2002). Blackbrush will form a short-term seedbank during drought conditions, but most, if not all, seeds will germinate when moisture is adequate for winter germination.

As with all dryland and desert species, successful establishment from seeds depends on the availability of moisture during the germination and establishment periods. Spring and early summer precipitation is not the norm for Mojave and Sonoran blackbrush communities, a fact that makes blackbrush seeding establishment in these areas difficult, especially in lower-elevation sites.

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Fabaceae—Pea family

Colutea L.

bladder-senna

Paula M. Pijut

Dr. Pijut is a research plant physiologist at the USDA Forest Service, North Central Research Station's Hardwood Tree Improvement and Regeneration Center, West Lafayette, Indiana

Growth habit, occurrence, and use. The genus *Colutea*—the bladder-sennas—includes about 26 species of deciduous shrubs or small trees, with a distribution ranging from the Mediterranean region and southeastern Europe to northwest Africa and the western Himalayas (Browicz 1963, 1967; Hillier 1991; Krüssmann 1984; LHBH 1976). The 3 taxa of interest in the United States are common bladder-senna (*C. arborescens* L.), *C. orientalis* Mill., and *C.* × *media* Willd. (table 1). Bladder-senna species are cultivated in temperate climates primarily for ornamental purposes but may also be used for erosion control (Krüssmann 1984). In Spain, the potential use of common bladder-senna as a forage crop has been investigated because of its ligneous nature and summer utility (Allue Andrade 1983a). Antifungal compounds have been isolated from root bark of common bladder-senna (Grosvenor and Gray 1998). The bladder-sennas are very distinct shrubs, and the common name is derived from their large, inflated legumes (pods).

Common bladder-senna is a vigorous shrub of bushy habit, with medium to fast growth. It prefers a sunny location (Dirr 1990) but is easily grown in almost any soil type (except waterlogged). The cultivar 'Bullata' is a dwarf form with dense habit (about $^1/_3$ to $^1/_2$ the size of the species at maturity) whose 5 to 7 leaflets are small, rounded, and somewhat bullate (Dirr 1990; Krüssmann 1984). The cultivar 'Crisp' is a low-growing form with leaflets that are sinuate (Dirr 1990; Krüssmann 1984). *Colutea orientalis* is a rounded shrub with attractive glaucous leaflets (Hillier 1991; Krüssmann 1984). *Colutea* × *media* is a recognized as a hybrid (*C. arborescens* × *C. orientalis*), with bluish green foliage, that originated before 1790 (Dirr 1990; Krüssmann 1984).

Flowering and fruiting. The papillionaceous flowers are about 2 cm in length, bloom from May to July (with scattered blossoms into September), and occur in axillary, long-stalked racemes (Dirr 1990; Krüssmann 1984; LHBH 1976). The pea-shaped flowers of common bladder-senna are yellow, the standard petal having red markings; the flowers of *C. orientalis* are a reddish brown or copper color; and those of *C. media* range in color from the typical yellow to those which blend through markings or tints of copper, pink, or reddish brown (Krüssmann 1984; LHBH 1976). The fruit is a inflated, bladder-like legume, 6 to 7.6 cm long and 2.5 to 3.8 cm wide, that varies in color from lime green to tints of pink or bronze and is very ornamental (Dirr 1990; Krüssmann 1984). Fruits mature from July to September (Dirr 1990) and each legume contains several small seeds (figure 1) (Rudolf 1974). The legumes of *C. orientalis* dehisce at the tip.

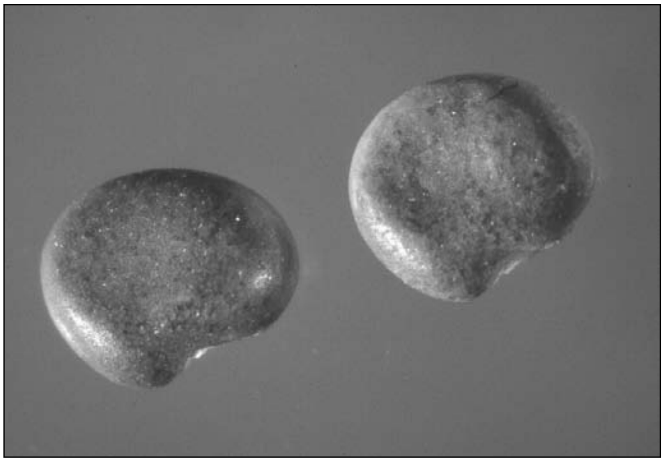
Collection of fruits; extraction, cleaning, and storage of seeds. Ripe legumes can be harvested from the shrubs in late summer or fall and then spread in a shed (with good air circulation) to dry (Rudolf 1974). The legumes are threshed to remove the seeds and the debris is fanned out (Rudolf 1974). Bladder-sennas average 74,956 seeds/kg (34,000/lb) (Allen 1995). Dry seeds stored at 5 °C in glass

Table 1—*Colutea*, bladder-senna: morphological characteristics, height at maturity, and date first cultivated

Scientific name	Leaflets	Flowers/ raceme	Height at maturity (m)	Year first cultivated
C. arborescens	9–13	6–8	1.8–4.5	1570
C. orientalis	7–11	2–5	2	1710
C. x media	11–13	Varies	1.8–3.0	1809

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

Figure 1—*Colutea* arborescens, common bladder-senna: seeds.



containers will be viable for 1 to 3 years, depending upon the species. Like most genera in Fabaceae, this genus is orthodox in storage behavior. Seeds can be stored in liquid nitrogen without a significant loss in germination percentage (Gonzalez-Benito and others 1994; Iriondo and others 1992).

Pregermination treatments. Bladder-senna seeds do not germinate readily unless the impermeable seedcoat is ruptured by mechanical or chemical scarification. Soaking the seeds in concentrated sulfuric acid for 30 to 60 minutes, before sowing in nursery beds, results in good germination (Dirr 1990; Dirr and Heuser 1987). Steeping seeds in water that was initially brought to 88 °C and then allowed to cool 24 hours also results in good seed germination (Allue Andrade 1983b; Dirr 1990; Dirr and Heuser 1987).

Germination tests. Pretreated bladder-senna seeds can be tested in germinators at 20 °C night and 30 °C day for 30 days (Rudolf 1974).

Nursery practice and seedling care. Untreated seeds may be sown in the fall, but scarified seeds are required for spring-sowing (Allen 1995; Dirr and Heuser 1987). Seedlings germinate within 1 to 2 weeks and grow rapidly. Bladder-senna species may also be propagated by cuttings. In England, 29% of half-ripened cuttings taken in early November rooted without treatment; the cuttings failed to respond to naphthaleneacetic acid (NAA); and 73% rooted after treatment with 0.1 g/liter (100 ppm) indole-3-butyric acid (IBA) solution for 18 hours (Dirr 1990; Dirr and Hesuer 1987). Summer softwood cuttings should be treated with about 1 to 3 g/liter IBA solution (1,000 to 3,000 ppm) or talc formulation (Dirr and Heuser 1987). Bladder-senna plants develop a thin, rangy root system that makes transplanting difficult. Growing plants in containers is the preferred production method.

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Cornaceae—Dogwood family

Cornus L.

dogwood

Kenneth A. Brinkman and Victor Vankus

Dr. Brinkman retired from the USDA Forest Service's North Central Experiment Station;
Mr. Vankus is a botanist at the USDA Forest Service's, National Seed Laboratory, Dry Branch, Georgia

Growth habit, occurrence, and use. About 40 species of dogwood—*Cornus* L.—are native to the temperate regions of the Northern Hemisphere, and 1 is found in Peru. Most species are deciduous trees or shrubs (2 are herbs) useful chiefly for their ornamental qualities—flowers, fruit, foliage, or color of twigs. Many varieties have been developed for a number of the species for their landscape or horticultural value. The wood of flowering dogwood, the most commercially important species in the United States, is hard and heavy and was used extensively by the textile industry earlier in the 20th century for shuttle blocks. Today the species is widely known due to its popular use as an ornamental landscape tree. Some species produce edible fruits (Edminster 1950; Edminster and May 1951), and the bark of others contains a substitute for quinine. Roots and bark of several species have long been known to have medicinal properties that can be used to fight fevers. Distribution data and chief uses of 17 species of present or potential importance in the United States are listed in table 1.

Flowering and fruiting. The small, perfect flowers—white, greenish white, or yellow in color—are borne in terminal clusters in the spring. In flowering and Pacific dogwoods, the clusters are surrounded by a conspicuous enlarged involucre of 4 to 6 white or pinkish petal-like, enlarged bracts. Fruits are globular or ovoid drupes 3 to 6 mm in diameter, with a thin succulent or mealy flesh containing a single 2-celled and a 2-seeded bony stone (figures 1 and 2). However, in many stones, only 1 seed is fully developed, but larger stones generally have 2 developed seeds. The fruits ripen in the late summer or fall (table 2). Data on minimum seed-bearing age and fruiting frequency are limited (table 3). Stones are dispersed largely by birds and animals.

Collection of fruits. Dogwood fruits should be collected when the fruit can be squeezed and the stone will pop out. To reduce losses to birds, fruits should be collected as soon as they are ripe by stripping or shaking them from the branches. Short ladders may be useful for collecting fruits from the taller species, but ordinarily this can be done from the ground. Fruits of flowering dogwood should not be collected from isolated trees because these seem to be self-ster-

Figure 1—*Cornus*, dogwood: cleaned seeds of *C. alternifolia,* alternate-leaf dogwood (**top left**); *C. amomum*, silky dogwood (**top center**); *C. sericea* ssp. *orientalis*, California dogwood (**top right**); *C. drummondii*, roughleaf dogwood (**middle left**); *C. florida*, flowering dogwood (**middle center**); *C. nuttallii*, Pacific dogwood (**middle right**); and *C. racemosa*, gray dogwood **(bottom)**.

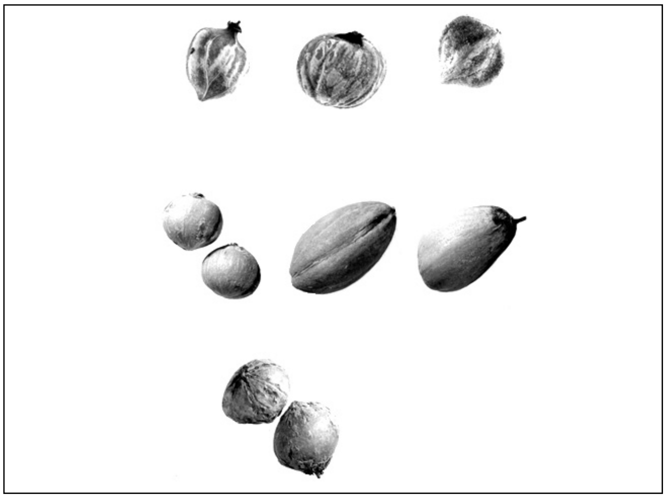


Figure 2—*Cornus sericea*, red-osier dogwood: longitudinal section through an embryo of a stone **(left)**; transverse section of a stone containing 2 embryos (**right top**) and transverse section of a stone containing 1 embryo (**right bottom**).

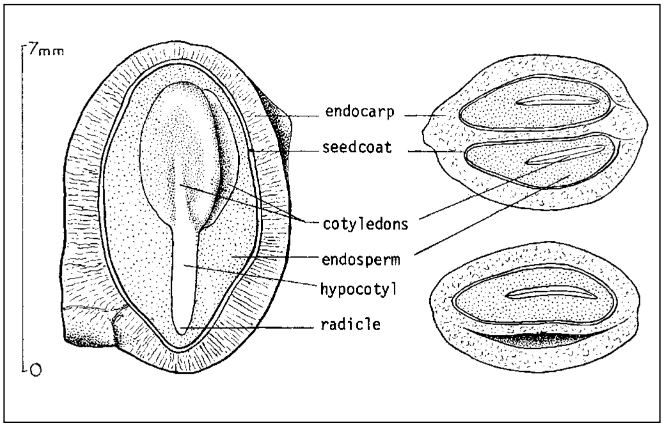


Table 1—*Cornus*, dogwood: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
C. alba **L.** *C. tatarica Mill.*	**Tatarian dogwood**	Siberia to Manchuria & North Korea
C. alternifolia **L. f.** *Swida alternifolia* (L.f.) Small	**alternate-leaf dogwood,** blue dogwood, pagoda dogwood	Newfoundland to SE Manitoba, S to Missouri & E Arkansas, E to Georgia
C. amomum P. Mill.	**silky dogwood,** kinnikinnik, red-willow	Maine to Indiana, S to Georgia & Florida
C. canadensis **L.** *Chamaepericlymenum canadense*(L.) Aschers & Graebn. *Cornella canadensis* (L.) Rydb.	**bunchberry,** bunchberry dogwood, dwarf cornel	S Greenland to Alaska, S to Maryland, W to South Dakota, New Mexico, & California
C. controversa **Hems.**	**giant dogwood**	Japan, China, & Nepal
C. drummondii **C. A. Mey.** *C. priceae Small*	**roughleaf dogwood**	S Ontario, Ohio, & Kentucky, W to Nebraska, S to Texas & Mississippi
C. florida **L.** *Cynoxylon floridum* (L) Raf. ex. B.D. Jackson	**flowering dogwood,** dogwood	E United States
C. kousa **Hance**	**Japanese dogwood,** kousa dogwood	Japan & Korea
C. macrophylla **Wall.**	**bigleaf dogwood**	Japan, China, & Nepal
C. mas **L.**	**cornelian-cherry,** cornelian-cherry dogwood	Central & S Europe & W Asia
C. nuttallii **Audubon ex Torr. & Gray**	**Pacific dogwood,** western flowering dogwood, mountain dogwood	SW British Columbia, W Washington & Oregon, S in mtns to S California; also in central W Idaho
C. officinalis **Siebold & Zucc.**	**Japanese cornelian-cherry,** Japanese cornel dogwood	Japan, Korea, & China
C. racemosa **Lam.** *C. foemina* ssp. *racemosa* (Lam.) *C. circinata* L'Herit. J.S. Wilson *C. paniculata* L'Herit.	**gray dogwood,** western dogwood	Maine to Manitoba, S to Florida, W to Missouri & Oklahoma
C. rugosa **Lam.** *C. circinata* L'Herit.	**roundleaf dogwood,** roundleaved dogwood, roundleaf cornel	Quebec to Manitoba, S to Virginia, W to NE Iowa
C. sanguinea **L.** *C. sanguinea* var. *viridissima* Dieck *Swida sanguinea* (L.) Opiz	**bloodtwig dogwood,** common dogwood, dogberry, pegwood	Europe
C. sericea **L.** *C. stolonifera* Michx. *C. baileyi* Coult. & Evans *Suida stolonifera* (Michx.) Rydb.	**red-osier dogwood,** American dogwood, kinnikinnik, squawbush	Newfoundland to Alaska, S to California, New Mexico, & Nebraska, in NE US from Wisconsin to New York
C. sericea **ssp.** ***occidentalis*** **(Torr.& Gray) Fosberg**	**western dogwood,** California dogwood, creek dogwood	S British Columbia to N Idaho, S to S California

Source: Brinkman (1974).

ile, and a high percentage of the stones will be empty (Mugford 1969).

Extraction and storage of seeds. The stones can be readily extracted by macerating the fruits in water and allowing the pulp and empty stones to float away (see chapter 3 on seed processing) (Brinkman 1974; Mugford 1969). Stone yields and weights are summarized in table 4. If the fruits cannot be extracted immediately after fruits are collected, they should be spread out in shallow layers to prevent excessive heating; however, slight fermentation facilitates removal of the fruit pulp (Brinkman 1974; NBV 1946). Clean air-dried stones may be stored in sealed containers at 3 to 5 °C (Heit 1967; Mugford 1969; Sus 1925; Swingle 1939). Stones of flowering dogwood have been successfully stored at 4% moisture content at –7 °C for 7 years by the Georgia Forestry Commission with only a 1% decrease in viability (Brock 1997), thus demonstrating the orthodox nature of seeds of this genus. Brinkman (1974) wrote that dogwood stones could be sown without extracting them from the fruit and that stones were cleaned when storage was required and that commercial seedlots may or may not have the dried fruit attached. Presently, however, all commercial lots of dogwood seeds now are cleaned (table 4) and some nursery managers report that if the stones are not cleaned, the fruits may inhibit germination (Brock 1997). After the fruits are collected and cleaned, the stones may be sown immediately or stratified for spring-planting.

Pregermination treatments. Natural germination of most species occurs in the spring following seedfall, but some seeds do not germinate until the second spring. Germination is epigeal (figure 3). Seeds of all species show delayed germination due to dormant embryos; in most species, hard pericarps also are present. Where both types of dormancy exist warm stratification for at least 60 days in a

Table 2—*Cornus,* dogwood: phenology of flowering and fruiting

Species	Flowering	Fruit ripening	Seed dispersal
C. alba	May–June	Aug–Sept	—
C. alternifolia	May–July	July–Sept	July–Sept
C. amomum	May–July	Aug–Sept	Sept
C. canadensis	May–July	Aug	Aug–Oct
C. controversa	May–June	Aug–Sept	Oct
C. drummondii	May–June	Aug–Oct	Aug–winter
C. florida	Mar & Apr (S US)–May (N US)	Sept (N US)–Oct (S US)	Sept–Nov
C. kousa	May–June	Aug–Oct	—
C. macrophylla	July–Aug	—	—
C. mas	Feb–March	Aug–Sept	—
C. nuttallii	April–May	Sept–Oct	Sept–Oct
C. officinalis	Feb–Mar	Sept	—
C. racemosa	late May–July	July–Oct	Sept–Oct
C. rugosa	May–July	Aug–Sept	—
C. sanguinea	May–June	Aug–Sept	—
C. sericea	May–July, June–Aug (N US)	July–Oct	Oct–winter
C. sericea ssp. *occidentalis*	Apr–Aug	July–Nov	—

Sources: Asakawa, (1969), Billington (1943), Brinkman (1974), Dirr (1990), Fernald (1950), Forbes (1956), Holweg (1964), Gordon and Rowe (1982), Lakela (1965), McMinn (1951), Ohwi (1965), Rehder (1940), Rosendahl (195), Rydberg (1932), Steyermark (1963), Van Dersal (1938), Vimmerstedt (1965), Weaver (1976), Wyman (1947).

Table 3—*Cornus,* dogwood: height, seed-bearing age, seedcrop frequency, and fruit ripeness criteria

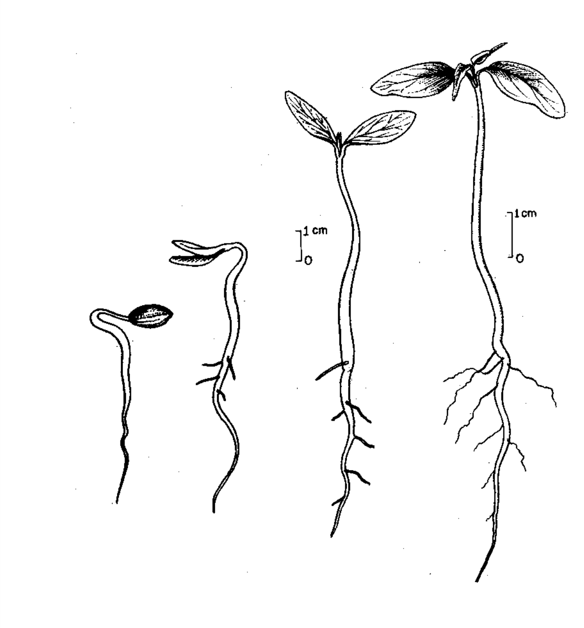
Species	Height at maturity (m)	Year first cultivated	Min seed-bearing age (yrs)	Years between large seedcrops	Ripe fruit color
C. alba	3	1741	—	—	Bluish white
C. alternifolia	5–8	1760	—	—	Dark blue
C. amomum	3	1658	4–5	1	Pale blue or bluish white
C. canadensis	0.3	—	—	—	Bright red or scarlet
C. controversa	9–18	1880	—	—	Red or purple to blue-black
C. drummondii	8–14	1836			White
C. florida	6–12	1731	6	1–2	Dark red
C. kousa	8	1875	—	2	Rose red pinkish
C. macrophylla	8–11	1827	—	—	Reddish purple to purple black
C. mas	8	Ancient	—	—	Scarlet
C. nuttallii	6–24	1835	10	2	Bright red to orange
C. officinalis	6–9	1877	—	—	Red
C. racemosa	4	1758	—	—	White
C. rugosa	3	1784	—	—	Light blue to white
C. sanguinea	2–5	—	—	—	Black
C. sericea	3–6	1656	—	—	White or lead colored
C. sericea ssp. *occidentalis*	5	—	—	—	White

Sources: Dirr (1990), Fernald (1950), Gordon and Rowe (1982), McMinn (1951), Rehder (1940), Weaver (1976).

moist environment followed by a longer period at a much lower temperature is required (table 5). A more complicated procedure has been recommended for cornelian-cherry by Tylkowski (1992). The warm phase of treatment is at alternating temperatures (15/25 °C) on 24-hour cycles for 18 weeks, then a cold phase at 3 °C for 15 to 18 weeks or until the first germination is observed. Immersion in concentrated sulfuric acid for 1 to 4 hours or mechanical scarification can be used in place of warm stratification for most species. Soaking stones in gibberellic acid for 24 hours also has been successful for roughleaf (Furuta 1960) and flowering dogwoods (Litvinenko 1959). In species having only embryo dormancy, this can be broken by low-temperature stratification.

Germination tests. Official testing rules for dogwoods call for germination tests for some species, but rapid tests, such as tetrazolium (TZ) staining or excised embryos, are also recommended (AOSA 1993; ISTA 1993). Flowering and western dogwoods can be tested on the top of moist blotters or creped paper for 28 days at alternating temperatures of 30 °C (day) and 20 °C (night). Excised embryo testing is an alternate method for flowering dogwood, and TZ is an alternate method for western dogwood (AOSA 1993). TZ

Figure 3—*Cornus florida*, flowering dogwood: seedling development at 2, 4, 8, and 31 days after germination.



staining is recommended for the European species Cornelian-cherry and bloodtwig dogwood (ISTA 1993). The seeds must be soaked in water for 48 hours, then cut transversely on the ends and soaked for another 6 hours. The TZ incubation should be for 48 hours in a 1% solution; presence of any unstained tissues is cause to consider the seeds non-viable (ISTA 1993).

Germination tests using 400 properly pretreated seeds per test can be performed using sand, soil, paper, or blotters, but long stratification periods of 3 to 5 months are usually required. The same diurnally alternated temperatures of 30/20 °C appear to be satisfactory for all species (table 6), although Heit (1968a) recommended 30 and 10 °C for silky dogwood. Estimating the viability of dogwood seed lots by TZ staining is the common practice at the USDA Forest Service's National Seed Laboratory and at other seed testing facilities. In many cases, this is the preferred testing method of seed collectors and dealers, nursery managers, and seed testing laboratories. TZ tests performed by trained personnel will provide accurate, reliable data that are comparable to field germination. A TZ test only takes a few days to conduct as compared to a germination test, which requires several months of stratification before the germination period. The quicker TZ test will provide nursery managers with more time to secure different seedlot for either fall-sowing or stratification if the first seedlot is substandard or dead. Excised embryos also have been used (Flemion 1948; Heit 1955).

Nursery practices. Best results for most species are obtained when freshly collected stones are sown in the fall as soon after cleaning as possible (Heit 1968a; Stevenson 1969). Seeds of most species will germinate the following spring. Seeds of species that require a warm-cold pretreatment (table 6) can be planted in the summer but should be left in the ground until the second spring because many will not germinate the spring following planting (Murphy 1997). Dry-stored stones probably should be soaked in water and sown before October (Heit 1968a). Fruits collected too late for fall-sowing should be cleaned, stored over winter and spring, stratified in summer and sown in the fall (NBV 1946; Shumilina 1949). An alternate procedure is to stratify the seeds at about 4 °C for 3 to 4 months during the winter

Table 4—*Cornus,* dogwood: seed yield data

	Stones/fruit wt		Cleaned stones/weight				
			Range		Average		
Species	kg/100 kg	lbs/100 lb	/kg	/lb	/kg	/lb	Samples
C. alba	—	—	27,900–40,900	12,700–18,600	33,000	15,000	33
C. alternifolia	—	—	13,000–20,500	5,900–9,300	17,600	8,000	6
C. amomum	15–18	17–20	22,400–30,800	10,200–14,000	26,800	12,200	6
C. canadensis	—	—	129,800–169,400	59,000–77,000	147,400	67,000	2
C. drummondii	16–24	18–27	18,900–46,200	8,600–21,000	34,500	15,700	5
C. florida	17–41	19–46	7,300–13,600	3,300–6,200	9,900	4,500	11
C. kousa	—	—	14,300–18,300	6,500–8,300	21,300	9,700	3
C. mas	13	15	3,500–7,500	1,600–3,400	5,000	2,300	22
*C. nuttallii**	11	12	8,800–13,400	4,000–6,100	10,300	4,700	4
C. racemosa	16–22	18–25	22,400–33,700	10,200–15,300	28,600	13,000	11
C. rugosa	—	—	—	—	41,800	19,000	1
C. sanguinea	—	—	16,100–26,000	7,300–11,800	20,200	9,200	70
C. sericea	13–18	15–20	30,400–58,700	13,800–26,700	40,700	18,500	9
C. sericea ssp. *occidentalis*	—	—	—	—	73,500	33,400	1

Sources: Asakawa (1969), Brinkman (1974), Edminster (1947), Forbes (1956), Gordon and Rowe (1982), Gorshenin (1941), Heit (1969), Mirov and Kraebel (1939), Mugford (1969), NBV (1946), Stevenson (1969), Swingle (1930).

* 0.036 cubic meters (1 bu) of fruit clusters weighed 15 kg (33 lb) and yielded 2 kg (4 lb) of stones (Brinkman 1974).

and sow them in the spring (Goodwin 1948; Shumilina 1949; Sus 1925). Seeds in nurserybeds should be covered with 6 to 13 mm ($^1/_4$ to $^1/_2$ in) of soil (Brinkman 1974; Heit 1968b; Mugford 1969; NBV 1946; Stevenson 1969). Seeds sown in the fall should be mulched during the winter with 13 to 25 mm ($^1/_2$ to 1 inch) of sawdust (Heit 1968a; Mugford 1969; Stevenson 1969).

Table 5—*Cornus,* dogwood: stratification treatments

	Warm period		Cold period		
Species	**Medium**	**Temp (C°)**	**Days**	**Temp (C°)**	**Duration (days)**
C. alba	—	—	—	5	90–120
C. alternifolia	Sand, peat, or mix	30–20	60	5	60
*C. amomum**	"Moist"	—	—	3–5	21–28
	Sand, peat, or moss	—	—	5	90–120
C. canadensis†	—	—	—	—	60–90
	Sand, peat, or mix	25	30–60	1	120–150
C. controversa	—	—	60–90	—	60–90
C. drummondii‡	Sand	21–27	1	5	30
		—	30–60	—	30–60
C. florida	Sand	—	—	5	120
C. kousa	Sand, peat, or vermiculite	—	—	1–5	40–120
C. macrophylla	—	—	90–150	—	90
C. mas	Soil or vermiculite	20–30	120	1–13	30–120
C. nuttallii§	Peat	—	—	3	90
C. officinalis	—	15–22	120–150	—	90
C. racemosa	Sand	20–30	60	5	60, 120
C. rugosa	Soil	—	—	Outdoors	Overwinter
C. sanguinea	—	—	60	—	60–90
C. sericea //	Sand	—	—	2–5	60–90
	Sand	—	—	5	60-90

Sources: Billington (1943), Brinkman (1974), Dirr and Heuser (1987), Emery (1988), Guan and others (1989), Goodwin (1948), Gordon and Rowe (1982), Heit (1967, 1968b), Jack (1969), Nichols (1934), Ohwi (1965), Pammel and King (1921), Peterson (1953), Soljanik (1961), Swingle (1939),

* Seeds were soaked for 3 hours in water at room temperature before stratification (Heit 1968b).

† Seeds were soaked for 1 hour in sulfuric acid before stratification (Dirr and Heuser 1987).

‡ Seeds were mechanically scarified before stratification (Brinkman 1974).

§ Seeds were soaked for 4 hours in sulfuric acid before stratification (Emery 1988).

// Seeds were soaked for 1 hour in sulfuric acid before stratification (Brinkman 1974).

Table 6—*Cornus,* dogwood: germination test conditions and results

	Germination test conditions*		Germination rate		Germination %		
Species	**Daily light (hrs)**	**Days**	**Amt (%)**	**Days**	**Average (%)**	**Samples**	**Purity (%)**
C. alba	—	—	—	—	—	—	—
C. alternifolia	8	60	8	50	10	2	63
C. amomum	8–24	14–28	86†	11	70	6	91
C. canadensis	—	60–90	6	26	16	5	90
C. drummondii	8	50	14	34	25	3	89
C. florida	8	60	14–45	15–20	35	7	97
C. kousa	—	30	—	—	85	2	—
C. macrophylla	—	—	—	—	—	—	—
C. mas	—	—	—	—	57	6	95
C. nuttallii	8–24	47	57	16	81	2	100
C. racemosa	8	60	22–30	14	20	8	83
C. rugosa	8	60+	8	15	46	4	95
C. sericea	—	60–90	35	13–18	57	18	99

Sources: Adams (1927), Asakawa (1969), Brinkman (1974), Heit (1968a&b, 1969), McKeever (1938), Nichols (1934), Peterson (1953), Soljanik (1961), Swingle (1939), Titus (1940).

*Temperatures were 30 °C for 8 hours and 20 °C for 16 hours each day. Sand was the medium used on all listed species. Additional tests were made on wet paper in germinators with seeds of *C. amomum, C. kousa,* and *C. nuttallii* (Brinkman 1974; Heit 1969).

†One test.

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Betulaceae—Birch family

Corylus L.

hazel

Jill Barbour and Kenneth A. Brinkman

Ms. Barbour is a germination specialist at USDA Forest Service's National Seed Laboratory, Dry Branch, Georgia; Dr. Brinkman retired from the USDA Forest Service's North Central Forest Experiment Station

Other common names. Filbert, hazelnut.

Growth habit, occurrence, and use. The hazels—*Corylus* L.—include about 15 species of large, deciduous shrubs (rarely small trees) that occur in the temperate parts of North America, Europe, and Asia. Some species are grown for their nuts or for ornament, and most species provide food for wildlife. In this country, 4 species have present or potential value for wildlife, shelterbelt, or environmental plantings (table 1). For many years, European hazel has been cultivated for the commercial production of its edible nutmeats, known as hazelnuts or filberts, mostly in Europe but to some extent in the United States, especially in the Willamette Valley of Oregon. Years of first cultivation for other species are as follows: American hazel (1798), beaked hazel (1745), and California hazel (1910).

Flowering and fruiting. Male and female flowers are borne separately on 1-year-old lateral twigs of the same plant. They are formed late in the summer and open the following spring before the leaves appear (table 2). The male flowers are borne in clusters of 2 to 5 pendulous catkins, consisting only of stamens. The female flower is budlike, each flower has a single ovary with 2 styles that are strikingly red at pollination (Hora 1981). By late summer or early fall, the fertilized female flowers develop into fruits. These are round or egg-shaped, brown or dark-tan, hard-shelled "nuts", each containing one embryo that is enclosed in a pericarp, or shell. These nuts are enclosed in an involucre (or husk) which consists of 2 more-or-less united hairy bracts (figures 1 and 2). The seeds are naturally dispersed by animals or birds. Large seedcrops are produced at irregular intervals, usually every 2 or 3 years (NBV 1946; Vines 1960).

Collection of fruits. Hazelnuts may be eaten by rodents, larger animals, or some birds even before they are fully mature. To reduce such losses, fruits should be picked as soon as the edges of the husks begin to turn brown, which may be as early as mid-August.

Extraction and storage of seeds. The fruits should be spread out in thin layers on wire-mesh screens to dry in a room with high humidity for about 1 month. A macerator can be used to separate the nut from the husk. The machine is operated without water, and the nuts and husks pour out of the spout (Horvath 1999). An aspirator or screen cleaning machine is then needed to separate the husk debris from the nut. Alternatively, a brush machine can be used to dehisce the nut in a square-wire cylinder and a vacuum to suck out the dust, with the seeds flowing out the opening in the door (Maloney 1999). Yields and number of seeds per weight vary even within the species (table 3).

Table 1—*Corylus,* hazel: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
C. americana Walt.	**American hazel,** American filbert	Maine to Saskatchewan, S to Georgia; W to Missouri & Oklahoma
C. avellana L.	**European hazel,** European filbert, common filbert	Europe, to 1,824 m in central Alps
C. cornuta **Marsh.** *C. rostrata* Ait.	**beaked hazel,** beaked filbert	Newfoundland to British Columbia, S to Georgia, Missouri, & E Colorado
C. cornuta **var.** ***californica*** **Marsh. (A.DC.) Sharp**	**California hazel,** California filbert	Coast ranges from Santa Cruz N to British Columbia

Source: Brinkman (1974).

Table 2—*Corylus,* hazel: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening
C. americana	—	Mar–May	July–Sept
C. avellana	Europe	Feb–Apr	Sept–Oct
C. cornuta	Tennessee	Jan–Feb	Aug–Sept
var. *california*	California	Jan–Feb	Sept–Oct

Sources: Fernald (1950), Loiseau (1945), Munz and Keck (1959), NBV (1946), Rosendahl (1955), Sus (1925), Van Dersal (1938), Vines (1960), Wappes (1932), Zarger

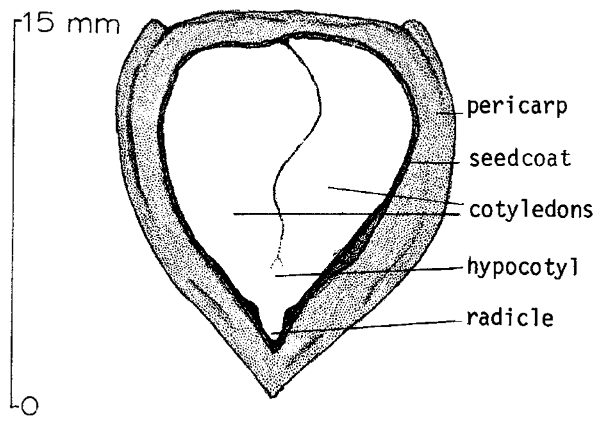
Table 3—*Corylus,* hazel: seed yield data

				Cleaned seeds/weight				
	Place of	Seed wt/fruit wt		Range		Average		
Species	**collection**	**kg/45 kg**	**lb/100 lb**	**/kg**	**/lb**	**/kg**	**/lb**	**Samples**
C. americana	—	11–14	25–30	434–1,623	197–736	1,083	491	11
C. avellana	Europe	27	60	353–1,180	160–535	803	364	244
C. cornuta	—	—	—	937–1,490	425–676	549	249	3
var. *californica*	California	—	—	882–922	400–418	410	186	—

Sources: Brinkman (1974), Gors~~henin (1941), NBV (1946), Rafn~~ (1928), Swingle (1939), Vines (1960), Zarger (1968).

Figure 1—*Corylus cornuta* var. *californica*, California hazel: mature fruit including husk.

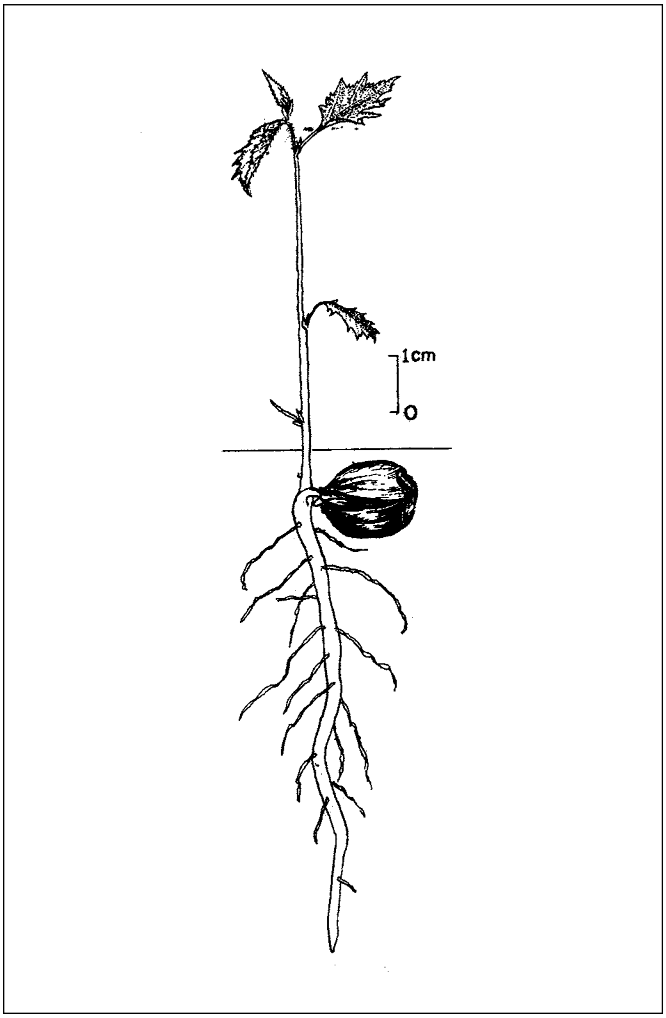
Figure 2—*Corylus cornuta* var. *californica*, California hazel: longitudinal section through a fruit.



Because some dormancy is apparently induced by drying the nuts, seeds of hazel species were once thought to be recalcitrant and intolerant of any drying (Hong and Ellis 1996). Recommendations usually were to keep the hazelnuts moist after collection and store them moist over winter (stratification) before planting in the spring (Heit 1967; NBV 1946). Seeds of hazel species are now considered as orthodox in storage behavior, even though moist storage will prevent deep embryo dormancy for at least several months. Seeds of this genus will also remain viable for a year in unsealed containers at room temperature. Most of the viability of American hazelnut and some of beaked hazelnuts (Brinkman 1974) will be retained if seeds are stored in sealed containers at 5 °C. There are no long-term storage data for hazelnuts.

Pregermination treatments. Newly harvested hazelnuts are not dormant, but inhibitors present in the testa and pericarp are carried to the cotyledons and subsequently through the cotyledonary petioles into the embryonic axis (Bradbeer 1978; Jarvis 1975). Numerous studies have been carried out on the nature of dormancy in European hazel, with most of them concerning the balance of gibberellins and inhibitors and starch synthesis (Arias and others 1976; Bradbeer and Pinnfield 1966; Jarvis 1975; Jarvis and Wilson 1978; Jeavons and Jarvis 1984; Li and Ross 1990). Stratification remains the method used to overcome dormancy, however. Hazel seeds require 2 to 6 months of prechilling before germination will occur (Heit 1968a&b). Three months of cold stratification has proven effective (Dirr and Heuser 1987). Stratification removes the block to gibberellin biosynthesis which begins when the seed is transferred to higher temperatures (Bradbeer and others 1978). In nurseries this can be accomplished by fall-sowing or by stratifying outdoors over winter before planting. Seeds may benefit from alternations of warm and cold stratification. Freshly harvested seeds of European hazel that were warm stratified for 3 weeks followed by 3 weeks at 4 °C germinated best (Dirr and Heuser 1987).

Figure 3—*Corylus cornuta* var. *californica*, California hazel: seedling development 30 days after germination.



Germination tests. Germination is hypogeal (figure 3). The seeds have a dormant embryo and germinate slowly without pretreatment. In one experiment, unstratified seeds of American hazel germinated throughout a year (Brinkman 1974). Giberillic acid (10^{-4} M) applied to European hazel seeds increased the germination from 64% for the control to 86% at 20 °C (Arias and others 1976). Seedlots of this species soaked in ethanol and then 0.1% (w/v) mercuric chloride, when put in a lighted chamber germinated 70% compared to seedlots germinated in total darkness, which germinated at only 9% (Jeavons and Jarvis 1996). Results of limited tests are listed in table 4.

Viability testing by staining the seeds with tetrazolium chloride (TZ) is the preferred method of ascertaining the seed's quality (ISTA 1993). Seeds should be cracked and soaked in water for 18 hours. After 1 to 2 mm of the cotyledons is cut off at the distal ends and the seeds are split longitudinally, the embryos should be incubated for 12 to 15 hours in 1% TZ, or 18 to 24 hours in a 0.5% solution. Some unstained tissue is allowed in viable seeds, but interpretation is difficult. Standard germination tests can also be performed once the pericarp is removed and the seeds are prechilled for 2 months at 3 to 5 °C (ISTA 1993).

Nursery practice. Although spring-sowing of stratified seeds is feasible, most nurseries plant hazel seeds in the fall (Sus 1925). In Holland, seeds of European hazel are mixed with moist sand for several months before sowing in the fall (NBV 1946). In Tennessee, good results with this species were obtained by storing fresh seed dry at 3 °C until planting in October; average tree percent was 98 based on 80% viability (Zarger 1968). Two seedlots of American hazel planted in November and December gave tree percents of 63 and 48, based on 70 and 60% viability. Seeds of both species were sown 2.5 cm (1 in) deep in drills and covered with 2.5 to 3.75 cm (1 to 1.5 in) of sawdust. In this report, the seedbeds had been fumigated with methyl bromide; other fumigants are now recommended. If seedling densities are kept low, from 43 to $65/m^2$ (4 to $6/ft^2$), hazel can be outplanted when 1 year old. European hazel and horticultural varieties are frequently propagated by cuttings, grafting, and tissue culture (Dirr and Heuser 1987).

Hazels are attacked by several fungi. The powdery mildew of hardwoods—*Phyllactinia guttata* (Wallr.:Fr.) Lév. (synonym *Phyllactinia corylea* (Pers.) P. Karst.)—will defoliate the plant. More serious attacks by the fungal parasite *Nematospora coryli* Peglion cause malformation of the nuts (Hora 1981). Hazelnuts are also attacked by the brown rot of pome and stone fruits—*Monilinia fructigena* Honey in Whetzel (synonym *Sclerotinia fructigena* Aderhold. ex Sacc.)—which enters through punctures caused by *Balaninus nuceum*, the nut weevil (Hora 1981).

Table 4—*Corylus*, hazel: germination test conditions and results

	Germination test conditions				Germinative energy		Germinative capacity		
		Temp (°)			Amt		Average		Purity
Species	Medium	Day	Night	Days	(%)	Days	(%)	Samples	(%)
C. americana	Sand	30	20	60	10	30	13	2	96
C. avellana	Sand or germinator	30	20	60	—	—	69	13	95
C. cornuta	Sand	30	20	60	1	26	1	1	99
var. *californica*	Sand	30	20	90	—	—	20	1	62

Sources: Brinkman (1974), NBV (1946), Rafn (1928), Shumilina (1949).

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Anacardiaceae—Sumac family

Cotinus P. Mill.

smoketree or smokebush

Paula M. Pijut

Dr. Pijut is a research plant physiologist at the USDA Forest Service's North Central Research Station, Hardwood Tree Improvement and Regeneration Center, West Lafayette, Indiana

Growth habit, occurrence, and use. The genus *Cotinus* P. Mill—smoketree— includes 3 or 4 species of deciduous, polygamous or dioecious, small trees or shrubs, widely distributed through central and southern Europe to the Himalayas, southwest China, and the southeastern United States (Hillier 1991; Krüssmann 1984). The smoketrees are cultivated primarily for ornamental purposes. The durable wood of American smoketree has been used for fence posts (Koller and Shadow 1991; LHBH 1976) and it also yields a yellow dye that was widely used during the Civil War (Vines 1960). Common smoketree is used in Bulgarian medicine for its anti-inflammatory, antibacterial, and wound-healing properties (Tsankova and others 1993). The 2 species of interest are described in table 1.

Common smoketree is an upright, spreading, multi-stemmed shrub that is grown because of its many ornamental landscape qualities and its adaptability to widely divergent soils and pH ranges (Dirr 1990). Several cultivars produce a long period of midsummer floral and fruit ornamentation, showy plumose inflorescences, and vivid autumn foliage color (Dirr 1990; Hillier 1991; Koller and Shadow 1991; Krüssmann 1984). Of special note are 'Nordine Red', the hardiest of the purple-leaf smokebushes, and 'Royal Purple', a cultivar with rich maroon-red foliage and purplish red inflorescences (Dirr 1990). The foliage of this last cultivar accumulates anthocyanin pigments in response to ultraviolet light of wavelengths between 300 and 400 nm and low temperatures (Oren-Shamir and Levi-Nissim 1997).

American smoketree is a large, upright shrub or small, round-headed tree with bluish to dark green leaves that turn a brilliant yellow, orange, red, and reddish purple color in the fall (Dirr 1990). The bark of the American smoketree is a beautiful gray to gray-brown, and scaly mature trunks (that is, with a fishlike scale effect), providing pattern and detail in the winter landscape (Dirr 1990; Koller and Shadow 1991). For a review of *Cotinus* and discussion of selected cultivars, see Tripp (1994).

Flowering and fruiting. The small, usually infertile, yellowish flowers, which bloom in June to July (April to May for American smoketree), are borne in large, terminal panicles (Krüssmann 1984). The pedicels and peduncles lengthen after flowering and are clad with fine hairs, creating the smokelike effect that gives the plant its common name (LHBH 1976). The plumelike inflorescences often persist through September (Dirr 1990). The fruit (figures 1 and 2) is a dry, reticulate drupe about 3 to 6 mm in length, light red-brown in color (ripening to near black), containing a thick, bony stone (Rudolf 1974). Seedcrops are produced annually but are often poor. The kidney-shaped drupe ripens in the fall, which is usually August to October for common

Table 1—*Cotinus*, smoketree: nomenclature, occurrence, growth habit, height at maturity, and date first cultivated

Scientific name(s)	Common name(s)	Occurrence	Growth habit	Height (m)	Year first cultivated
C. coggygria **Scop.** *C. americanus* Nutt. *C. cotinoides* (Nutt. ex Chapm.) Britt.	**common smoketree**, smokebush, European smoketree, Venetian sumac	Central & S Europe, Himalayas & to SW China	Shrub	2.5–4.6	1656
C. obovatus **Raf.**	**American smoketree**, yellowwood	Tennessee, S to Alabama & Missouri, W to Texas	Tree	6.1–9.1	1882

Sources: Dirr (1990), LHBH (1976).

Figure 1—*Cotinus obovatus*, American smoketree: seeds.

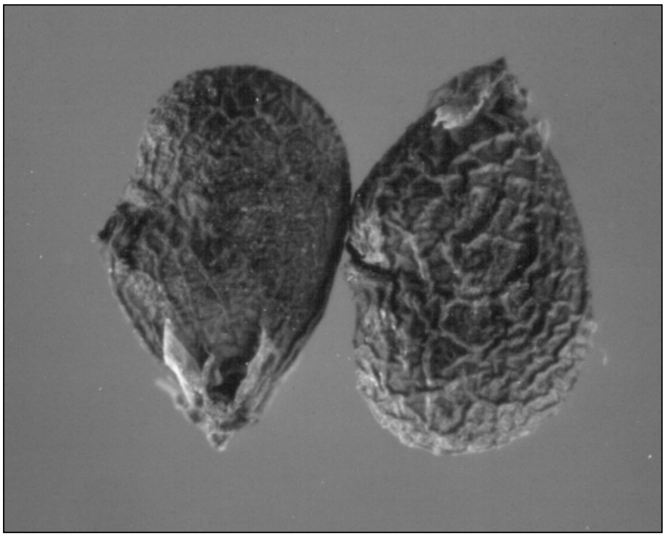
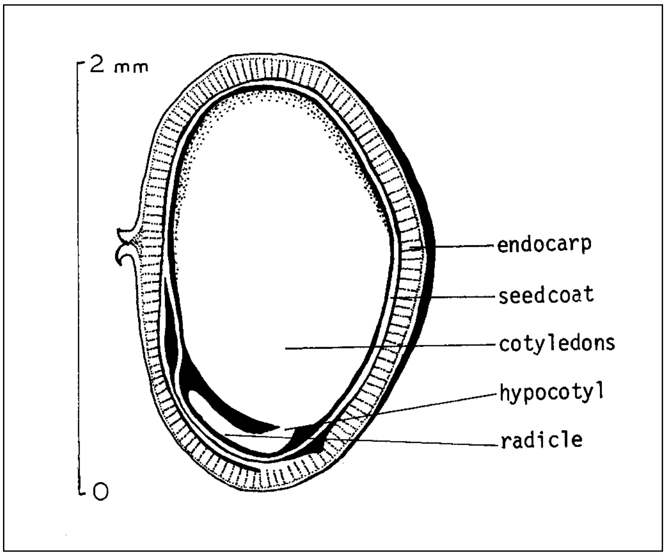


Figure 2—*Cotinus obovatus*, American smoketree: longitudinal section through a seed.



smoketree and June to September for American smoketree (Rudolf 1974).

Collection of fruits; extraction, cleaning, and storage of seeds. The fruits should be harvested by hand as soon as they are ripe (Rudolf 1974). Seeds of common smoketree that are collected green during late August–September and sown immediately can produce high germination percentages the following spring (Dirr and Heuser 1987). Seeds collected from purple-leaf forms produce a mixture of green-leaf and purple-leaf seedlings (Dirr and Heuser 1987). Dry fruits should be run through a hammermill and the debris fanned out (Rudolf 1974). The number of cleaned seeds per seed weight for common smoketree ranges between 99,978 to 118,999/kg (45,350 to 53,978/lb) with 75% germination and 97% purity, depending upon cleaning techniques (Allen 1994). The average number of cleaned seeds per weight for American smoketree is 111,111/kg (50,400/lb) (Rudolf 1974).

Information on smoketree seed storage is limited, but the indications are strong that these seeds are orthodox in storage behavior. One report states that seeds of common smoketree can be stored dry for several years in open or sealed containers at room temperature (Heit 1967, cited by Rudolf 1974). However, the best practice is to store dry seeds in a metal or rigid plastic container that is then sealed and stored in a refrigerator at 0 to 5 °C (Macdonald 1986). Seeds stored in this manner will be viable for a number of years.

Pregermination treatments. Smoketree seeds have both a hard seedcoat and an internal dormancy, thus causing slow and irregular germination. Seeds can be stimulated to germinate more uniformly by sulfuric acid scarification followed by cold stratification (table 2). Seeds from a recent introduction (Dummer hybrids) that were acid-scarified for 3 hours (no cold stratification given) and then planted germinated in 12 days (Dirr 1990).

Germination tests. Pretreated smoketree seeds may be tested for 30 days in sphagnum flats or in seed germinators (Rudolf 1974). Average test results for 2 species are shown in table 3. Tetrazolium staining can be used for rapid estimates. Seeds should be soaked in water for 24 hours before breaking open the seed coat and staining 24 hour at 30 °C in a 1% solution (Enescv 1991).

Nursery practice and seedling care. Smoketree seeds are fall-sown without pretreatment if the fruits are slightly green (Dirr 1990; Macdonald 1986; Rudolf 1974) or with pretreatment in the spring at a rate of 430/m^2 (40/ft^2) (Rudolf 1974). The seed should be covered with 6 to 9 mm ($^1/_4$ to $^3/_8$ in) of soil, and fall-sown beds should be mulched with sawdust (Rudolf 1974). Seedlings may be planted as 1+0 stock (Rudolf 1974).

Several references noted that common smoketree should be propagated by vegetative methods, because many seedlings are male plants lacking the showy flowering panicles (Dirr 1990; Dirr and Heuser 1987; Hartmann and others 1990; Macdonald 1986). In general, softwood cuttings taken in early June to July, treated with 1 to 3 g/liter (1,000 to 3,000 ppm) indole-3-butyric acid solution, and placed in a well-drained medium under mist will root in about 4 to 8 weeks (Blakesley and others 1991, 1992; Dirr 1990; Dirr and Heuser 1987; Hartmann and others 1990; Kelley and

Table 2—*Cotinus,* smoketree: seed pregermination treatments

	Scarification	Stratification treatments		
Species	in H_2SO_4 (min)	Moist medium	Temp (°C)	Days
C. coggygria	30	Sand	3	45–60
	30/60	Sphagnum moss	5	90
	20/80	Peat	3	60–80
C. obovatus	20/40	Plastic bag	3	60

Sources: Dirr and Heuser (1987), Gonderman and O'Rourke (1961), Heit (1968) cited by Rudolf (1974), Stilinovic and Grbic (1988).

Table 3—*Cotinus,* smoketree: germination test conditions and results with pretreated seed

	Germination test conditions				Germination rate		Germination		Sound-
		Temp (°C)							
Species	Medium	Day	Night	Days	%	Days	%	Samples	ness (%)
C. coggygria	Germinator	20	20	30	—	—	80	2	70
	Sphagnum	21	21	21	—	—	93	2	—
C. obovatus	Kimpak in germinator	30*	20	46	37	11	39	3	60†

Source: Rudolf (1974).
* With light for 8 hours.
† Purity was 96%.

Foret 1977; Macdonald 1986; Siftar 1981). Rooted cuttings must be overwintered without disturbance and transplanted in the spring. Spellerberg (1985, 1986) reported improved shoot growth and higher rooting percentages of common smoke tree cv. 'Royal Purple' cuttings when they were taken in April from mother plants forced under glass than cuttings taken in June from outdoor-grown plants. After rooting, shoot growth was promoted by longer photoperiods, higher carbon dioxide levels, and gibberellic acid treatments. Howard (1996) reported that rooting of 'Royal Purple" cuttings was confined to the period of active shoot growth (late May to early August), and a small benefit was noted with severe stock plant pruning. Common smoketree can also be successfully propagated by French or continuous layering (Macdonald 1986).

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Rosaceae—Rose family

Cotoneaster Medik.

cotoneaster

Paul E. Slabaugh and Nancy L. Shaw

Dr. Slabaugh (deceased) retired from the USDA Forest Service's Rocky Mountain Forest and Range Experiment Station; Dr. Shaw is a research botanist at the USDA Forest Service's Rocky Mountain Research Station, Forestry Sciences Laboratory, Boise, Idaho

Growth habit, occurrence, and use. The genus *Cotoneaster* includes about 50 species of shrubs and small trees native to the temperate regions of Europe, northern Africa, and Asia (excepting Japan) (Cumming 1960). Growth habits range from nearly prostrate to upright. Cold-hardy types are more or less deciduous, whereas those native to warmer regions are evergreen (Heriteau 1990). Cotoneasters are valued as ornamentals for their glossy green foliage, attractive fruits, and interesting growth habits. Fall foliage color is often a showy blend of orange and red. Cotoneasters are adapted to sunny locations with moderately deep and moderately well-drained silty to sandy soils. Several hardy species are commonly used in mass plantings, hedges, shelterbelts, wildlife plantings, windbreaks, recreational areas, and along transportation corridors on the northern Great Plains, the southern portions of adjoining Canadian provinces, and occasionally in the Intermountain region and other areas (Plummer and others 1968; Shaw and others 2004; Slabaugh 1974). They require little maintenance and provide ground cover, soil stabilization, snow entrapment, and aesthetic values. Peking cotoneaster provides food and cover for wildlife (Johnson and Anderson 1980; Kufeld and others 1973; Leach 1956; Miller and others 1948). Six species used in conservation plantings are described in table 1 (Hoag 1965; Nonnecke 1954; Plummer and others 1977; Rheder 1940; USDA SCS 1988; Zucker 1966). Use of cotoneasters in some areas may be limited due to their susceptibility to fire blight (infection with the bacterium *Erwinia amylovora*), borers (*Chrysobothris femorata* (*Olivier*)), lace bugs (*Corythucha cydonia* (Fitch)), and red spiders (*Oligonychus platani* (McGregor)(Griffiths 1994; Krüssmann 1986; Wyman 1986).

Cotoneasters are apomictic and will, therefore, propagate true from seed (Wyman 1986). However, because of the apomictic habit, many variants occur within each species (Everett 1982). This variability has been exploited in cultivar

Table 1—*Cotoneaster,* cotoneaster: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
***C. acutifolius* Turcz.** *C. acutifolia* Turcz. *C. pekinensis* Zab.	**Peking cotoneaster**	North China; introduced from North Dakota to Nebraska & upper mid-West, S Canadian prairie provinces
***C. apiculatus* Rehd. & Wilson** *C. apiculata* Rehd. & Wilson	**cranberry cotoneaster**	W China; introduced from North Dakota to Nebraska & upper mid-West
***C. horizontalis* Dcne.** *C. davidiana* Hort.	**rock cotoneaster,** rockspray cotoneaster, quinceberry	W China; introduced from North Dakota to Nebraska & upper mid-West, S central Washington
***C. integerrimus* Medic.** *C. vulgaris* Lindl.	**European cotoneaster**	Europe, W Asia, Siberia
***C. lucidus* Schltdl.** *C. acutifolia* Lindl., not Turcz. *C. sinensis* Hort.	**hedge cotoneaster**	Altai Mtns & Lake Baikal region of Asia
***C. niger* (Thunb.) Fries** *C. melanocarpus* Lodd.	**black cotoneaster,** darkseed cotoneaster	Europe to NE & central Asia, introduced from North Dakota to Nebraska

Source: Krüssmann (1986), LHBH (1976), Slabaugh (1974).

development (Krüssmann 1986; LHBH 1976). A number of hybrids have also been developed as ornamentals.

Flowering and fruiting. Cotoneaster flowers are perfect, regular, and white to pink. They develop singly or several to many together in corymbs produced at the ends of leafy lateral branchlets. Flowers are small, but in some species attractive due to their abundance. Fruits are black or red berrylike pomes that ripen in late summer or early fall and often persist into winter (Wyman 1949) (figure 1). The fruits contain 1 to 5 seeds (Rehder 1940) (figures 2 and 3), averaging 3 for Peking, hedge, and black cotoneasters; 2 for cranberry and rock cotoneasters; and 2 or 3 for European cotoneasters (Uhlinger 1968, 1970). Phenological data are provided in table 2.

Collection of fruits. Ripe fruits are collected by hand stripping or flailing in early autumn, preferably after leaf fall. Fruit firmness and color (table 3) are good criteria of ripeness. Leslie (1954) recommends that fruits of Peking, hedge, and black cotoneasters be collected slightly green. The minimum fruit-bearing age of hedge cotoneaster is 3 years. Fruit crops are produced annually.

Extraction, cleaning, and storage of seeds. Seeds may be extracted by macerating fresh fruits and skimming off or screening out the pulp. Seeds are best cleaned while fresh, because it is difficult to remove dry fleshy material by maceration. Most empty seeds can be eliminated by floating the seedlot twice in water (Uhlinger 1968, 1970). Seeds may be removed from dried fruits by abrasion (Slabaugh 1974) and the debris separated using a 2-screen fanning machine. Number of seeds per weight for 3 species are provided in table 4. About 0.5 kg (1 lb) of cleaned seeds of European cotoneaster are obtained from 2.7 kg (6 lb) of fruits (USDA SCS 1988). Seeds of the cotoneasters are orthodox in storage behavior. Leslie (1954) and USDA SCS (1988) recommend that seeds of cotoneasters be stored dry in sealed containers in a cool place. Seeds of European cotoneaster, however, can be stored in an unheated warehouse for at least 16 years without loss of viability (Jorgensen 1996; Plummer 1968).

Pregermination treatments. Seeds of many cotoneasters exhibit double dormancy due to their hard, impermeable seedcoats and the physiological condition of their embryos. First-year germination is enhanced by acid scarification followed by warm incubation and wet prechilling (USDA SCS 1988) (table 5). Addition of a commercial compost activator to the wet prechilling medium reportedly improved emergence of spreading cotoneaster—*C. divaricatus* Rehd. & Wilson (Cullum and Gordon 1994).

Figure 1—*Cotoneaster*, cotoneaster: fruits.

Figure 2—*Cotoneaster*, cotoneaster: seeds **(from top to bottom)** of *C. apiculanyus,* cranberry cotoneaster; *C. horizontalis,* rock cotoneaster; *C. lucidus,* hedge cotoneaster; *C. niger,* blackcotoneaster.

Figure 3—*Cotoneaster horizontalis*, rock cotoneaster: longitudinal section through a seed.

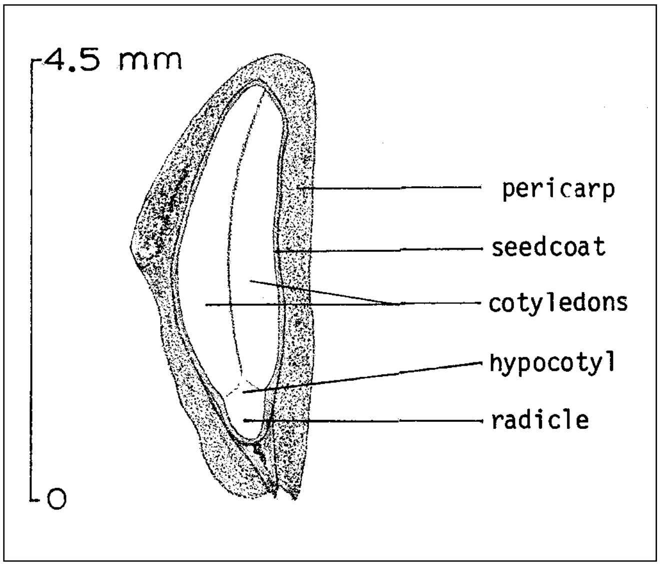

Table 2—*Cotoneaster,* cotoneaster: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening	Seed dispersal
C. acutifolius	N Great Plains	May–June	Sept–Oct	Sept–winter
C. apiculatus	S Michigan	May–June	Aug–Sept	Fall–winter
C. horizontalis	—	June	Sept–Nov	Sept–winter
C. integerrimus	Great Plains	May–June	Aug–Sept	—
C. lucidus	North Dakota	May–June	Sept	—
C. niger	—	May–June	—	—

Sources: Krüssmann (1986), Macdonald (1986), Slabaugh (1974), USDA SCS(1988), Zucker (1966).

Table 3—*Cotoneaster,* cotoneaster: height, year first cultivated, and color of flowers and ripe fruit

Species	Height at maturity (m)	Year first cultivated	Flower color	Color of ripe fruit
C. acutifolius	1.8–3.9	1883	Pink	Black
C. apiculatus	0.3–1.5	1910	Pink	Scarlet
C. horizontalis	0.9–1.2	1880	White-pink	Light to dark red
C. integerrimus	1.2–3.6	—	Pinkish	Red
C. lucidus	1.8–2.7	1840	White, tinged w/pink	Black
C. niger	1.5–2.4	1829	Pinkish-white	Blackish red

Sources: Griffiths (1994), Hoag (1958, 1965), LHBH (1976), Leslie (1954), Krüssmann (1986), Rehder (1940), Rosendahl (1955), USDA SCS (1988).

Table 4—*Cotoneaster,* cotoneaster: seed yield data

	Cleaned seeds/weight			
	Range		Average	
Species	/kg	/lb	/kg	/lb
C. acutifolus	48,466–58,212	21,984–26,405	59,300	26,900
C. horizontalis	—	—	141,094	64,000
C. integerrimus	—	—	35,274	16,000
C. lucidus	—	—	51,560	23,390

Sources: Cumming (1960), McDermand (1969), Plummer and others (1968), Slabaugh (1974), Uhlinger (1968, 1970), USDA SCS (1988).

Table 5—*Cotoneaster,* cotoneaster: pregermination treatments

		Wet prechill at 4 °C	
Species	Immersion time in conc H_2SO_4 (min)	Medium	Period (days)
C. acutifolius	10–90	Peat	30–90
C. apiculatus	60–120	Sand & peat	60–90
C. horizontalis	90–180	Peat	90–120
C. integerrimus	120	—	120*
C. lucidus	5–20	Sand & perlite	30–90
C. niger	10–90	Peat	30–90

Sources: Dirr and Heuser (1987), Fordham (1962), Leslie (1954), McDermand (1969), Slabaugh (1974), Smith (1951), Uhlinger (1968, 1970), USDA SCS (1988).
* Wet prechilling was preceded by 90 days of warm incubation at 21 °C.

Duration of effective pretreatments varies with species, seedlot, and year due to differences in seedcoat thickness and degree of embryo dormancy. Meyer (1988), for example, found that seeds of cranberry and spreading cotoneasters scarified for 1.5 hours in concentrated sulfuric acid germinated over an increasing range of incubation temperatures as the duration of wet prechilling at 2 °C increased from 0 to 4 months. After 4 months of prechilling, germination of both species occurred at constant incubation temperatures from 4.5 to 26.5 °C. This variability in response adds to the difficulty of securing prompt, consistent germination (Uhlinger 1968, 1970).

Germination tests. Table 6 lists germination test conditions and results for 4 cotoneaster species (see table 5 for pretreatments). The effect of light on germination of seeds of Peking, hedge, and black cotoneasters varies among seedlots, but germination of black cotoneaster was generally improved by exposure to cool-white fluorescent light (Uhlinger 1968, 1970). Pretreatment with gibberellic acid partially replaced the effect of light (Uhlinger 1968, 1970).

Because of the dormancy in these seeds, the International Seed Testing Association recommends use of tetrazolium staining rather than germination tests for evaluation of seed quality (ISTA 1993). Seeds are stained by first soaking them in water for 18 hours, then removing the distal third of the seeds with a transverse cut; and finally placing the seeds in a 1.0% solution of tetrazolium chloride for 20 to 24 hours. Viable seeds usually stain completely, but seeds are considered viable if only the radicle tip and the distal third of the cotyledons are unstained (ISTA 1993).

The excised embryo method may also be used to test seed germinability of spreading cotoneaster (Smith 1951). Seeds are first scarified in sulfuric acid for 3 hours, then soaked in 27 °C tapwater for 2 days before the embryos are excised and incubated under conditions favorable for germination.

Nursery practice. Seeds of cotoneaster species may be given appropriate scarification pretreatments and seeded in midsummer to provide the warm incubation and overwinter wet-prechilling required to relieve dormancy and permit germination in the spring. Scarified seeds provided with warm incubation pretreatment in the laboratory may be fall-planted; however, scarification, warm incubation, and wet prechilling in the laboratory are required for spring-planting. A seeding rate of 250 seeds/m^2 (23/ft^2) is recommended for producing lining-out stock of rock cotoneaster (Macdonald 1993); 100 to 130 seeds/m^2 (10 to 12/ft^2) are recommended for European cotoneaster var. 'Centennial' (USDA SCS 1988). Seeds of this variety are planted 0.3 cm (0.1 in) deep and covered with 1.5 to 2 cm (3/5 to 4/5 in) of soil (USDA SCS 1988). European and hedge cotoneaster seedbeds may be mulched with hay or other suitable material (Hinds 1969; USDA SCS 1988). Filtered shade until August is recommended for seedlings of Peking, hedge, and black cotoneasters (Leslie 1954). For hedge cotoneaster, an average seedling yield of 30% was obtained in a North Dakota nursery (Hinds 1969). Seedlings of this species are usually ready for outplanting after 2 growing seasons.

Cotoneasters are propagated vegetatively from softwood and occasionally from hardwood cuttings (Dirr and Heuser 1987; Wyman 1986). Cuttings are taken from June to August (Dirr and Heuser 1987) and treated with 1,000 to 3,000 ppm IBA. Macdonald (1993) recommended that heel cuttings be used when evergreen species are rooted in cold frames. Cuttings, particularly those of evergreen species, root readily and are easily transplanted and overwintered. Layering and grafting are also used to obtain small numbers of plants.

Field planting. Nursery stock is generally used to establish conservation plantings. Wildland seedings of Peking cotoneaster have been only marginally successful

Table 6—*Cotoneaster,* cotoneaster: germination test conditions and results

	Germination test conditions				Percentage germination		
			Temp (°C)				
Species	Daily light (hrs)	Medium	Day	Night	Days	Avg (%)	Samples #
C. acutifolius	9	Wet paper	25	10	—	70–80	—
C. horizontalis	24	Wet paper	27	—	—	100	—
	24	Sand	30	20	100	30	5+
C. lucidus	9	Wet paper	25	10	—	70	—
C. niger	9	Wet paper	25	10	—	80	—

Sources: Smith (1951), Slabaugh (1954), Uhlinger (1968 & 1970).

(Shaw and others 2004). Germination is erratic and seedlings grow slowly, particularly if the site is not kept weed-free.

Bareroot plantings of European cotoneaster Centennial may be established using 1+0 or 2+0 bareroot seedlings with stem diameters of 0.5 to 1.3 cm ($^1/_5$ to $^1/_3$ in) just above the root collar (USDA SCS 1988). Seedlings should be planted in fallowed ground at 1.2- to 1.5-m (4- to 5-ft) spacings immediately after the soil thaws in spring. At least 5 years of weed control are often required. Average survival ranges from 70 to 95% (USDA SCS 1988). Fruit-producing stands are obtained in 3 to 4 years.

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Rosaceae—Rose family

Purshia DC. ex Poir.

bitterbrush, cliffrose

D. Terrance Booth, Susan E. Meyer, and Nancy L. Shaw

Dr. Booth is a rangeland scientist at the USDA Agricultural Research Service's High Plains Grasslands Research Station, Cheyenne, Wyoming; Dr. Meyer is a research ecologist at the USDA Forest Service's Rocky Mountain Research Station, Shrub Sciences Laboratory, Provo, Utah; Dr. Shaw is a research botanist at the USDA Forest Service's Rocky Mountain Research Station, Forestry Sciences Laboratory, Boise, Idaho

Growth habit, occurrence, and use. The bitterbrush genus—*Purshia*—as presently circumscribed comprises 8 species of decumbent to arborescent shrubs of interior western North America. Three are common in the United States (table 1). The type species—antelope bitterbrush—has an essentially northern distribution, whereas cliffrose has an essentially southern distribution, and desert bitterbrush occurs in parts of the geographic area where the other 2 species have overlapping distributions. Cliffrose, along with the 5 Mexican species of the genus, has been traditionally referred to the genus *Cowania* D. Don. Cliffrose regularly forms hybrids with antelope bitterbrush, and desert bitterbrush could be interpreted as a stabilized hybrid between these species (Stutz and Thomas 1964). In fact, molecular genetics work by Jabbes (2000) indicates that *Purshia* was derived from *Cowania*. We follow Welsh and others (1987) in treating the group as congeneric under the name *Purshia*.

Members of the genus are erect, spreading or decumbent, freely branched shrubs up to 6 m in height. They have small, alternate, simple, apically lobed leaves that may be evergreen (cliffrose) to winter deciduous (antelope bitterbrush). Layering forms of bitterbrush (principally antelope bitterbrush) may resprout after fire, but erect forms are usually not fire tolerant. Because of their interesting habits, attractive foliage, and showy flowers, bitterbrush species have potential as ornamentals in low-maintenance landscapes.

Bitterbrush species are hardy and drought tolerant. Antelope bitterbrush occurs mainly on well-drained soils over a wide elevational range and is often a principal component of mixed shrub, pinyon–juniper, ponderosa pine, and sometimes lodgepole pine communities, where it is notable as a nurse plant for conifer seedlings (Geier-Hayes 1987; McArthur and others 1983; Nord 1965; Tew 1983). It is valued as a high-protein browse for domestic and wild ungulates, being especially important on winter ranges (Bishop and others 2001; Scholten 1983). It also supplies high-quality forage during spring and summer months (Austin and Urness 1983; Ngugi and others 1992). Cliffrose grows primarily on rocky sites in blackbrush–joshua tree woodland, sagebrush–grassland, piñyon–juniper woodland, mountain brush, and ponderosa pine communities, sometimes forming extensive stands on south-facing ridge slopes (McArthur and others 1983). It is also an important browse species, especially for mule deer (*Odocoileus hemionus*) (Plummer and others 1968). Desert bitterbrush is a component of blackbrush, chaparral, and piñyon–juniper communities.

The bitterbrush species form actinorhizal root nodules that fix nitrogen when soil water is adequate (Bond 1976;

Table 1—*Purshia,* bitterbrush, cliffrose: common names and geographic distributions

Scientific name & synonym(s)	Common name	Geographic distribution
P. glandulosa **Curran** *P. tridentata* var. *glandulosa* (Curran) M.E. Jones	**desert bitterbrush**	SW Utah, S Nevada, & S California
P. mexicana **(D. Don) Henrickson** *Cowania mexicana* D. Don	**cliffrose**	S Colorado W through Utah to S California & S to New Mexico, Arizona, Sonora, & Chihuahua
P. tridentata **(Pursh) DC.**	**antelope bitterbrush**	British Columbia to W Montana, S to New Mexico, California, & N Arizona

Sources: Little (1979), Sargent (1965), Vines (1960).

Kyle and Righetti 1996; Nelson 1983; Righetti and others 1983). They readily function as pioneer species that colonize harsh, steep disturbances and have been used extensively in revegetation and disturbed-land reclamation. An ethanol extract of antelope bitterbrush aerial stems was found to inhibit reverse transcriptase of HIV-1 and to contain the cyanoglucosides pushianin and menisdaurin (Nakanishi and others 1994). Unfortunately, the cyanoglucosides lacked the inhibitory activity of the original extract. Cliffrose has also been examined for beneficial secondary products (Hideyuki and others 1995; Ito and others 1999). Specific populations of antelope bitterbrush with distinctive attributes are recognized and are commercially harvested and sold, although to date only two ('Lassen' and 'Maybell') have been formally named (Davis and others 2002; Shaw and Monsen 1995).

Flowering and fruiting. Most of the medium to large, perfect, cream to sulfur yellow flowers of this genus appear during the first flush of flowering in April, May, or June, depending on elevation. In areas where they co-occur, antelope bitterbrush usually flowers 2 to 3 weeks before cliffrose. The flowers are borne on lateral spurs of the previous year's wood (Shaw and Monsen 1983). In cliffrose, summer rains may induce later flowering on current-year leaders, but these flowers rarely set good seeds (Alexander and others 1974). The flowers have a sweet fragrance and are primarily insect-pollinated. Each has 5 sepals, 5 separate petals, numerous stamens, and 1 to 10 pistils borne within a hypanthium. Flowers of antelope and desert bitterbrushes usually contain a single pistil with a relatively short, non-plumose style, whereas those of cliffrose contain multiple pistils. The pistils develop into single-seeded achenes with papery pericarps. In cliffrose the achenes are tipped with persistent) plumose styles, 22 to 50 mm (1 to 2 in) in length, that give the plants a feathery appearance in fruit.

The main fruit crop ripens from June through August, depending on species and elevation. Plants begin to bear seeds as early as 5 years of age. At least some fruits are produced in most years, and abundant seedcrops are produced on average every 2 to 3 years (Alexander and others 1974; Deitschman and others 1974). Cliffrose seeds (figure 1) are apparently dispersed principally by wind (Alexander and others 1974). Scatter-hoarding rodents such as chipmunks (*Tamias* spp.), disperse bitterbrush seeds (figure 2) and seedlings from rodent caches appear to account for nearly all (99%) natural recruitment as survivors from seedling clumps containing 2 to >100 individuals (Evans and others 1983; Vander Wall 1994).

Seed collection, cleaning, and storage. Bitterbrush plants produce more leader growth in favorable water years, and leader length is an indicator of the potential for seed production the following year (McCarty and Price 1942; Young and Young 1986). Fruits may be hand-stripped or beaten into hoppers or other containers when fully ripe; harvesters should take care to protect themselves from the fiberglass-like style hairs in the case of cliffrose. The window of opportunity is quite narrow, as ripe fruits are easily detached by wind and do not persist long on the plant, making close monitoring during ripening advisable. Plants in draws and other areas protected from wind may retain their seeds longer. Maturation dates for antelope bitterbrush have been predicted with reasonable accuracy using elevational and latitudinal predictors (Nord 1965). Well-timed harvests of antelope bitterbrush average 168 to 224 kg/ha (150 to 200 lb/acre) but may range up to 560 kg/ha (500 lb/acre) (Nord 1965). Fill percentages are usually high, although insects or drought stress during filling can damage the crop (Shaw and Monsen 1983). Krannitz (1997a) reported the variation in seed weight from 240 bitterbrush plants representing 10 sites in the southern Okanagan Valley of Canada varied from

Figure 1—*Purshia*, mexicana, cliffrose: achenes:

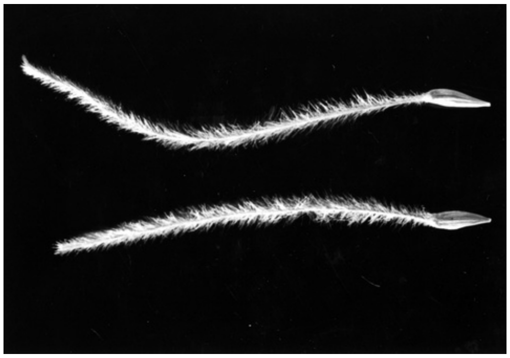
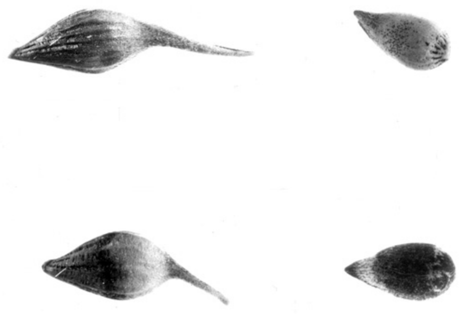


Figure 2—*Purshia*, bitterbrush: achenes (**left**) and cleaned seeds (**right**) of *P. glandulosa*, desert bitterbrush (**top**) and



5 to 46 mg/seed with the population being skewed toward the small seeds. The representative weights given in table 2 are of cleaned seeds (the smaller fraction is removed in cleaning). Krannitz also found that larger seeds had greater concentrations of nitrogen than smaller seeds and that shrubs that had been browsed most intensively the winter before seed-set had seeds with greater concentrations of magnesium (Krannitz 1997b).

A seed cleaner or barley de-bearder may be used to break the styles from cliffrose achenes and to remove the papery pericarps of bitterbrush species. The achenes (cliffrose) or seeds (bitterbrush species) may be separated from the inert material—which usually comprises from one-third (antelope bitterbrush) to two-thirds (cliffrose) of the total weight—using a fanning mill (Alexander and others 1974; Giunta and others 1978). In cliffrose, the achene is considered the seed unit, as the seed is held tightly within the pericarp and cannot be threshed out without damage. In bitterbrush species, the seeds are easily threshed free of their papery pericarps, and the seed unit is the seed itself. If properly dried (<10% moisture content), seeds of bitterbrush species can be warehouse-stored for 5 to 7 years (Belcher 1985) or even up to 15 years without losing viability (Stevens and others 1981).

Germination and seed testing. Bitterbrush and cliffrose seeds are mostly dormant but the inhibiting mechanism(s) is not understood (Booth 1999; Booth and Sowa 2001; Dreyer and Trousdale 1978; Meyer 1989; Meyer and Monsen 1989; Young and Evans 1976, 1981). Moist chilling is preferred for breaking dormancy (table 3). Although some collections are less dormant than others are—as indicated by germination percentages for untreated or partially treated seeds (table 3) (Booth 1999; Meyer and Monsen 1989)—there is no obvious relationship between collection site and chilling requirement (Meyer and Monsen 1989). Dormancy might be affected by high seed temperature (30 °C) while in the dry state (Meyer 1989) and is certainly affected by imbibition temperature (Booth 1999; Meyer 1989).

Young and Evans (1981) reported the required chilling period was shorter at 5 °C, than at 2 °C for all 3 species, and that adequately chilled seeds could germinate over a wide range of temperatures. A 28- to 30-day chill at 1 to 3 °C is highly recommended (AOSA 1993; Belcher 1985; Booth 1999; Meyer 1989) followed by post-chill incubation at 15 °C (10/20 °C for cliffrose). Desert bitterbrush needs only 14 days of chilling (Belcher 1985). Germination of antelope bitterbrush seeds can be facilitated by 24 hours of soaking in cold (2 °C) water prior to moist chilling, but soaking in

Table 2—*Purshia,* bitterbrush and cliffrose: seed yield data (seeds/weight) for mechanically cleaned seeds*

	Mean		Range	
Species	**/kg**	**/lb**	**/kg**	**/lb**
P. glandulosa	50,850	26,540	45,000–90,000	20,300–40,900
P. mexicana	129,000	58,600	108,000–210,000	49,000–95,000
P. tridentata	35,000	15,750	29,000–51,000	13,400–23,200

Sources: Alexander and others (1974), Belcher (1985), Deitschman and others (1974), Meyer (2002), Meyer and others (1988).

Table 3—*Purshia,* bitterbrush and cliffrose: germination data

	Mean percentage of initially viable seeds							
Species	**0**	**2 wk**	**4 wk**	**6 wk**	**8 wk**	**10 wk**	**12 wk**	**Samples**
P. glandulosa	—	—	—	93	—	—	100	1*
	10	56	81	100	65	—	32	1†
P. mexicana	6	33	83	94	100	—	—	6
	6	64	91	100	32	—	19	1†
P. tridentata	2	43	88	98	100	—	—	13
	13	60	100	100	36	—	37	1†

Sources: Deitschman and others (1974), Meyer (2002), Meyer and Monsen (1989), Young and Evans (1981).

Note: Values are expressed as percentage of initially viable seeds after moist chilling at to 2 °C for 0 to 12 weeks followed by incubation at 15 °C or 10/20 °C for 4 weeks.

* These seeds were chilled at 3 to 5 °C and germination was scored during chilling.

† Decrease in germination percentage after 6 weeks was due to seed mortality during the test.

warm water (>10 °C), or holding imbibed seeds at warm temperatures, decreases seedling vigor and increases pre-germination seed-weight loss (Booth 1999; Booth and Sowa 2001). Longer, colder chilling periods (28 days, 2 °C vs 14 days, 5 °C) increases seedling vigor (Booth 1999; Booth and Morgan 1993). Recommended germination test periods are 28 days for antelope bitterbrush and cliffrose (AOSA 1993).

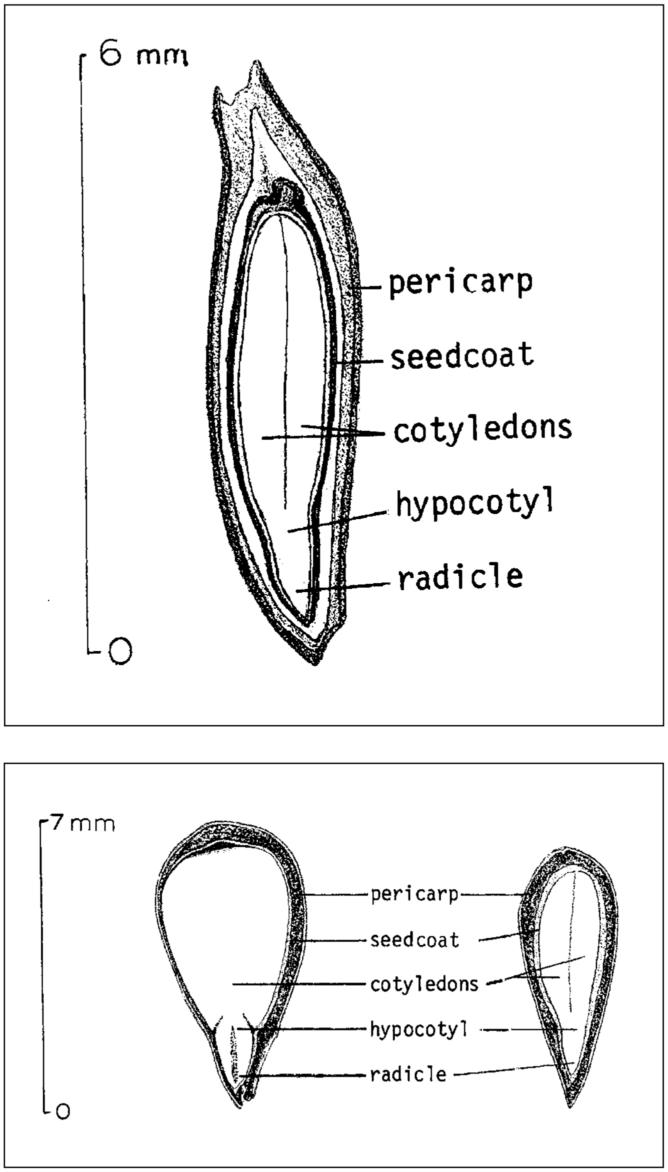
Soaking seeds in hydrogen peroxide (Everett and Meeuwig 1975) or a 1 to 3% solution of thiourea (Pearson 1957; Young and Evans 1981) will induce germination but these methods have not proven useful for field plantings. Booth (1999) found thiourea-treated seeds to have the lowest seedling vigor among 8 dormancy-breaking treatments and attributed the lower vigor to residual dormancy and to weight loss resulting from accelerated respiration (Booth 1999; Booth and Sowa 2001).

Tetrazolium (TZ) staining is acceptable for evaluating seed quality of bitterbrush (AOSA 1993; Weber and Weisner 1980). Meyer (2002) found no significant difference between TZ viability estimates and germination perentages after 8 weeks of chilling for either cliffrose or antelope bitterbrush. For TZ viability testing, seeds should be clipped at the cotyledon end (figure 3) and soaked in water for 6 to 24 hours. Then, the embryos can be popped-out of the cut end by gentle finger pressure and immersed in 1% TZ solution for 4 to 12 hours at room temperature before evaluation. Cliffrose must be soaked longer than bitterbrush before the embryos can be popped out.

Field seeding and nursery practice. Bitterbrush species are generally sown in fall or early winter in a mixture with other shrubs and forbs. They are used in upper sagebrush, piñyon-juniper woodlands, and mountain brush vegetation types to improve degraded wildlife habitat or revegetate bare roadcuts, gullies, south slopes, and other difficult sites (Alexander and others 1974). Because of the chilling requirement, spring-seeding should be avoided. Seeds may be drilled at a depth of 6 to 12 mm ($^{1}/_{4}$ to $^{1}/_{2}$ in) or deeper. Deeper seeding may provide some protection from rodent depredation, which can be a serious problem (Alexander and others 1974; Evans and others 1983; Vander Wall 1994). Seeding in late fall or early winter, when rodents are less active, may also alleviate this problem.

Broadcast-seeding is generally unsuccessful unless provision is made for covering the seeds. Aerial seeding is not recommended. The seedlings do not compete well with weedy annual grasses such as red brome (*Bromus rubens* L.) and cheatgrass (*B. tectorum* L.), or with heavy stands of perennial grasses. They are sensitive to frost and drought during establishment (Plummer and others 1968). Recommended (drill) seeding rates for cliffrose are 5 to 10% of the shrub mix at 8 to 10 kg/ha (7 to 9 lb/ac) (Alexander and others 1974; Plummer and others 1968) and 16 to 65 seeds/m (5 to 20 seeds/ft) for bitterbrush. The higher rates are advisable for both species when seeding in crust-forming soils. The most effective method of seeding large areas in conjunction with chaining is with a seed dribbler that drops seeds in front of the bulldozers pulling the chain.

Figure 3—*Purshia*: longitudinal section of *P. mexicana*, cliffrose (**top**) and *P. tridentata*, antelope bitterbrush (**bottom**).



Hand-planting into scalped sites with a tool such as a cased-hole punch planter can be very effective on a small scale (Booth 1995). The purpose of scalping is to control herbaceous competition within a half-meter (1 $^{1}/_{2}$-ft) radius of the planting spots. Treating seeds with fungicide, planting seeds in groups, and planting with vermiculite to aid in moisture retention have all improved emergence and establishment of antelope bitterbrush (Booth 1980; Evans and others 1983; Ferguson and Basile 1967). Good emergence depends on adequate snowcover (Young and others 1993).

Bitterbrush species are readily grown as bareroot or container stock, and outplanting may succeed where direct seeding has failed (Alexander and others 1974). Care must be taken to lift or transplant stock only when the plants are hardened or dormant, as survival of actively growing plants is generally low (Landis and Simonich 1984; Shaw 1984). Plants are easier to handle and have higher survival rates if allowed to reach sufficient size before field transplanting. One-year-old bareroot stock or container seedlings 16 to 20 weeks of age are usually large enough (Alexander and others 1974; Shaw 1984). On more level terrain, a conventional tree-planter may be used (Alexander and others 1974). Transplanting should be carried out at a time and in such a way as to assure that the transplants will have adequate moisture for root development for 4 to 6 weeks after planting. This may be accomplished by planting in very early spring or by watering at the time of planting. Fall-planted seedlings may require supplemental watering. Controlling competition from weedy annual or perennial grasses before planting will enhance survival and first-season growth.

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Rosaceae—Rose family

Crataegus L.

hawthorn, haw, thorn, thorn-apple

F. Todd Lasseigne and Frank A. Blazich

Dr. Lasseigne is executive director of the Paul J. Ciener Botanical Garden,
Kernersville, North Carolina; Dr. Blazich is alumni distinguished graduate professor
of plant propagation and tissue culture at North Carolina State University's
Department of Horticultural Science, Raleigh, North Carolina

Growth habit, occurrence, and uses. The genus *Crataegus* L. is a complex group of trees and shrubs native to northern temperate zones (Mabberley 1997), mostly between latitudes 30° and 50°N (Phipps 1983). Although most species can attain tree-sized proportions, hawthorns in general do not form large trees or exist as canopy dominants in forests (Little 1980a&b). Some species are decidedly shrubby, whereas others can grow to heights of 12 m (table 1). There are about 250 currently recognized species, with most native to the New World (about 200 species), and the remainder (about 50 species) native to the Old World (Christensen 1992; Phipps and others 1990). Species native to the United States, as well as those that have been introduced and naturalized and some of those grown horticulturally, are included herein (table 1).

Historically, the taxonomy of the hawthorn genus has been rife with disagreement and confusion. The circumscriptions of species have varied widely, and authors of various floristic treatments have misidentified species that occur in regions treated in their works (Phipps 1998c). The genus has vexed so many authors that early experts on the group termed the situation "the Crataegus problem" (Eggleston 1910; Palmer 1932). Nearly 1,500 "species" were described in North America alone, mostly by W. W. Ashe, C. D. Beadle, and C. S. Sargent, from the 1890s through the 1910s (Christensen 1992; Phipps 1988; Phipps and others 1990; Robertson 1974). Palmer later reduced the number of species of hawthorns, such that only 20 to 100 were recognized, a range followed by subsequent authors (Phipps 1988). Recently, taxonomists have taken a middle approach, recognizing 100 to 200 species in North America (Kartesz 1994a&b; Phipps and others 1990), a larger number than that accepted in treatments of 20 to 30 years ago. Two primary references—Kartesz (1994a) and Phipps and others (1990)—offer the most complete survey of North American hawthorns (excluding Mexico).

Crataegus belongs to the subfamily Maloideae in the Rosaceae, a natural group of complex genera with the ability to interbreed freely (or hybridize), as they all possess the basal chromosome number of 17 (Phipps and others 1991; Robertson 1974; Robertson and others 1991). Authors have long regarded hybridization and apomixis as potential explanatory factors for the speciation phenomenon existing in hawthorns (Phipps 1988; Radford and others 1968; Vines 1960). Robertson (1974) related empirically derived data that implicated apomixis and hybridization as causes of the variation found within the genus. Specifically, he cited (1) widespread occurrence of pollen sterility; (2) cytological proof of triploidy or polyploidy in > 75% of plants observed; (3) similarity between offspring produced from triploid or pollen-sterile plants and parental plants; and (4) the ability of flowers that have stigmas removed at anthesis to set fruit.

Many authors allude to the existence of putative hybrids in New World hawthorns (Elias 1987; Harlow and others 1996; Jacobson 1996; Kartesz 1994a&b; Knees and Warwick 1995; LHBH 1976; Little 1980a&b; Phipps 1984; Vines 1960). However, despite widespread documentation of hybrid species complexes existing in Eurasia (Christensen 1992), few scientifically verified examples of hybrid species in North American hawthorns are known (Phipps 1998a). Several recent studies now demonstrate unequivocal proof that both apomixis and polyploidy are implicated in the complex variation seen in this genus in North America (Dickinson 1985; Muniyamma and Phipps 1979a&b, 1984, 1985; Phipps 1984). Apomixis and hybridization are also known in other Rosaceous genera, including *Alchemilla* L. (lady's-mantle), *Cotoneaster* Medik. (cotoneaster), *Potentilla* L. (cinquefoil), and *Rubus* L. (blackberries and raspberries) (Mabberley 1997).

Around the world, hawthorns are used for a wide range of purposes. Many hawthorn species are grown for their

Table 1—*Crataegus,* hawthorn: nomenclature, occurrence, and heights at maturity

Scientific name & synonym(s)	Common name(s)	Occurrence	Height at maturity (m)
***C. aestivalis* (Walt.) Torr. & Gray** *C. cerasoides* Sarg. *C. luculenta* Sarg.; *C. maloides* Sarg.	**eastern mayhaw**, shining, may, or apple hawthorn	N Florida & SE Alabama, N to E North Carolina	3–12
***C. x anomala* Sarg. (pro sp.)** *C. arnoldiana Sarg.*	**Arnold hawthorn**, anomalous hawthorn	Quebec & New England, S to New York	5–10
***C. berberifolia* Torr. & Gray**	**barberry hawthorn,** bigtree hawthorn	Virginia to Kansas, S to Georgia & Texas	5–11
***C. brachyacantha* Sarg. & Engelm.**	**blueberry hawthorn**, blue haw, pomette bleu	Arkansas to Oklahoma, S to Mississippi & Texas; Georgia also	6–15
***C. brainerdii* Sarg.**	**Brainerd hawthorn**	Quebec to Michigan, S to New England, North Carolina & Ohio	2–7
***C. calpodendron* (Ehrh.) Medik.** *C. calpodendron* var. hispidula (Sarg.) Palmer *C. fontanesiana* (Spach) Steud. *C. hispidula* Sarg.; *C. tomentosa* L.	**pear hawthorn,** sugar or black hawthorn	Ontario to Minnesota & Kansas, S to Georgia & Texas	4–6
C. chrysocarpa* Ashe var. *chrysocarpa *C. brunetiana* Sarg. *C. doddsii* Ramalay; *C. faxonii* Sarg. *C. praecoqua* Sarg.; *C. praecox* Sarg. *C. rotundifolia* Moench; *C. sheridana* A. Nelson	**fireberry hawthorn,** roundleaf or golden-fruit hawthorn	Newfoundland to British Columbia, S to North Carolina & New Mexico	5–10
***C. coccinoides* Ashe**	**Kansas hawthorn,** Eggert thorn	Indiana to Kansas, S to Arkansas & Oklahoma	4–7
***C. crus-galli* L.** *C. acutifolia* Sarg.; *C. bushii* Sarg. *C. canbyi* Sarg.; *C. cherokeensis* Sarg. *C. mohrii* Beadle; *C.operata* Ashe; *C.palmeri* Sarg. *C. regalis* Beadle; *C. sabineana* Ashe *C. salicifolia Medik.* *C. signata* Beadle *C. subpilosa* Sarg.; *C. vallicola* Sarg. *C. warneri* Sarg.	**cockspur hawthorn,** Newcastle thorn, hog-apple	Quebec to Michigan & Kansas, S to Florida & Texas	5–10
***C. dilatata* Sarg.** *C. conspecta* Sarg. *C. locuples* Sarg.	**broadleaf hawthorn,** apple-leaf hawthorn	Quebec to Michigan, S to New York, Kentucky, & Missouri	4–8
***C. douglasii* Lindl.** *C. columbiana* Howell	**black hawthorn,** Douglas or western black hawthorn, black thornberry	Alaska to S California, Ontario to Dakotas, S to Michigan & Nevada	7–12
***C. erythropoda* Ashe** *C. cerronis* A. Nelson	**cerro,** chocolate hawthorn	Wyoming to Washington, S to New Mexico & Arizona	2–6
***C. flabellata* (Spach) Kirchn.** *C. densiflora* Sarg.; *C. grayana* Egglest.	**fanleaf hawthorn**	Maine to Quebec to Michigan, S to Florida & Louisiana	4–6
***C. flava* Ait.** *C. cullasagensis* Ashe	**yellow hawthorn** summer haw	Maryland & West Virginia, S to Florida & Mississippi	5–8
***C. greggiana* Egglest.**	**Gregg hawthorn**	Texas & NE Mexico	3–6
***C. harbisonii* Beadle**	**Harbison hawthorn**	Tennessee, S to Georgia & Alabama	3–8
***C. intricata* Lange**	**thicket hawthorn,** entangled or Allegheny hawthorn	New England to Michigan to Missouri, S to Florida & Alabama	1–7
***C. lacrimata* Small**	**Pensacola hawthorn,** weeping or sandhill hawthorn	Florida	3–6
***C. laevigata* (Poir.) DC.** *C. oxyacantha* L., in part *C. oxycanthoides* Thuill.	**English hawthorn,** English midland or English woodland hawthorn	Central & W Europe	2–4
***C. marshallii* Egglest.** *C. apiifolia* (Marshs.) Michaux	**parsley hawthorn,** parsley haw	Virginia to Illinois, S to Florida & Texas	2–8
***C. mollis* (Torr. & Gray) Scheele** *C. albicans* Ashe *C. arkansana* Sarg. *C. brachyphylla* Beadle *C. cibaria* Beadle *C. coccinea* var. *mollis* Torr. & Gray *C. invisa* Sarg.; *C. lacera* Sarg.; *C. limaria* Sarg.	**downy hawthorn,** summer hawthorn red haw, turkey-apple	Ontario to the Dakotas, S to Alabama & Texas	6–12
***C. monogyna* Jac.** *C. oxyacantha* L. ssp. *monogyna* (Jacq.) Rouy & Camus	**oneseed hawthorn,** single-seed or common hawthorn, may, quickthorn	Europe, N Africa, & W Asia	5–12

Table 1—*Crataegus,* hawthorn: nomenclature, occurrence, and heights at maturity (continued)

Scientific name & synonym(s)	Common name(s)	Occurrence	Height at maturity (m)
C. nitida (Engelm.) Sarg. *C. viridis* var. *nitida* Engelm.	**shining hawthorn,** glossy hawthorn, & shining thorn	Ohio to Illinois, S to Arkansas	7–12
C. opaca Hook. & Arn. *C. nudiflora* Nutt. ex Torr. & Gray	**western mayhaw**, apple haw, may, or riverflat hawthorn	W Florida to Texas, N to Arkansas	6–10
C. pedicellata Sarg. var. _pedicellata_ *C. aulica* Sarg.; *C. caesa* Ashe *C. coccinea* L. in part	**scarlet hawthorn,** Ontario hawthorn	Maine to Michigan, S to Virginia & Illinois; South Carolina & Florida also	4–8
C. persimilis Sarg. *C. laetifica* Sarg.; *C. prunifolia* Pers.	**plumleaf hawthorn**	New York to Ontario, S to Pennsylvania & Ohio	7–10
C. phaenopyrum (L. f.) Medik. *C. cordata* (Mill.) Ait. *C. populifolia* Walt. *C. youngii* Sarg.	**Washington hawthorn**, Virginia hawthorn, Washington thorn, hedge thorn, red haw	New Jersey to Missouri, S to Florida, Mississippi & Louisiana	4–10
C. piperi Britt. *C. chrysocarpa* Ashe var. *piperi* (Britt.) Krushke *C. columbiana* auct. *C. columbiana* var. *columbiana* T.J. Howell *C. columbiana* Howell var. *piperi* (Britt.) Egglest.	**Columbia hawthorn,** Piper hawthorn	British Columbia, S to Idaho & Oregon	4–6
C. pruinosa (Wendl. f.) K. Koch *C. formosa* Sarg. *C. georgiana* Sarg. *C. lecta* Sarg.; *C. mackenzii* Sarg. *C. leiophylla* Sarg.; *C. porteri* Britt. *C. rugosa* (Ashe) Kruschke; *C. virella* Ashe	**frosted hawthorn,** waxy-fruited hawthorn	Newfoundland to Wisconsin, S to West Virginia & Oklahoma	2–8
C. pulcherrima Ashe *C. flava* Ait., not auctt. *C. opima* Beadle; *C. robur* Beadle	**beautiful hawthorn**	Florida to Mississippi	4–8
C. punctata Jacq. *C. fastosa* Sarg. *C. punctata* var. *aurea* Ait. *C. verruculosa* Sarg.	**dotted hawthorn,** flat-topped, thicket, or large-fruited hawthorn	Quebec to Minnesota & Iowa, S to Georgia & Arkansas	5–10
C. reverchonii Sarg.	**Reverchon hawthorn**	Missouri to Kansas, S to Arkansas & Texas	1–8
C. rufula Sarg.	**rufous mayhaw**	N Florida, SW Georgia, & SE Alabama	3–9
C. saligna Greene	**willow hawthorn**	Colorado	4–6
C. sanguinea Pall.	**Siberian hawthorn**	E Russia & Siberia, S to Mongolia & China	5–8
C. spathulata Michx. *C. microcarpa* Lindl.	**littlehip hawthorn,** small-fruited or pasture hawthorn	Virginia to Missouri, S to Florida to Texas	5–8
C. succulenta Schrad. ex Link *C. florifera* Sarg.; *C. laxiflora* Sarg.	**fleshy hawthorn,** longspine or succulent hawthorn	Nova Scotia to Montana, S to North Carolina & Utah	5–8
C. tracyi Ashe ex Egglest. *C. montivaga* Sarg.	**Tracy hawthorn,** mountain hawthorn	Texas & NE Mexico	3–5
C. triflora Chapman	**three-flower hawthorn**	Tennessee, S to Georgia & Louisiana	4–6
C. uniflora Münchh. *C. bisculcata* Ashe; *C. choriophylla* Sarg. *C. dawsoniana* Sarg.; *C. gregalis* Beadle	**dwarf haw,** one-flowered hawthorn, & dwarf thorn	New York to Missouri, S to Florida & NE Mexico	$^{1}/_{2}$–4
C. viridis L. *C. amicalis* Sarg. *C. ingens* Beadle	**green hawthorn**, southern or tall hawthorn, green haw, green or southern thorn	Pennsylvania to Kansas, S to Florida & Texas	5–12

Sources: Beadle (1913), Brinkman (1974), Dirr (1998), Flint (1997), Foote and Jones (1989), Griffiths (1994), Jacobson (1996), Little (1980a&b), Palmer (1950, 1952), Phipps (1988, 1995, 1998a&b), Phipps and O'Kennon (1998), Phipps and others (1990), Sargent (1933), Strausbaugh and Core (1978), Tidestrom (1933), Vines (1960), Wasson (2001), Weakley (2002).

edible fruits in Asia, Central America, and various Mediterranean countries (Everett 1981; Guo and Jiao 1995; Mabberley 1997; Usher 1974). The fruits of some species contain higher concentrations of vitamin C than do oranges (*Citrus* L. spp.) (Morton 1981).

In recent years, cultivation of mayhaws native to the southeastern United States—including eastern, western, and rufous mayhaws—has increased (Bush and others 1991; Payne and Krewer 1990; Payne and others 1990). Mayhaws are atypical among the hawthorns in their early flowering period (from late February through mid-March) and their early fruit ripening dates (May) (table 2) (Payne and Krewer 1990). At least 12 cultivars have been selected for improved fruit size, yield, and ease of harvest, and these are grown for production of jellies, juices, preserves, and wine. Vitamin contents are comparable to those found in manzanilla (*Crataegus mexicana* Moc. & Sesse ex DC.) (Payne and others 1990), a species used for medicinal purposes in Central America (Morton 1981). However, until propagation, production, and harvest techniques are improved, limited supplies of fruits derived from orchard-grown plants will necessitate further collection of fruit from native stands (Bush and others 1991). Other North American *Crataegus* species cultivated for fruit production are black, yellow, and downy hawthorns (Mabberley 1997; Usher 1974).

Many hawthorn taxa are grown in North America and Europe solely as ornamental plants because of their small stature, brilliant flowers in spring, and brightly colored fruits in fall (Bean 1970; Christensen 1992; Dirr 1998; Everett 1981; Flint 1997; Griffiths 1994; Jacobson 1996; Knees and Warwick 1995; Krüssmann 1984; Mabberley 1997). In the United States, the most commonly encountered hawthorn taxa in cultivation include Washington,‘Winter King’, cockspur, plumleaf, and Lavalle hawthorns (*C.* × *lavallei* Henriq. ex Lav.) (Bir 1992; Dirr 1998; Everett 1981; Flint 1997). One caution, however, is necessary with regard to cultivated hawthorns. Because only a partial understanding of the taxonomy of native populations of hawthorns now exists, especially in North America, it is likely that identities of many cultivated hawthorns may be either incorrect or imprecisely defined.

Hawthorns are important for wildlife. They offer good nesting sites for birds because of their dense branching and their thorns, which deter predators (Martin and others 1961). Fruits of many species are consumed by songbirds, game birds, small mammals, and ungulates (Shrauder 1977). Hawthorns are recommended commonly by professionals as landscaping and shelterbelt plants that can attract wildlife (Bir 1992; Elias 1987; Foote and Jones 1989; Morgenson 1999; Petrides 1988).

Flowering and fruiting. Flowers always appear after leaf emergence and are borne either in flat-topped inflorescences termed corymbs or in globular inflorescences termed umbels (Phipps 1988). Flower color is usually white, but rarely, pink-flowered variants are found in horticultural selections. From 1 to 25 flowers can be produced per inflorescence (Christensen 1992; Phipps 1988). Flowers usually contain 5 petals and 5 to 20 stamens and have a fetid odor in many species.

Hawthorn fruits are known as pomes, although the seeds and their bony endocarps are termed pyrenes, or nutlets (figures 1 and 2). Between 1 and 5 pyrenes are produced in each pome. Although most species produce flowers in spring and fruits in fall, mayhaws are notable for their early flowering and fruit ripening period. Some species drop fruits in autumn, and others have fruits that persist through winter. Timing of these events is important to horticulturists and wildlife and game managers (table 2).

Collection of fruits, seed extraction, cleaning, and storage. Mature fruits of most hawthorn species are collected readily from the ground in autumn, whereas fruits of species that tend to hold their fruits through the winter must be hand-picked from the trees (Brinkman 1974). Harvested fruits can be macerated to separate the seeds from the fleshy pericarp (Munson 1986). The macerated pericarp material can be removed by water flotation, and the seeds should then be air-dried. Seed yield data are available for only a few species, and there is considerable variability among them (table 3).

As an alternative to macerating the fruits and subsequently storing the seeds, fermenting freshly collected, undried fruits of western mayhaw for 4 or 8 days yielded 93% germination. However, fermentation periods > 8 days adversely affected seed germination (Baker 1991). Most other reports stated that acid scarification and/or cold stratification are obligatory to enhance seed germination. Fermentation treatments may prove extremely beneficial in reducing the time required to produce seedlings of hawthorns. However, further research on a wide range of hawthorns is needed before making general conclusions about the usefulness of such treatments.

After extracting, cleaning, and drying, the seeds should be stored under refrigerated conditions (Dirr and Heuser 1987; Hartmann and others 2002). All indications are that hawthorn seeds are orthodox in storage behavior, but reports on long-term seed viability during storage do not all agree.

Table 2—*Crataegus,* hawthorn: phenology of flowering and fruiting, and color of ripe fruit

Species	Flowering	Fruit ripening	Color of ripe fruit*
C. aestivalis	Mar	May–June	Lustrous, scarlet
C. x anomala	May	Sept–Oct	Bright crimson
C. berberifolia	Mar–Apr	Oct	Orange with red face
C. brachyacantha	Apr–May	Aug	Bright blue with white wax
C. brainerdii	May–June	Sept–Oct	Red
C. calpodendron	May–June	Sept–Oct	Orange-red to red
C. chrysocarpa	May–June	Aug–Sept	Yellow to orange to crimson
C. coccinoides	May	Oct	Glossy, dark crimson
C. crus-galli	June	Oct	Dull red
C. dilatata	May	Sept	Scarlet with dark spots
C. douglasii	May	Aug–Sept	Lustrous, black to chestnut-brown
C. erythropoda	Apr–May	Oct	Red to wine purple, brown, or black
C. flabellata	May	Sept	Crimson
C. flava	Apr	Oct	Dark orange-brown or yellow
C. greggiana	Apr	Oct–Nov	Bright red
C. harbisonii	May	Oct	Bright red or orange-red
C. intricata	May–June	Oct	Greenish or reddish brown
C. lacrimata	Apr	Aug	Dull yellow or orange or red
C. laevigata	Apr–May	Sept–Oct	Deep red
C. marshallii	Apr–May	Oct	Bright scarlet
C. mollis	May	Aug–Sept	Scarlet with large dark dots
C. monogyna	May	Sept–Oct	Bright red
C. nitida	May	Oct	Dull red covered with white wax
C. opaca	Feb–Mar	May	Lustrous scarlet with pale dots
C. pedicellata	May	Sept	Glossy, scarlet
C. persimilis	May–June	Oct	Bright red
C. phaenopyrum	May	Sept–Oct	Lustrous scarlet
C. piperi	May–June	Aug–Sept	Salmon-orange to scarlet
C. pruinosa	May–June	Oct–Nov	Dark purple-red
C. pulcherrima	Apr–May	Sept–Oct	Red
C. punctata	May–June	Sept–Oct	Dull red or bright yellow
C. reverchonii	May	Oct	Shiny or dull red
C. rufula	Mar–Apr	June–July	Red
C. saligna	May	Oct	Red to blue-black
C. sanguinea†	May	Aug–Sept	Bright red
C. spathulata	Apr–May	Sept–Oct	Red
C. succulenta	May–June	Sept–Oct	Bright red
C. tracyi	Apr–May	Sept–Oct	Orange-red
C. triflora	May	Oct	Red, hairy
C. uniflora	Apr–May	Sept–Oct	Yellow to dull red to brown
C. viridis	Apr–May	Sept–Oct	Bright red, orange-red, yellow

Sources: Beadle (1913), Brinkman (1974), Dirr (1998), Everett (1981), Flint (1997), Foote and Jones (1989), Jacobson (1996), Little (1980a&b), Palmer (1950, 1952), Phipps (1988, 1998a), Phipps and O'Kennon (1998), Sargent (1933), Vines (1960).

* Color of ripe fruit is highly arbitrary and varies in interpretation amoung authors due to lack of standardization. Accurate determinations of fruit color cannot be ascertained from herbarium specimens.

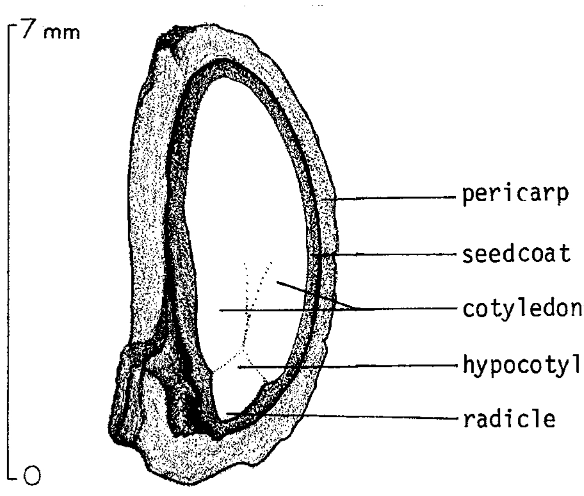
† Plants growing in Boston, Massachusetts, not in native habitat.

Dirr and Heuser (1987) stated that seeds of hawthorns, in general, can remain viable for 2 to 3 years in cold storage. St. John (1982), however, noted decreased seed viability in oneseed, cockspur, plumleaf, and scarlet hawthorns after storage for 2 years and recommended that seeds be stored for no more than 1 year. Bir (1992) found decreases in seed viability of Washington hawthorn after cold storage for 1 year. However, Christensen (1992) observed that under natural conditions, seeds of Eurasian species may require from 2 to 6 years to germinate.

Pregermination treatments and germination tests. Seeds of many hawthorns exhibit double dormancy (Hartmann and others 2002). Therefore, pregermination treatments usually consist of acid scarification followed by a period of cold stratification (Brinkman 1974; Hartmann and others 2002). Many authors also recommend periods of warm stratification for selected species (Brinkman 1974; Dirr and Heuser 1987; Morgenson 1999; St. John 1982; Young and Young 1992). Brinkman (1974) stated that "all" seeds of hawthorns exhibit embryo dormancy, therefore

Figure 1—*Crataegus*, hawthorn: cleaned pyrenes (nutlets) of *C. crus-galli*, cockspur hawthorn (**top**), *C. douglasii*, black hawthorn (**second**), *C. mollis*, downy hawthorn (**third**), *C. phaenopyrum*, Washington hawthorn (**fourth**), *C. punctata*, dotted hawthorn (**fifth**), *C. succulenta*, fleshy hawthorn (**bottom**).

Figure 2—*Crataegus*, hawthorn: longitudinal section of a pyrene (nutlet).



requiring cold stratification. This is reflected in the general recommendation by Hartmann and others (2002) that, following acid scarification, seeds should be stratified for 5 months at 4 °C. However, Kosykh (1972) reported that acid scarification and cold stratification for 6 months did not enhance germination of several species of hawthorns occurring in the Russian Crimea. In *C. mexicana*, cold stratification failed to enhance germination in seeds that were pretreated with 1 or 3 minutes of hot-water soaking at 80 °C (Felipe Isaac and others 1989). The fermentation work by Baker (1991) with western mayhaw also demonstrated high germination percentages without pretreating the seeds via acid scarification or cold stratification. Phipps (1998c) commented that hawthorns native to warm temperate climates possessed only endocarp dormancy, whereas those species native to regions with colder climates displayed embryo dormancy in addition to endocarp dormancy. In a large and geographically widely distributed group such as hawthorn, these different observations are not surprising.

Differences in endocarp thickness have been noted by several authors. Endocarp thickness in oneseed hawthorn varies not only among individual trees, but also over years (St. John 1982). Some species (for example, Washington hawthorn) lack thickened endocarps and can germinate without acid scarification (Bir 1992; Brinkman 1974; Dirr and Heuser 1987; Hartmann and others 2002). In contrast, other hawthorns exhibit highly thickened endocarps (up to 0.5 cm) and require up to 7 to 8 hours of acid scarification (Dirr and Heuser 1987) before other germination pretreatments can be imposed. Table 4 summarizes pregermination treatments that have been tested on various species of *Crataegus*.

Tipton and Pedroza (1986) studied germination requirements of Tracy hawthorn and failed to achieve germination > 54% in seeds pretreated with acid scarification for up to 4.5 hours, in combination with other pretreatments (table 4). They speculated that a combination of longer durations of acid scarification (for example, > 4.5 hours), lower germination chamber temperatures (for example, < 16 °C), shorter durations of warm stratification (for example, 0 to 60 days), and longer durations of cold stratification (for example, 100 to 322 days) might improve germination in this species. The low germination percentages observed may have been due to embryo decay caused by excessively long periods of warm stratification or high temperatures in the germination chamber, in combination with incomplete modification of the endocarp due to an inadequate duration of acid scarification. Interestingly, some seeds germinated during cold stratification before being placed into the germination chambers.

Morgenson (1999) noted differential responses of 3 hawthorns to acid scarification, as well as warm and cold stratification pretreatments. Specifically, he found that

Table 3—*Crataegus,* hawthorn: seed yield data

Species	Provenance	Seed wt/fruit wt kg/kg	Seed wt/fruit wt lb/100 lb	Average cleaned seeds/wt /kg	Average cleaned seeds/wt /lb	Samples
C. chrysocarpa	South Dakota	—	—	21,500	10,750	1
C. douglasii	Washington, Idaho, Oregon	0.15	15.2	45,200	22,600	6
C. phaenopyrum	—	—	—	59,600	29,800	1
C. punctata	Minnesota	0.11	11.3	9,400	4,700	2
C. sanguinea	Russia	0.15	15.0	—	—	—
C. succulenta	—	—	—	41,200	20,600	1

Source: Brinkman (1974).

although 2 hours of acid scarification did not enhance seed germination of Arnold and downy hawthorns, some beneficial effects on seed germination in fireberry hawthorn were noted, especially in combination with warm and cold stratification pretreatments. Germination of both Arnold and downy hawthorn seeds was optimized under a 60-day warm and 120-day (or more) cold stratification regime, with 37 and 51% germination occurring, respectively. For fireberry hawthorns, 90 to 120 days of warm stratification, followed by 120 to 180 days of cold stratification resulted in 18 to 27% germination. In all 3 species tested, extreme radicle elongation was observed in some treatments, for example, in all 120-day cold stratification combination treatments for fireberry hawthorns, and in some 60 and 120-day cold stratification combination treatments for Arnold and downy hawthorns.

In *C. azarolus* L., cold stratification treatments reduced abscisic acid (ABA) content in seeds, especially during the first 20 days, but only yielded 24% germination (Qrunfleh 1991). Work in England with oneseed, cockspur, plumleaf, and scarlet hawthorns resulted in as much as 80% germination (see table 4 for pregermination treatments) (St. John 1982). Using alternating 3-month periods of warm stratification at 21°C and cold stratification at 4 °C, seeds of oneseed hawthorn exhibited 31% germination after a warm-cold cycle and 55% after a cold-warm-cold-warm-cold cycle (Deno 1993). Utilizing these alternating cold-warm regimes with Washington hawthorn, 50% germination was attained with a warm-cold scheme, and 51% germination occurred with cold stratification only (Deno 1993). This latter result for Washington hawthorn agreed with data reported by Brinkman (1974). Studying seeds of downy hawthorn sown into old-field vegetation patches, Burton and Bazzaz (1991) noted a negative correlation between germination percentage and the quantity of plant litter on the soil surface. This suggested that seed germination in downy hawthorn may be inhibited by the presence of organic acids or allelochemicals released by decaying organic matter.

Official seed testing prescriptions are in place for only 2 species. AOSA (1993) recommends 2 hours of soaking in concentrated sulfuric acid, followed by 90 days of incubation at room temperature and then 120 days of moist-prechilling for downy hawthorn. Germination should then be tested on moist blotters or creped paper at 20/30 °C for 14 days. For oneseed hawthorn, ISTA (1993) prescribes 90 days of incubation at 25 °C, followed by 9 months of moist-prechilling at 3 to 5 °C. Germination is to be tested in sand at 20/30 °C for 28 days. Both organizations also allow tetrazolium staining to determine viability as an alternative to actual tests. For all hawthorn species, ISTA (1996) recommends cutting transversely one-third from the distal end of the seeds, then incubating for 20 to 24 hours in a 1% solution at 30 °C. The embryos must be excised for evaluation. Maximum unstained tissue is one-third the distal end and the radicle tip. Some germination test results are summarized in table 5.

Because hawthorns produce apomictic seeds, reports have appeared on clonal production of plants by seed propagation (Hartmann and others 2002). In western mayhaw, this phenomenon occurs widely because of the production of nucellar embryos and may be exploitable for production of superior clones (Payne and Krewer 1990; Payne and others 1990). Further study of apomixis in *Crataegus* is needed.

Nursery practice. Hawthorns are produced in nurseries utilizing both sexual and asexual propagation techniques. In horticulture, sexual propagation of hawthorns (via seeds) is important for production of large numbers of rootstocks, to which superior, clonal scions (often cultivars) are budded (Bush and others 1991; Dirr and Heuser 1987). In particular, this is necessary for rapid build-up of clonal orchards of desirable species of hawthorns (such as those with potential pomological interest), for which there are limited scion material and little knowledge of vegetative propagation by stem cuttings. Western mayhaw is a good example of such a species (Bush and others 1991). Brinkman (1974) recommended that if controlled seed pretreatment regimes (such as stored refrigerated conditions) are not used by nurseries, seeds should be sown in early fall (versus spring) to satisfy any potential requirements for cold stratification. This may be an adequate generalization for many hawthorns, although it is important to note the aforementioned exceptions for those species (for example, those from warm temperate climates) that will germinate either in shorter time periods without the cumbersome waiting periods involved in cold stratification or through innovative seed pretreatment techniques such as fermentation.

Research on vegetative propagation of hawthorns by stem cuttings is limited. Dirr and Heuser (1987) reported previous efforts as being "rarely successful," whereas Dirr

Table 4—*Crataegus,* hawthorn: pregermination treatments

		Stratification treatments			
	Scarification*	Warm period		Cold period	
Species	(hrs)	Temp (°C)	Days	Temp (°C)	Days
C. anomala	4.5	—	—	2–9	180
	0	21–27	30–90	2–9	90–180
C. crus-galli	2–3	21–25	21	Low†	21–135
	0	21	120	7	135
C. douglasii	0.5–3	—	—	5	84–112
C. mollis	2	25	90	5	120
	0	30	21	10	180
C. monogyna	—	25	90	3–5	270
	0.5–2	20	14–28	2–4	70–84
C. pedicellata	2	20	28	2–4	84
C. persimilis	4	20	14–28	2–4	70–84
C. phaenopyrum	0	—	—	5–10	135
C. punctata	0	21	120	5	135
C. sanguinea	2	21–25	21	5	21
	0	20–25	30	4–7	—
C. succulenta	0.5	—	—	4	110–140
C. tracyi	0, 0.5, 2.5, 4.5	21–27	0, 20, 60, 120	4	0, 20, 100

Sources: Brinkman (1974), Felipe Isaac and others (1989), Qrunfleh (1991), St John (1982), Tipton and Pedroza (1986), Young and Young (1992).

* Immersion time in sulfuric acid (H_2SO_4).

† Outdoor winter temperatures.

Table 5—*Crataegus,* hawthorn: germination test conditions and results

	Germination test conditions*					
		Temp (°C)			Germination	
Species	Medium	Day	Night	Days	Avg (%)	Samples
C. anomala	Soil	8	2	180	35	1
C. crus-galli	Soil	21	21	21	73	1
C. douglasii	Peat or sand	21	21	35–45	30†	6
C. mollis	Soil	21	21	—	42–50	3
C. phaenopyrum	Soil	21	21	—	71	2
	Peat	5	5	135	92	1
C. punctata	Peat	21	21	21	60	1
C. sanguinea	Peat	21	21	21	73	1
	Peat	4	7	30	50	2
C. succulenta	Soil	—	—	—	35–40	2
C. tracyi ‡	Germination blotters	16	16	28	0	2

Sources: Brinkman (1974), Tipton and Pedroza (1986).

* Light provided ≥ 8 hours per day. For each species, seeds were pretreated as shown in table 4.

† Sound seed was approximately 45% of total seeds sown.

‡ 16/8 hour light/dark cycle used.

(1998) and Hartmann and others (2002) make no mention of stem cutting propagation. However, 35% rooting was achieved utilizing softwood stem cuttings of 2 cultivars of western mayhaw—'Super Spur' and 'T.O. Super Berry'—treated with 8,000 ppm of the potassium (K) salt of indolebutyric acid (IBA) in combination with 2,000 ppm of the K salt of naphthaleneacetic acid (NAA) (Payne and Krewer 1990). Hardwood stem cuttings of this species (no clone specified) exhibited poor rooting, with callus visible 12 weeks after sticking cuttings, and ultimately only 10% rooting occurring (Bush and others 1991). However, softwood stem cuttings taken from new growth in mid-spring (in Calhoun, Louisiana) rooted in percentages > 80% in 8 weeks under intermittent mist. No differences in rooting occurred for cuttings treated with talc formulations of 0, 3,000, or 8,000 ppm IBA (Bush and others 1991). Clearly, these latter results suggest potential for developing readily producible clonal hawthorns by stem cuttings. If so, this could reduce the importance of seed-propagated hawthorns.

Vegetative propagation of hawthorns by grafting and budding is used widely in the horticulture industry. T-budding is one of the most viable vegetative propagation procedures employed for a wide range of cultivars of hawthorns (Dirr and Heuser 1987; Hartmann and others 2002). Root-grafting is also mentioned (Hartmann and others 2002) but rarely practiced. In the United States, Washington hawthorn is the "universal" rootstock, due to the fact that (a) seedlings are commonly available (because seeds of this hawthorn species germinate more easily than those of other species) and (b) bark-slippage occurs over a long season (late summer to early fall) (Dirr and Heuser 1987). Cultivars budded onto Washington hawthorn can be expected to grow 0.9 to 1.2 m in the growing season following budding (Dirr and Heuser 1987). Cultivars of European species (for example, English and oneseed hawthorns) should be budded onto rootstocks of European species, whereas hawthorns native to North America should be budded onto rootstocks of North American species (Dirr and Heuser 1987; Hartmann and others 2002). Aside from these constraints, T-budded hawthorns appear to be highly compatible across many species.

Several grafting procedures are employed (rather than budding procedures) in production of plants of mayhaw. Cleft grafts for larger rootstocks or whip-and-tongue grafts for small diameter rootstocks are used widely in late winter (Payne and Krewer 1990). In Louisiana, cleft grafting is the most popular grafting method used for western mayhaw (Bush and others 1991). Other species, such as parsley, cockspur, Washington, and yellow hawthorns, also can be used as rootstocks for mayhaws (in particular, western mayhaw) due to graft compatibility (Payne and Krewer 1990).

Brinkman (1974) called for additional trials on hawthorns to acquire more knowledge on seed biology. However, little comprehensive research has been conducted in the intervening 30 years on this subject. Much work remains to be done before a comprehensive understanding of propagation of hawthorns will be possible.

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Taxodiaceae—Redwood family

Cryptomeria japonica (L. f.) D. Don

sugi or cryptomeria

Gerald A. Walters and John K. Francis

Dr. Walters retired from the USDA Forest Service; Dr. Francis retired from the USDA Forest Service's International Institute of Tropical Forestry

Synonyms. *C. fortunei* Hooibrenk, *C. mairei* (Leveille) Nakai, *C. kawaii* Hayata, *Cupressus japonica* L. f., *Cupressus mairei* Leveille.

Other common names. Japanese cryptomeria, Japanese-cedar, goddess-of-mercy-fir, peacock-pine.

Growth habit, occurrence, and use. *Cryptomeria* is a monotypic genus native of Japan and China (Streets 1956). *Sugi—Cryptomeria japonica* [L. f.] D. Don—has been cultivated there since about 1300 A.D. for timber, shelterbelts, and environmental forestry. It was introduced to Hawaii for the same purposes about 1870 by Japanese immigrants (Carlson and Bryan 1959). An evergreen tree, it reaches heights of 36 to 46 m (Carlson and Bryan 1959; Dallimore and Jackson 1967; Troup 1921). Its wood is soft and fragrant; the red heartwood is strong and durable (Dallimore and Jackson 1967). It is used for boxes, poles, and general construction (Tsutsumi and others 1982). This species is also used for Christmas trees (Carlson and Bryan 1959; Dallimore and Jackson 1967).

Flowering and fruiting. Sugi is a monoecious species, with the male and female strobili located on different parts of the same branch. The female strobili are formed in fall and are fertilized when pollen is shed the following spring (Dallimore and Jackson 1967). Seed weight and percentage filled seeds are higher and seedling growth rate is greater when flowers are wind-pollinated (outcrossed) rather than selfed (Tabachi and Furukoshi 1983). In the native range in Japan, female cones begin to open between late January and mid-February and flower for 54 to 57 days. The male strobili begin to open about 25 days after the female strobili (Hashizume 1973). The solitary cones are globular and measure 13 to 19 mm in diameter. In Hawaii, cones ripen from July to September.

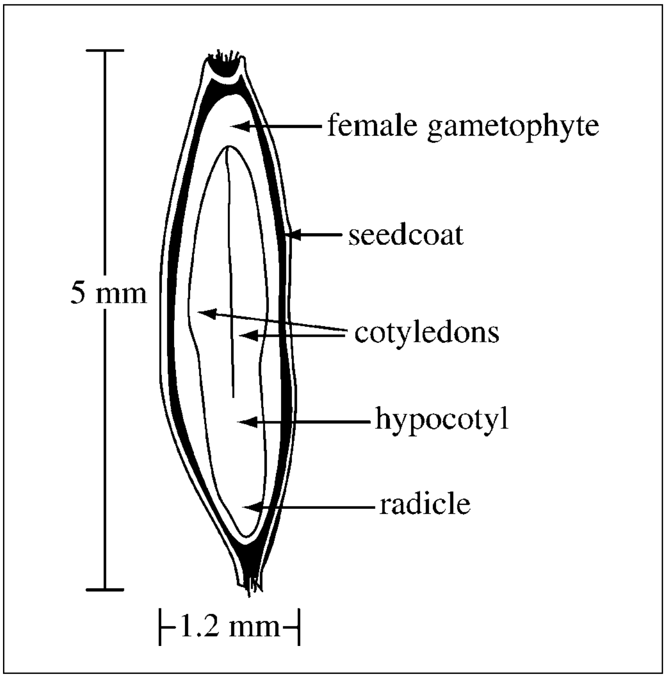
Seeds are shed during the same periods (Walters 1974). The seeds are dark brown and triangular, measuring 4 to 6 mm long and about 3 mm wide (Dallimore and Jackson 1967) (figures 1 and 2). Trees generally begin to produce seeds when 15 to 20 years old (Carlson and Bryan 1959). A 3-year-old orchard of rooted cuttings in Japan that was sprayed with gibberellic acid produced 1,082 kg/ha of seeds (967 lb/ac) 11 months later; 46% of the seeds were sound and 45% germinated (Itoo and Katsuta 1986).

Collection, cleaning, and storage. When the cones turn from grayish brown to reddish brown, they are ripe and should be picked. Cones should be immediately spread out to finish ripening. As the cones dry, seeds fall into trays; agitation aids in seed extraction. Seeds can be separated from chaff by winnowing. The number of seeds per weight ranges from 700,000 to 1,200,000/kg (320,000 to 550,000/lb) (Walters 1974; Ohmasa 1956). The optimal moisture content for storage is 10% (Shi 1985). After drying, the seeds should be stored in sealed polyethylene bags at 2 to 5 °C (Walters 1974). A drying agent placed in the bag aids storage (Ohmasa 1956).

Germination. Sugi seed germination is considered poor to very poor (Parry 1956). In Japan, the standard of sowing—30 g/m^2 (0.1 oz/ft^2)—is based on 30% germination (Ohmasa 1956). Sugi seeds should be soaked in cold water (0 °C) for about half a day, then put moist into plastic bags, and stored at 1 °C for 60 to 90 days before sowing (Walters 1974). Bags should be left open for adequate aeration. A mild fungicide can be added (Ohmasa 1956). Constant day/night temperature, whether high or low, adversely affects germination (RFC 1973). Germination is better in

Figure 1—*Cryptomeria japonica*, sugi: seed.

Figure 2—*Cryptomeria japonica*, sugi: longitudinal section through a seed.



seeds kept in the light than seeds kept in the dark (Chettri and others 1987). Official test prescriptions for sugi call for germination on top of moist blotters at alternating temperatures of 20 and 30 °C for 28 days; no pretreatment is necessary (ISTA 1993).

Nursery and field practice. Sugi seeds are sown in Hawaii from November to March. Sowing is by the broadcast method or by using a planter that has been adjusted to the proper seed size. The planter places seeds in rows about 15 to 20 cm (6 to 7 in) apart. Seeds are covered with 3 to 6 mm (1 to $2^1/_2$ in) of soil (Ohmasa 1956; Walters 1974). No mulch is used in Hawaii (Walters 1974), but a single layer of straw is used in Japan (Ohmasa 1956). The seedbeds are given about 75% shade for about 2 months (Walters 1974). Seedling density in the beds is about 220 to 330 seedlings/m^2 (20 to 30/ft^2). Frost damage to seedlings in early winter can be avoided by shading or shortening the daily period of exposure to solar radiation (Horiuchi and Sakai 1978). Seedlings are outplanted as 1+0 stock in Hawaii (Walters 1974). Sugi can be started from cuttings (Carlson 1959). In an experiment in which the trees were measured after more than 26 years, there was no significant difference in any measure of growth between trees started from seeds and those started from cuttings (Yang and Wang 1984).

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Cupressaceae—Cypress family

Cupressus L.

cypress

LeRoy C. Johnson and Robert P. Karrfalt

Mr. Johnson retired from the Pacific Southwest Forest and Range Experiment Station; Mr. Karrfalt is director of the USDA Forest Service's National Seed Laboratory, Dry Branch, Georgia

Growth habit, occurrence, and use. The true cypresses—genus *Cupressus* L.—are evergreen trees or shrubs native to the warm temperate areas of the Northern Hemisphere. The genus comprises about 15 species distributed throughout the western United States, Mexico, northern Central America, the Mediterranean region, northern Africa, and from southern Asia to Japan (Bailey 1923; Dallimore and Jackson 1967; Little 1966, 1979; Raizada and Sahni 1960; Wolf and Wagener 1948). The species native to North America are referred to as New World cypresses and those native to Europe, Africa, and Asia, as Old World cypresses (Gauseen 1968; Wolf and Wagener 1948).

Most New World cypresses are restricted in their occurrence (table 1). McNab and Sargent cypresses are often associated with serpentine soils (Griffin and Stone 1967; Hardham 1962). Arizona cypress and its subspecies are found in large stands confined mainly to north slopes, coves, benches, and canyon bottoms (Sudworth 1915). All reach their maximum sizes in moist, sheltered canyon bottoms. Of the 10 species, subspecies, and varieties native to California, none grow in large pure stands.

In the United States, cypresses are commercially propagated mainly for landscaping, Christmas trees, erosion control, windbreaks, and to a minor extent for lumber, fenceposts, fuelwood, and railroad ties. A factor limiting the widespread planting of cypresses in some parts of the United States is the cypress canker—*Seiridium cardinale* (W. Wegener) Sutton & I. Gibson—which attacks most species of cypress (Wolf and Wagener 1948). In California, this disease has eliminated some plantations of Monterey cypress. Only resistant species or strains of cypress should be planted where the cypress canker disease exists. In the South, Arizona cypress was at one time seriously considered for Christmas tree production (Goggans and Posey 1968; Grigsby 1969; Linnartz 1964; Posey and Goggans 1967), but its susceptibility to a foliage blight caused by *Cercospora sequoiae* Ellis & Everh. (Hepting 1971) has eroded this interest.

In Africa and New Zealand, Mexican and Monterey cypresses are planted for lumber and pulp production (Bannister 1962; Bannister and Orman 1960; Paterson 1963). Mexican cypress has become commercially important in Ethiopia, Kenya, and Tanzania (Bergsten and Sundberg 1990). Himalayan cypress is planted for timber, fuelwood, windbreaks, and animal fodder in Asia (Von Carlowitz 1986).

Italian cypress is the most widely planted of all the cypresses. It has been cultivated since ancient times (Bailey 1923; Bolotin 1964a); its columnar form and dark green foliage make it a popular tree for planting in formal gardens, along roads, and in cemeteries. This variety is propagated by seeds or cuttings. Seeds collected from pure stands or isolated columnar form varieties will breed true (Bolotin 1964b). The unusually narrow crown results from the ascending branches, which almost parallel the main trunk (table 2).

Monterey cypress is also extensively used in landscaping in spite of its high susceptibility to cypress canker disease. The rapid growth, lush green foliage, and dense crown make it ideally suited for planting around buildings, in windbreaks, and along roadsides.

Flowering and fruiting. Cypresses are monoecious. Staminate and ovulate strobili are produced on the ends of short twigs or branchlets. The staminate strobili are 3 to 7 mm long, cylindrical or oblong, and light green or rarely red. They become yellow as pollen-shedding time nears. Ovulate strobili at time of pollination are less than 6 mm long, subglobose to cylindrical, erect, greenish, and have 6 to 12 (rarely 14) distichously arranged scales. At maturity they may be 15 to 25 mm long.

Pollen is shed in late fall, winter, and spring. Planted trees of Arizona and Guadeloupe cypresses growing in the Eddy Arboretum, Placerville, California, shed their pollen in

Table 1—*Cupressus,* cypress: nomenclature and occurrence

Scientific name & synonym(s)	Common names	Occurrence
C. abramsiana C.B.Wolf *C. goveniana* var. *abramsiana* (C.B.Wolf) Little	**Santa Cruz cypress**	California: Santa Cruz & San Mateo Co.
C. arizonica Greene	**Arizona cypress**	Small, scattered areas in mtns of Arizona, New Mexico, S Texas, & N Mexico at 915–2,450 m
C. arizonica ssp. _arizonica_ Greene *C. arizonica* var. *glabra* (Sudsworth) Little *C. glabra* Sudsworth	**Arizona smooth cypress**	Mtn areas of central Arizona
C. arizonica ssp. _nevadensis_ (Abrams) E. Murray *C. arizonica* var. *nevadensis* (Abrams) Little *C. macnabiana* var. *nevadensis* (Abrams) Abrams *C. nevadendsis* Abrams	**Piute cypress**	California: Kern Co., Piute Mtns
C. arizonica ssp. _stephensonii_ (C.B.Wolf) Beauchamp *C. arizonica* var. *stephensonii* (C.B.Wolf) Little *C. stephensonii* C. B.Wolf	**Cuyamaca cypress**	California: San Diego Co., Cuyamaca Mtns
C. bakeri Jepson *C. bakeri* ssp. *matthewsii* C.B.Wolf	**Modoc cypress,** Baker cypress, Siskiyou cypress	California & Oregon in Siskiyou Mtns & NE California
C. forbesii Jepson *C. guadalupensis* var. *forbesii* (Jepson) Little *C. quadalupensis* ssp. *forbesii* (Jepson) Beauchamp	**tecate cypress**	San Diego Co., California, & Baja California, Mexico
C. goveniana Gord.	**Gowen cypress**	California coast from Mendocino Co. to San Diego Co.
C. goveniana ssp. _pygmaea_ (Lemmon) Bartel *C. goveniana* var. *pygmaea* Lemmon *C. pygmaea* (Lemmon) Sarg.	**Mendocino cypress,** pygmy cypress	California coast in Mendocino Co.
C. guadalupensis S.Wats.	**Guadalupe cypress**	Mexico, Guadalupe Island
C. lusitanica Mill.	**Mexican cypress,** cedar-of-Gog, Portuguese-cedar	Central Mexico, S to Guatemala & Costa Rica
C. macnabiana A. Murr.	**MacNab cypress**	N California: Sierra Nevada foothills & interior coastal range from Siskiyou to Napa Co.
C. macrocarpa Hartw. ex Gord.	**Monterey cypress**	Central California coast in Monterey Co. between Monterey & Carmel Bays; scattered on inland ridges
C. sargentii Jepson stands	**Sargent cypress**	California, in the coastal range in scattered from Mendocino Co. S to Santa Barbara Co.
C. sempervirens L. *C. sempervirens* var. *stricta* Aiton	**Italian cypress,** Mediterranean cypress	Mediterranean area
C. sempervirens var. _horizontalis_ (Mill.) Gord.	**spreading Italian cypress**	Mediterranean area
Cupressus torulosa D.Don *C. torulosa* var. *corneyana* Carr.	**Himalayan cypress,** *surai*	Temperate China to tropical India & Queensland, Australia

Sources: Johnson (1974), Sargent (1965), Sudworth (1967).

October and November (Johnson 1974). Native trees of Sargent cypress at Bonnie Doon, California, pollinate in December, and Monterey cypress at Point Cypress, California, pollinate in March (Johnson 1974).

The ovulate cones and their seeds ripen the second year, some 15 to 18 months after pollination. Mature cones (figure 1) are up to 30 mm in diameter, woody or leathery, and the peltate scales usually have a central mucro. Each cone produces 12 to 150 seeds (table 3).

Precocious cone production characterizes this genus. Male cones have developed on 1- and 2-year-old seedlings of Gowen and Mendocino cypresses, respectively (McMillan 1952). Female cone production often begins on trees younger than 10 years (Magini and Tulstrup 1955; McMillan 1952), but collectable quantities are usually not produced at such an early age. Treatment of seedlings of some species with various gibberellins can stimulate precocious flowering to a great degree. Mendocino, Mexican, and

Table 2—*Cupressus,* cypress: growth habit, height, cone and seed ripeness criteria

Species	Growth habit	Height at maturity (m)	Year first cultivated	Color ripeness criteria Cones	 Seed
C. arizonica	Straight central leader	15–21	1882	Dull gray to brown, sometimes purple	Medium to dark brown or deep purplish brown
ssp. *arizonica*	Straight trunk with or without turned up side branches	8–15	ca. 1909	Glaucous bloom over rich dark brown	Medium tan to brown or red brown
ssp. *nevadensi*	Erect tree with pyramidal crown	6–15	1930	Glaucous or silver gray	Rich light tan
ssp. *stephensonii*	Erect tree with straight central leader	9–15	1900	Dull gray or brown	Very dark brown
C. bakeri	Single stem, narrow crown	9–15	1917	Grayish to dull brown	Light tan
C. forbesii	Erect, irregularly branched tree	4.5–9	1927	Dull brown or gray	Rich dark brown
C. goveniana	Shrublike to small tree with single stem	6–18	1846	Brown to gray brown	Dull dark brown to nearly black
ssp. *pygmaea*	Shrublike to medium-sized tree	9–46	—	Weathered gray	Jet black to brownish
C. guadalupensis	Broad crown, trunk forking	12–20	ca. 1879	—	Dark brown & glaucous
C. lusitanica	Erect straight trunk with drooping branches	30	ca. 1670	Dull brown	Rich light tan
C. macnabiana	Brown crown lacking main trunk	6–12	1854	Brownish gray	Medium brown to glaucous brown
C. macrocarpa	Single trunk; symmetrical in sheltered areas	8–27	1838	Brown	Dark brown
C. sargentii	Single stem, slender or bushy tree	9–23	1908	Dull brown or gray often glaucous	Dark brown
C. sempervirens	Columnar with branches parallel to main trunk	46	B.C.	Shiny brown or grayish	—
var. *horizontalis*	Single stem with spreading crown	46	B.C.	Shiny brown or grayish	—

Sources: Dallimore and Jackson (1967), Raizada and Sahni (1960), Sargent (1965), Sudworth (1915), Wolf and Wagener (1948).

Arizona cypresses produced staminate strobili on seedlings 7 to 9 months old after foliar applications of gibberellin (GA_3), whereas the latter 2 species produced ovulate strobili at ages less than 24 months (Pharis and Morf 1967). Most native and planted cypresses produce an abundance of female cones. Guadelupe cypress rarely produces female cones under cultivation (Wolf and Wagener 1948), but the close relative, tecate cypress from Baja California, does (Johnson 1974). Occasional trees appear sterile, but this phenomenon is usually correlated with extremely heavy male cone production (Johnson 1974). Most cypresses have serotinous cones. Arizona and Italian cypresses open and shed their seeds when the cones ripen (Wolf and Wagener 1948). Cones on some trees within a stand will open and shed their seeds in July (Posey and Goggans 1967).

There is little information on the cone and seed insects that damage seed production in cypress. Larvae of the cypress bark moth—*Laspeyresia cupressana* Kearfott—are known to feed on maturing seeds of Gowen and Monterey cypresses and have earned the common name of "seedworm." Similar damage has been recorded on Monterey cypress from larvae of the moth *Henricus macrocarpana* Walsingham (Hedlin and others 1980). Over a dozen microorganisms have been identified in association with cypress seeds (Mittal and others 1990), but their effects on seed production, if any, are not known.

Cypress seeds vary widely in shape and size (figures 2 and 3). Length with wings attached ranges from 2 to 8 mm; width dimensions are slightly less. Seeds are flattened or lense shaped, and the wings are tegumentary extensions of the seedcoat. Seed length within a cone of Sargent cypress ranged from 2 to 5 mm (Johnson 1974). X-ray examination of bulk collections of several species showed that the smallest seeds were hollow (Johnson 1974). This phenomenon is most likely caused by lack of pollination or abortion after pollination (Johnson 1974).

Seed color is an important criterion for determining ripeness and an aid in differentiating species (table 2). Some species and geographic sources within a species can be distinguished by seed color. Seeds of Mendocino cypress from the central coast of Mendocino Co., California, have shiny jet-black seedcoats; seeds of the Anchor Bay strain from southern Mendocino Co. have brownish black seedcoats. Seeds of Guadalupe and tecate cypresses have the same

Table 3—*Cupressus,* cypress: seed yield data

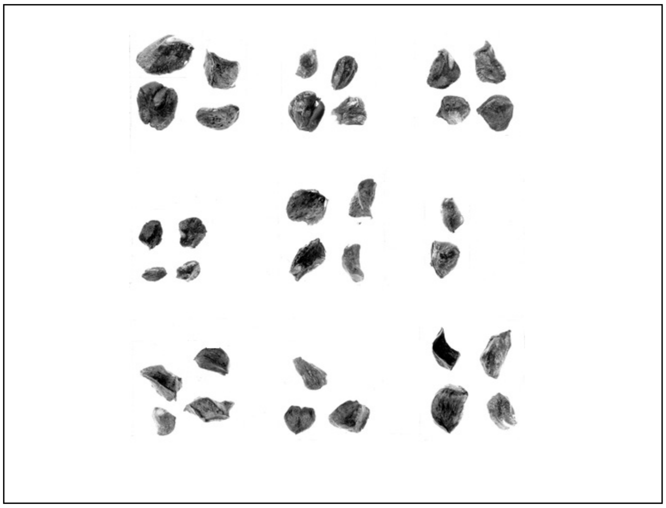
	Seeds (x1,000)/weight*						
	Range		Average				
Species	/kg	/lb	/kg	/lb	Samples	Scales/cone	Seeds/cone
C. arizonica	387.6–103.2	176.2–46.9	182.6	83.0	77+	6–8	90–120
ssp. *arizonica*	210.3–65.1	95.6–29.6	121	55.0	22+	5–10	90–100
ssp. *nevadensis*	201.1–86.7	91.4–39.4	126.5	57.5	11+	6–8	93
ssp. *stephensonii*	123.2–95.0	56.0–43.2	109.1	49.6	2	6–8	100–125
C. bakeri	387.2–316.8	176.0–144.0	359.9	163.6	4	6–8	50-85
C. forbesii	112.6–84.5	51.2–38.4	98.6	44.8	2	6–10	—
C. goveniana	313.3–253.4	142.4–115.2	283.4	128.8	2	3–5	90–110
ssp. *pygmaea*	246.4-232.3	112.0–105.6	239.4	108.8	2	8–10	130
C. guadalupensis	—	—	55.0	25.0	1	8-10	>100
C. lusitanica	—	—	261.8	119.0	1	6-10	75
C. macrocarpa	356.4–100.1	162.0–45.5	167.4	76.1	20	8–12	140
C. macnabiana	200.6–147.8	91.2–67.2	174.2	79.2	2	6–8	75–105
C. sargentii	147.8–98.6	67.2–44.8	123.2	56.0	2	6–10	100
C. sempervirens	149.6–118.8	68.0–54.0	137.9	62.7	9	8–14	64–280
C. torulosa	238-200	108–91	219	99	—	—	—

Sources: Goggans and Posey (1968), Rafn (1915), Raizada and Sahni (1960), Toumey and Stevens (1928), Von Carlowitz (1986), Wolf and Wagener (1948).

* Figures are for samples that have foreign matter (twigs, leaves, cone scales, etc.) removed but no attempt was made to separate sound from hollow and other nonviable seeds.

Figure 1—*Cupressus* goveniana, Gowen cypress: cones.

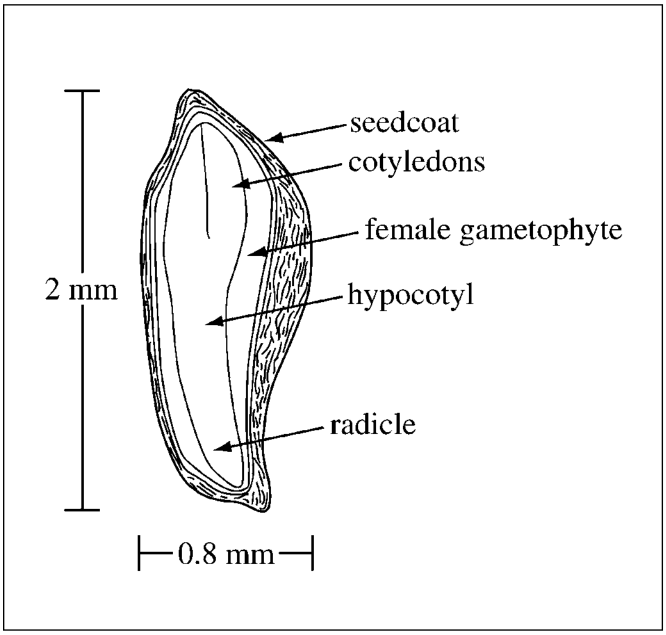
Figure 2—*Cupressus*, cypress: seeds of *C. arizonica*, Arizona cypress (**upper left**); *C. bakeri*; Modoc cypress (**upper center**); *C. goveniana*, Gowen cypress (**upper right**); *C. goveniana* ssp. *pygmaea*; Mendocino cypress (**middle left)**; *C. forbesii*, tecate cypress (**middle center**); *C. lusitanica*, Mexican cypress (**middle right**); *C. macnabiana*, MacNab cypress (**bottom left**); *C. macrocarpa*, Monterey cypress (**bottom center**); and *C. sargentii*, Sargent cypress (**bottom right**).



color, but seeds of Guadalupe cypress have a glaucous bloom and those of tecate cypress are shiny (Wolf and Wagener 1948).

Collection of cones. Mature cones are normally collected by hand from standing trees, usually by cutting clusters of cones with hand clippers. To ensure that the seeds are mature, only seeds from cones that matured the previous season or seeds with thoroughly darkened coats from the current season should be collected (Wolf and Wagner 1948). Goggans and others (1974) confirmed this rule in collections of Arizona cypress made in Alabama plantations. Cones that had turned gray in Alabama were over 5 years old and yielded seeds of quality not much better than immature cones. It is advisable, therefore, to collect only cones 4 years old and younger. Insect-damaged cones should not be collected because they do not readily open, and many of the seeds have been destroyed by the insects (Wolf and Wagener 1948). The time of year for collecting cones for most species is not critical if older cones are collected. Seeds of Italian cypress and some Arizona cypress sspp. are

Figure 3—*Cupressus arizonica*, Arizona cypress: longitudinal section through a seed.



shed when the cones are mature, so they must be collected as soon as they ripen. Cone and seed color aid in determining when the seeds are ripe (table 2).

Extraction and storage of seed. Cypress cones must be dried for the seeds to be released. Cones dried at room temperature 22 °C require 1 to 2 months for the scales to separate and the seeds to fall out (Posey and Goggans 1967). The process of cone opening can be speeded up by boiling the cones for 30 to 60 seconds or cutting each cone in half. Either method hastens the process of cone opening by several weeks. Clusters of cones should be cut apart so the scales can freely separate.

Sun-drying is another good method, provided the weather is hot and dry. Ripe cones of Gowen and Monterey cypresses collected in July were stored in a refrigerator at 1 °C for 2 days, then placed in trays. The cones opened and shed their seeds within 2 weeks when sun-dried in day temperatures of 32 to 35 °C with relative humidity ranging from 20 to 39% (Johnson 1974). Case-hardening is a potential hazard when sun-drying. This problem is minimized or eliminated by storing the cones for several days in a refrigerator, which will act as a desiccator.

Seeds fall out readily from completely mature cones with little or no tumbling. Insect-attacked and immature cones keep their seeds tightly attached to the cone scales, but such seeds usually have low viability and are best discarded with the cones. De-winging is not necessary, as the seeds have minute or no wings.

The percentage of filled seeds varies widely among species and among individuals within a species (table 4). Values for Arizona cypress ranged from 10 to 29% filled seeds, whereas those for the subspecies Arizona smooth cypress ranged from 1 to 49% filled seeds (Goggans and Posey 1968). Major improvements in seed quality can result from careful cleaning that minimizes loss of good seeds. This may be done with either a well-controlled air separation or a specific gravity table. Bergsten and Sundberg (1990) reported an upgrading of a Mexican cypress seedlot from 20 to 60% filled seeds with incubate-dry-separate (IDS) techniques (see chapter 3).

Cypress seeds are orthodox in storage behavior and maintain viability very well at low temperatures and moisture contents. There are no long-term storage test data available, but seeds of 7 species of cypress retained good viability during 10 to 20 years storage at temperatures of 1 to 5 °C (Johnson 1974; Schubert 1954; Toumey and Stevens 1928).

Pregermination treatments. Seeds of most cypress species exhibit some dormancy, and treatments are required for prompt germination. Ceccherini and others (1998) reported that, for 14 species of cypress, 30 days of stratification at 20 °C stimulated seed germination of all except Guadalupe cypress, and that the greatest benefit was shown by Monterey and Arizona smooth cypresses. At the USDA Forest Service's Institute of Forest Genetics at Placerville, California, seeds were stratified for 30 days at 1 °C (Johnson 1974). Stratification for 60 to 90 days has been recommended for Monterey cypress (Von Carlowitz 1986). Goggans and others (1974) also found 30 days of prechilling effective in breaking dormancy of Arizona cypress. The stratification was supplemented slightly by first soaking the seeds in a 0.1% citric acid solution. When time was short, a 72-hour water soak gave some benefit over just a 24-hour water soak. Seeds are often heavily contaminated with mold and bacteria, but control of the mold is feasible with fungicides during stratification and germination. Local extension experts should be consulted for current treatment recommendations. Treated medium and seeds can then be stored in plastic bags, jars, or petri dishes for the duration of the stratification period. Seeds stratified in a petri dish can be germinated in the same dish.

Germination. Germination should be tested with seeds placed on the top of moist blotters. ISTA (1993) recommends alternating temperatures of 30 °C (day) for 8 hours and 20 °C (night) for 16 hours for Arizona and Monterey cypresses, and a constant 20 °C for Italian cypress. Test periods of 28 days are prescribed for Arizona and Italian cypresses and 35 days for Monterey cypress.

Table 4—*Cupressus*, cypress: germination test results on stratified seeds

		Germination		Soundness	
Species	**Days**	**Average (%)**	**Samples**	**Average (%)**	**Samples**
C. arizonica	20	26	9	30	4
ssp. *nevadensis*	6	6	1	38	1
C. bakeri	30	12	2	36	2
C. forbesii	30	12	2	54	1
C. goveniana	30	22	2	93	2
ssp. *pygmaea*	30	31	2	—	—
C. macnabiana	30	1	1	5	1
	—	—	15	2	—
C. macrocarpa	30	24	4	82	4
	30	14	37	—	—
C. sargentii	30	13	2	41	2
C. sempervirens	—	—	—	27	9

Source: Johnson (1974).

* Soundness was determined by x-radiography examination before stratification (Johnson 1974).

Because of variable dormancy, AOSA (1998) also recommends paired tests for Arizona cypress, using unstratified and stratified (21 days) samples for each lot. Official test prescriptions have not been developed for the other cypress species, but similar conditions should be sufficient. For unstratified Himalayan cypress seeds (Rao 1988), the use of the alternating germination temperatures of 21 °C daytime and 9 °C night time gave 60% germination compared to 33% germination at constant 25 °C. Although light appears to be important, prechilling and alternating temperatures are the more significant promoters of germination. Light did not appear necessary for seeds of Arizona cypress (Goggans 1974). The seeds can be watered throughout the test with a mild solution of fungicide (the same formulation used above) with no phytotoxicity. Germination test results (table 4) have been low primarily because of the low percentages of sound seed that are common among seed lots of cypress. Good estimates of germination can be made with x-ray analysis of fresh seeds of Italian, Mexican, and Arizona cypresses (Bergsten and Sundberg 1990; Chavagnat and Bastien 1991).

Nursery practice. Fall-sowing of cypress seeds has been recommended (Johnson 1974; Wolf and Wagener 1948), but spring-sowing of stratified seeds is preferred. Germination of cypress is epigeal. Seeds should be sprinkled on the nurserybed and covered with a 4 to 5 mm (0.15 to 0.20 in) layer of soil and a light mulch. A density of 320 to 640/m^2 (30 to 60/ft^2) is recommended. Zeide (1977) was successful in direct seeding Italian cypress in Israel—annual precipitation, 447 mm (18 in)—in plots that were "mulched" with light colored stones of 2 to 5 cm ($^3/_4$ to 2 in) diameters. The seedlings grew out from under the stones.

Newly germinated cypress seedlings are particularly susceptible to damping-off fungus. When possible, nursery soil should be fumigated. As an added precaution, a fungicide should be used immediately after sowing and until the seedling stems become woody, which takes about 1 month's time. Cypresses can be outplanted as 1- or 2-year-old seedlings. A well-defined taproot and numerous lateral roots are formed in the first year. One-year-old seedlings of most species have only juvenile foliage.

Some species can be propagated vegetatively. Monterey cypress cuttings treated with indole-butyric acid (IBA) root well, whereas Italian cypress cuttings root well without treatment (Dirr and Heuser 1987).

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Fabaceae—Pea family

Cytisus scoparius (L.) Link

Scotch broom

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 uses. The genus *Cytisus* comprises about 80 species native to Eurasia and North Africa. Many are cultivated as ornamentals, and several of these have become more or less naturalized in the United States, especially in California (Munz and Keck 1959). Scotch broom—*C. scoparius* (L.) Link—was planted extensively for erosion control during the first half of the century (Gill and Pogge 1974) but is now considered a serious invasive weed throughout the range of its introduction in North America, Australia, and New Zealand (Bossard 1991). It has become the dominant species on several hundred thousand hectares of coastal and cis-montane vegetation, from Santa Barbara, California, north to British Columbia. It is a drought-deciduous shrub with angled, photosynthetic stems that is able to root-sprout following fire (Bossard and Rejmanek 1994; Gonzales-Andres and Ortiz 1997). It is largely useless as a browse-plant because of its toxic foliage, a feature that may permit it to increase at the expense of more palatable species (Bossard and Rejmanek 1994; Gill and Pogge 1974). It increases in response to disturbance of native vegetation and is also a serious weed problem in pine plantations in California and New Zealand.

However, because of its beauty and exceptional summer drought-hardiness, Scotch broom is considered valuable as an ornamental shrub for low-maintenance landscapes. The species is very showy in flower and its evergreen stems add interest to winter landscapes. There are over 60 named varieties (Wyman 1986).

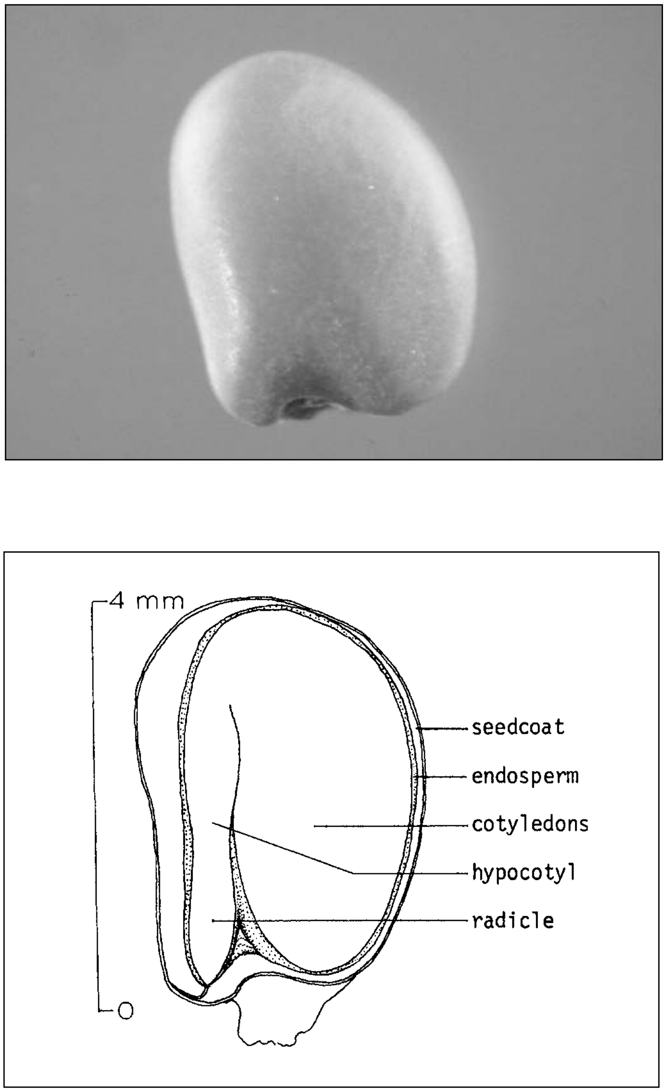
Flowering and fruiting. The perfect flowers are of typical pea-family form and appear on the plants in great profusion in May and June. Each flower must be "tripped" by an appropriate pollinator for fertilization to take place, so the mutualistic relationship with honey bees (*Apis mellifer*a L.) and native bumble bees is essentially obligatory (Parker 1997). Other native North American insects seem to ignore its fragrant blossoms, preferring to work the flowers of indigenous species. The result is that seed production may be severely pollinator-limited (Parker 1997). In spite of this, the plants may produce a prodigious number of seeds; the estimated mean annual production per plant was about 10,000 seeds in 2 California populations (Bossard and Rejmanek 1994). Host-specific pre-dispersal seed predators from Europe (a seed weevil and a bruchid beetle) have been introduced for biocontrol of Scotch broom in the Northwest, but so far these introductions have been largely ineffective, possibly because of asynchrony in the phenology of host and seed predator (Bravo 1980).

Plants reach reproductive maturity at about 4 years of age (Gill and Pogge 1974). The 5- or 6-seeded legumes (pods) ripen in August, and seeds are dispersed in September. The legumes open abruptly with a springing motion, vaulting the seeds some distance from the plant (Bossard 1991; Bossard and Rejmanek 1994). The seeds possess a strophiole or elaiosome at the hilar end (figure 1) and are secondarily dispersed by ants (Bossard 1991; Weiss 1909). At 2 California study sites, seeds were taken by mice and by ground-feeding birds, but these organisms were strictly seed predators and did not function as dispersers (Bossard 1991).

Seeds of Scotch broom have the capacity to form a persistent seed bank. Bossard (1993) found in seed retrieval experiments that 65% germinated the first year after dispersal, 20% germinated the second year, and 10% germinated the third year. About 5% of the seed population carried over for more than 3 years.

Seed collection, cleaning, and storage. After the fruits ripen but before they disperse, the legumes may be hand-stripped or picked up from beneath plants. They should be spread to dry, threshed, and screened to separate the seeds (Gill and Pogge 1974). Reported seed weights have averaged 125 seeds/g (57,500/lb) in 9 samples, and viability averaged 80% in 5 samples (Gill and Pogge 1974).

Figure 1—*Cytisus scoparius*, Scotch broom: longitudinal section through a seed (**bottom**) and exterior view (**top**).



No long-term storage data are available, but the seeds are orthodox and remain viable for many years in storage.

Germination and seed testing. Scotch broom seeds have water-impermeable (hard) seedcoats and require pretreatment in order to germinate. Once the seedcoats have been made permeable, the seeds germinate well over a wide range of temperatures and do not require any further pretreatment (Bossard 1993). Mechanical and acid scarification have been used to remove hard-seededness in this species, and the official seed-testing rules call for cutting or nicking the seedcoat at the cotyledon end, then soaking in water for 3 hours (ISTA 1993). Tests should be carried out on the tops of moist paper blotters for 28 days at 20/30 °C. More recently, the effect of heat on hard-seededness in Scotch broom has received attention. Tarrega and others (1992) report that dry-heating the seeds was as effective as mechanical scarification in terms of final percentage. Optimum time of heating varied with temperature from 1 minute at 130 °C to 15 minutes at 70 °C. Abdullah and others (1989) reported that repeated brief (3-second) immersion in boiling water resulted in complete elimination of hard-seededness, but low germination percentages indicated that some damage was occurring. They found that alternating the boiling water treatments with freezing treatments (immersion in liquid nitrogen for 15 seconds) resulted in the highest germination percentages as well as in complete removal of hard-seededness. This result was confirmed by Bossard (1993), who found that vigor of seedlings from hot/cold–treated seeds was much higher than that of seedlings from seeds subjected to dry heat only.

Nursery practice. Scotch broom is normally propagated from cuttings for ornamental planting in order to preserve varietal characters (Wyman 1986). If seed propagation is desired, seeds should be pretreated to remove hard-seededness prior to planting (Gill and Pogge 1974). The roots are delicate, and plants are more easily produced in container culture than as bareroot stock (Wyman 1986).

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