H

Styracaceae—Storax family

Halesia carolina L.

Carolina silverbell

Franklin T. Bonner

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

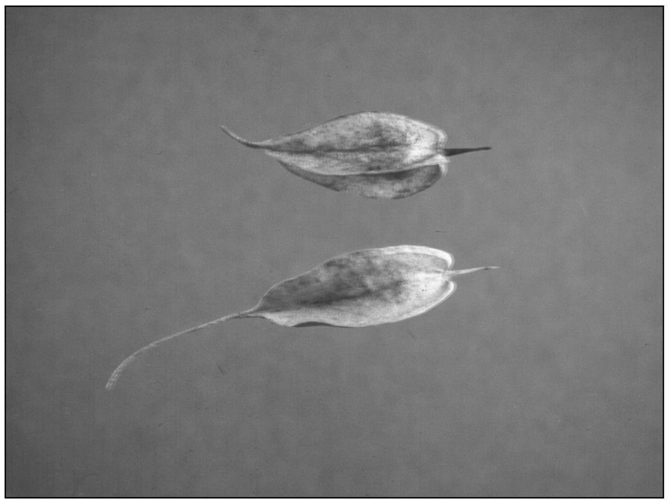
Other common names. opossum-wood, silverbell, snowdrop-tree.

Growth habit, occurrence, and uses. Carolina silverbell—*Halesia carolina* L.—is a small, deciduous tree found naturally from West Virginia and southern Illinois south to South Carolina, northwestern Florida, and Alabama, with small pockets as far west as Arkansas and Oklahoma (Sargent 1965; Sluder 1990). It has been successfully planted in Massachusetts and California and also to some extent in northern and central Europe. Carolina silverbell was first cultivated in 1756 (Bonner and Mignery 1974). It is highly valued as an ornamental, and its fruits are a source of food for wildlife.

Flowering and fruiting. The perfect, white (sometimes pink), bell-shaped, axillary flowers of Carolina silverbell are borne in fascicles of 1 to 5 in March to May (Brown and Kirkman 1990; Sluder 1990). The species is precocious and seedlings may flower when only a little over 1 m in height (Dirr and Heuser 1987). The fruit, which matures in August and September, is an oblong or oblong-ovate, dry, 4-winged, reddish brown, corky drupe 2.5 to 5 cm long. The ovary is a 4-celled ellipsoid stone, 13 to 16 mm long, usually containing only 1 seed (figures 1 and 2) (Bonner and Mignery 1974; Brown and Kirkman 1990; Sluder 1990). The fruits are persistent on the branches, and dispersal occurs well into the winter.

Collection, extraction, and storage. Carolina silverbell fruits may be collected from the trees in late fall and early winter. De-winging can be done mechanically (Thornhill 1968) and is recommended to reduce bulk and facilitate handling. Complete extraction of stones from the fruits is not necessary. Bonner and Mignery (1974) found about 2,600 to 5,500 de-winged fruits/kg (1,200 to 2,500/lb) using 2 samples. Although no data on long-term storage are available, dry cold storage of cleaned fruits has been recommended (Chadwick 1935).

Figure 1—*Halesia carolina*, Carolina silverbell: fruits.



Germination tests. The seeds are extremely dormant and they response best to warm stratification followed by cold stratification. Moist stratification at 13 °C for 60 to 120 days, then 60 to 90 days at 1 to 5 °C, has worked for seeds from many sources (Bonner and Mignery 1974), although more northern collections may require more than 90 days of cold (Dirr 1977). Other sources, however, seem to need only cold stratification (Chadwick 1935). Stratified seeds can be germinated in flats of sand or sand–peat mixtures for 60 to 90 days with 30 °C day and 20 °C night temperatures. Seven samples germinated in this manner averaged 53% germination (Bonner and Mignery 1974). Germination is epigeal (figure 3).

Nursery practice. Because of uncertain response to stratification, the most practical method of propagation from seeds may be to plant in the fall and allow 2 years for full germination (Dirr and Heuser 1987). Stratified seeds should always be used for spring-sowing. Some growers in the North have planted seeds in flats of soil and have kept them in a greenhouse during the early winter months. In January,

Figure 2—*Halesia carolina*, Carolina silverbell: longitudinal section through 2 embryos of a stone.

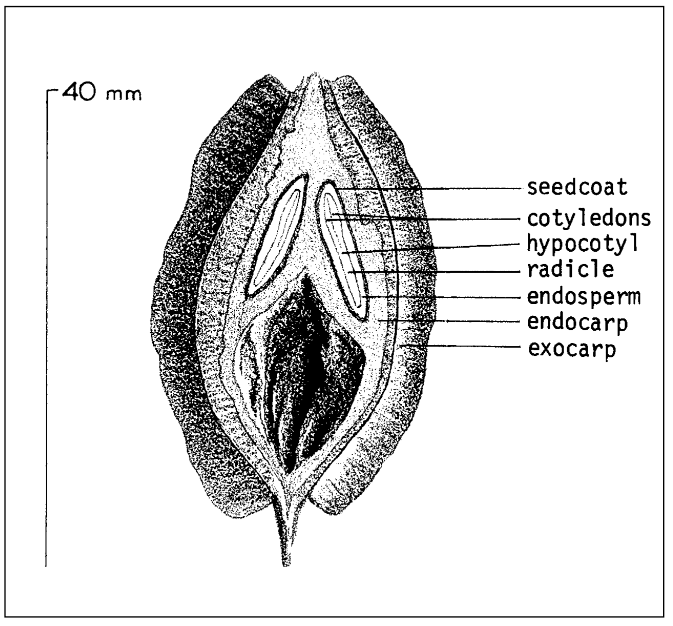
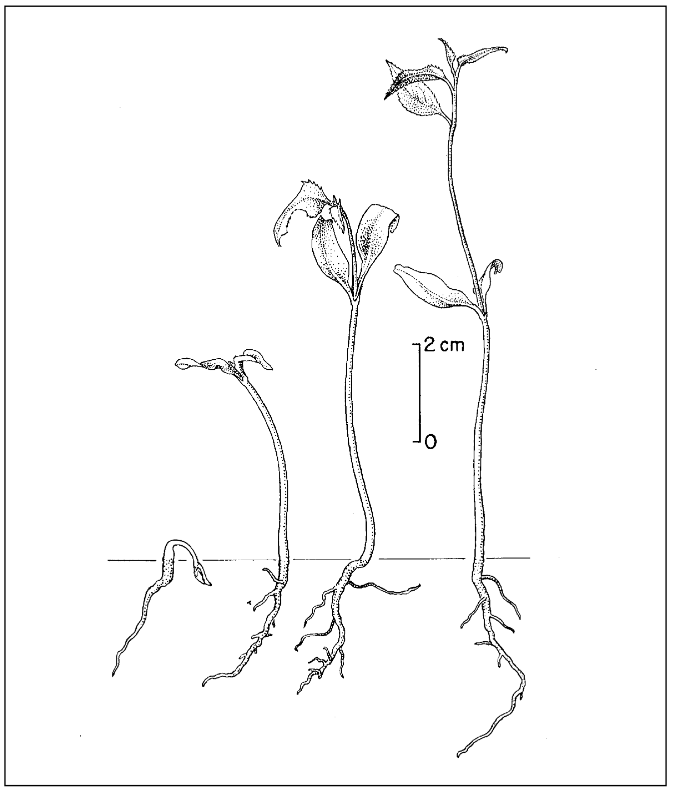


Figure 3—*Halesia carolina,* Carolina silverbell: seedling development after 1, 4, 16, and 40 days.



the flats are then moved to an outdoor cold frame for the cold part of the after-ripening treatment. The flats are protected by mulch or by board covers on the coldframes (Bonner and Mignery 1974).

Carolina silverbell can also be propagated by cuttings taken in mid-summer after shoot elongation has ceased but before fall hardening sets in. It is best to treat cuttings with 2,500 or 10,000 ppm indole butyric acid (IBA) solution and place them in peat or perlite under mist. Rooting success at levels of 80 to 100% can be expected (Dirr and Heuser 1987; Sluder 1990). Micropropagation techniques are also under study (Brand and Lineberger 1986).

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H

Hamamelidaceae—Witch-hazel family

Hamamelis L.

witch-hazel

Jill R. Barbour and Kenneth A. Brinkman

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

Growth habit, occurrence, and use. Witch-hazels are deciduous shrubs or small trees that attain heights of 2 to 10 m (table 1). American witch-hazel is native from Nova Scotia to southeastern Minnesota, south to Missouri, southeastern Oklahoma and Texas, and east to central Florida (Little 1953). First cultivated in 1736 (Rehder 1940), American witch-hazel is used in environmental plantings largely because it flowers in late autumn. The species provides seeds for birds and browse for wildlife (Van Dersal 1938). Bark, leaves, and twigs have been used medicinally in the form of extracts. Another species—Ozark witch-hazel—is a shrub of the Ozark region of Missouri, Arkansas, and Oklahoma but is seldom planted. Japanese and Chinese witch-hazels are popular introduced ornamentals that bloom in the spring. *Hamamelis* × *intermedia* 'Arnold Promise', a hybrid of Japanese and Chinese witch-hazels, was first produced at the Arnold Arboretum (Hora 1981).

Flowering and fruiting. The spider-like yellow or rusty red flowers of American witch-hazel open in September or October, but the ovules are not fertilized until the following May. The fruits ripen early the next autumn (Rehder 1940; Van Dersal 1938). Members of the witch-hazel family have catkins for flowers and they are wind-pollinated (Johnson 1973). Ozark witch-hazel flowers from midwinter to spring (Fernald 1970). Capsules (figure 1) burst open when dry, each discharging 2 shiny black seeds (figure 2). There is limited dispersal by birds. Annual fruit production is highly variable, with abundant fruit crops occurring 1 out of 4 years (DeSteven 1982).

Developing witch-hazel seeds are the larval food of the beetle *Pseudanthonomus hamamelidis* Pierce (Curculionidae) (DeSteven 1982). Weevil eggs are laid on the fruits from mid-June to early July and the newly hatched larvae feed on the fruits from mid-July to September. Lepidopteran caterpillars may also consume the seeds. The 2 most abundant species are in the families Gelechiidae and Pyralidae, and 3 other "occasional" species are in the families Nolidae,

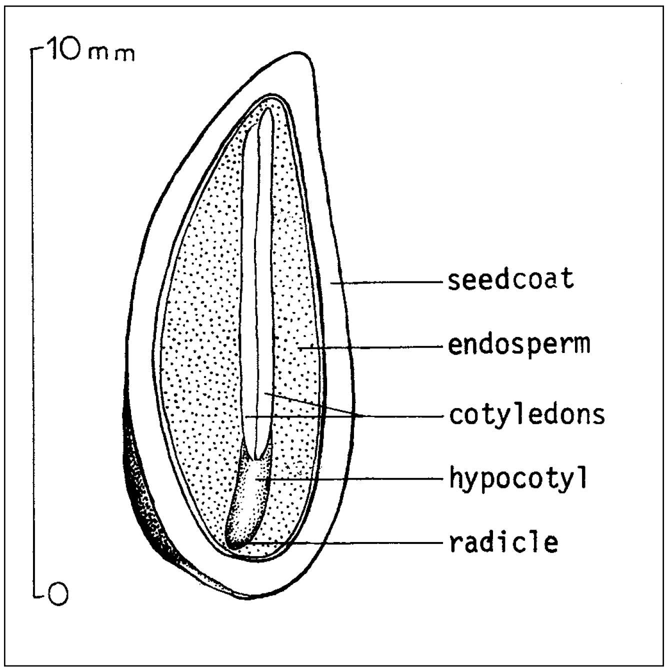
Figure 1—*Hamamelis virginiana,* witch-hazel: fruits (capsules) before and after seeds were discharged.

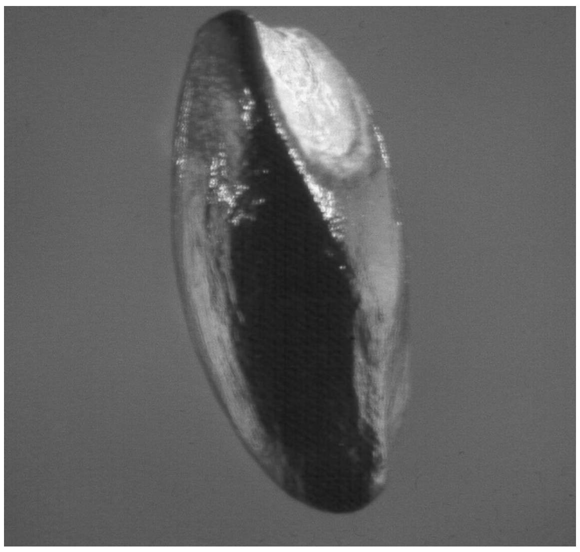
Table 1—*Hamamelis,* witch-hazel: nomenclature and occurrence

Scientific name	Common name	Occurrence	Height (m)
H. japonica **Siebold & Zucc.**	**Japanese witch-hazel**	Japan	10
H. mollis **D. Oliver**	**Chinese witch-hazel**	W central China	10
H. vernalis **Sarg.**	**Ozark witch-hazel**	Ozark Mtns of Missouri & Arkansas	2
H. virginiana **L.**	**American witch-hazel**	E US & Canada	7–10

Sources: LHBH (1976), Hora (1981).

Figure 2—*Hamamelis virginiana*, witch-hazel: longitudinal section through a seed (**top**) and exterior view (**bottom**).





Phalaenidae, and Geometridae (DeSteven 1982). Small mammals begin feeding on the seeds once they mature in the autumn. Only 14 to 16% of the seeds survive the predation by insects and mammals (DeSteven 1982)

Collection, extraction, and storage. Witch-hazel fruits should be picked in early autumn before they split open and discharge their seeds. Ripe fruits are dull orange-brown with blackened adhering fragments of floral bracts, but the seeds apparently mature as early as August before the fruit coat is fully hardened (Sandahl 1941). Fruits should be spread out to dry so the seeds may be separated from the capsules by screening. Two samples had 19,200 and 24,000 seeds/kg (8,727 and 10,909 seeds/lb) (Brinkman 1974). Fresh seeds can be stored dry in sealed containers at 5 °C for at least 1 year without loss of viability. For over-winter storage before spring-planting, seeds should be stratified in a 1:1 mixture of dampened sand and peat moss at 5 °C. The stratification medium should be 2 to 3 times the volume of seeds (Fordham 1976).

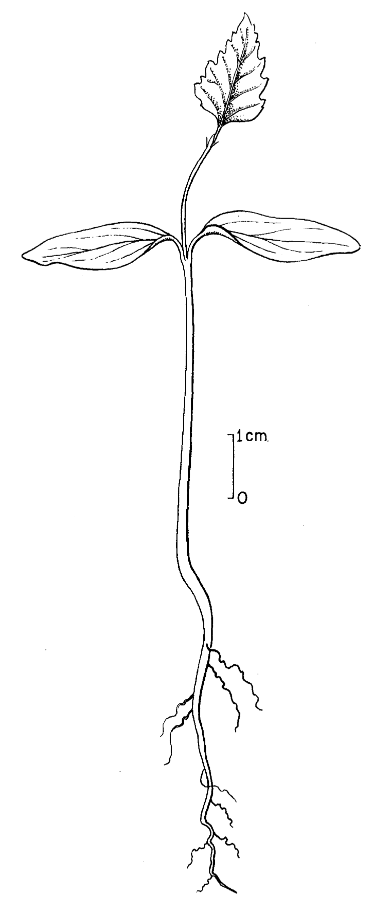
Germination and seed testing. Some stratified seeds germinate in the first spring but many remain dormant until the following year. Dormancy is due to conditions in both seedcoat and embryo, but satisfactory methods for overcoming dormancy have not been found. Rivera and others (1937) subjected witch-hazel seeds to pressures of 2,070 to 413,700 kN/m^2 (300 to 60,000 lb/in^2) at temperatures of 0, 25, and 50 °C and found that none of these conditions resulted in germination. In a series of tests, Brinkman (1974), stratified seeds for 60 days at 30 °C (day) and 20 °C (night) plus 90 days at 5 °C. When tested in sand at 30 °C (day) and 20 °C (night), 17% of these seeds germinated in 60 days.

Work in England on American witch-hazel seeds stratified for 2 months of warm and 2 months of cold, then 2 months of warm and 4 months of cold, produced 88% germination. The same seeds stratified for 2 months of warm and 1 month of cold, then $^1/_2$ month of warm and 4 months of cold, produced 84% germination. Chinese witch-hazel seeds stratified for 3 months of warm and 3 months of cold resulted in 88% germination. Ozark witch-hazel seeds germinated 70% after 3 months of cold stratification; 75% after 3 months of warm and 3 months of cold; 81% after 4 months of warm and 3 months of cold; and 85% after 5 months of warm and 3 months of cold (Dirr and Heuser 1987). A study at the Arnold Arboretum showed that Ozark witch-hazel germinated about as well after cold stratification only as it did after 2 stages of pretreatment (Fordham 1976). The Arnold Arboretum has found that 5 months of warm stratification followed by 3 months of cold treatment was satisfactory for witch-hazel seeds (Fordham 1991).

Chemical staining with 2,3,5-triphenyl tetrazolium chloride (TZ) is the preferred laboratory method for testing the viability of witch-hazel (Moore 1985). One-fourth of the seed opposite the radicle is clipped off to allow for the seed to imbibe the chemical. After staining, the seed is cut longitudinally to expose the embryo for observation. The average viability of 19 samples of witch-hazel seeds was 59% with a range of 0 to 97% (Brinkman 1974).

Nursery practice. Witch-hazel seeds may be fall-sown in the nursery as soon as collected, or stratified seeds may be sown in the spring. Limited trials show that seeds collected as early as August and sown by early October results in as much as 90% germination the following spring (Heit 1968; Sandahl 1941). Fall-sowing is recommended; the seedbeds should be mulched over winter and uncovered at germination time in the spring. For spring-sowing of stratified seeds, seedbeds should be prepared as early as soil conditions permit. Sowing in drills spaced 20 to 30 cm (8 to 12 in) apart will facilitate weeding and cultivating. Secondary leaves may develop on a seedling within 21 days after germination (figure 3). Propagation by layering also is possible.

Figure 3—*Hamamelis virginiana*, witch-hazel: seedling at 21 days after germination.



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Rosaceae—Rose family

Heteromeles arbutifolia (Lindl.) M. Roemer

Christmasberry

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 *Heteromeles* has only a single species—*H. arbutifolia* (Lindl.) M. Roemer, also known as *H. salicifolia* (K. Presl.) Abrams (Phipps 1992). It is closely related to the large tropical genus *Photinia* Lindl., to which it has sometimes been referred. Christmasberry, also known as toyon, California-holly, and hollywood, is a long-lived shrub or small tree, 2 to 10 m in height, that sprouts freely after fire from a subterranean burl. It has shiny, leathery, evergreen leaves that are sharply toothed along the margins. A common constituent of chaparral vegetation throughout California and Baja California, it is usually found on less harsh, more mesic microsites. Christmasberry is useful for erosion control, is a source of honey, and has leaves and fruits that provide food for wildlife. It has also been widely planted in California as an ornamental for park, freeway, and home landscape use (Magill 1974). The attractive foliage and fruits are cut and used for their decorative value.

Flowering and fruiting. Unlike many chaparral shrubs, Christmasberry is summer-flowering (Magill 1974). The numerous, small flowers are borne in flat-topped or convex terminal inflorescences. The flowers are perigynous and have 5 separate petals, 10 stamens, and a 2- to 3-carpellate ovary. The fruits are bright red to orange, 2- or 3-seeded juicy pomes that are 6 to 10 mm in diameter. They ripen from October through January and are dispersed by birds. Good crops are reported to occur yearly (Keeley 1992).

Seed collection, cleaning, and storage. Christmasberry fruits may be clipped or stripped from the branches once they have attained a bright red or orange color (Magill 1974). They should be soaked in water and allowed to ferment slightly. (However, too-long a soaking period can damage the seeds, which have soft, pliable seedcoats.) The seeds (figure 1 and 2) may then be separated from the pulp by passage through a macerator, followed by flotation to remove the pulp. Small lots can be hand-rubbed through a large-holed screen. The seeds may then be allowed to dry. Once dry, any flat, unfilled seeds can be removed by screening.

Figure 1—*Heteromeles arbutifolia,* Christmasberry: seeds.

Christmasberry seed weight is apparently highly variable. Magill (1974) reported a mean seed weight of 19 mg and count of 52,630 seeds/kg (23,900/lb), whereas Keeley (1991) reported a seed weight of 5.5 mg and count of 181,820/kg (82,500/lb). Martineja and Bullock (1997) examined Christmasberry seed weight as a function of habitat variables. They found no correlation with latitude or annual precipitation but did find a significant increase in seed weight with increasing elevation. Overall mean seed weight for their 12 Christmasberry populations was 36 mg and seed count was 27,800 seeds/kg (12,600/lb), with weight ranges of 28 to 49 mg and counts of 20,400 to 35,700 seeds/kg (9,200 to 16,200/lb).

Christmasberry seeds have limited longevity at room temperature, but they are probably orthodox in storage behavior. Keeley (1991) reported a shelf life of less than 1 year in laboratory storage. The seeds were also damaged or killed by high temperature treatments. One hour at 70 °C reduced viability from 99 to 33%, and 5 minutes at 120 °C resulted in essentially complete mortality (Keeley 1987). Magill (1974) recommended sealed storage at low tempera-

Figure 2—*Heteromeles arbutifolia*, Christmasberry: longitudinal section through a seed.

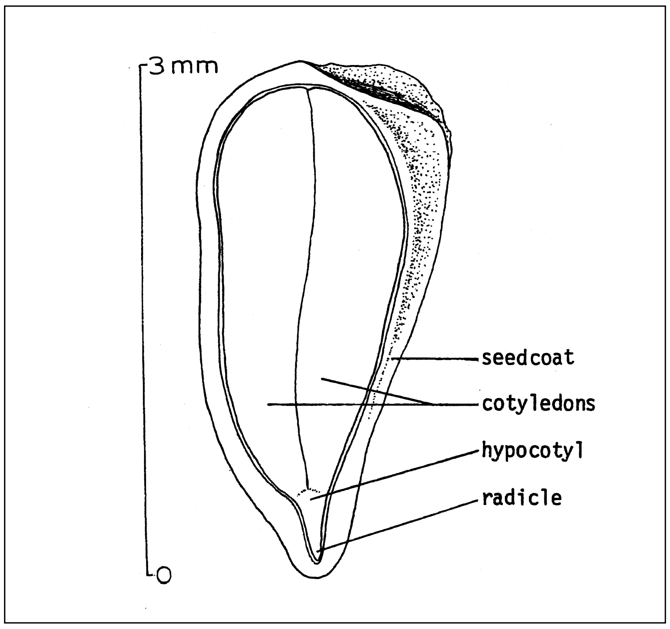
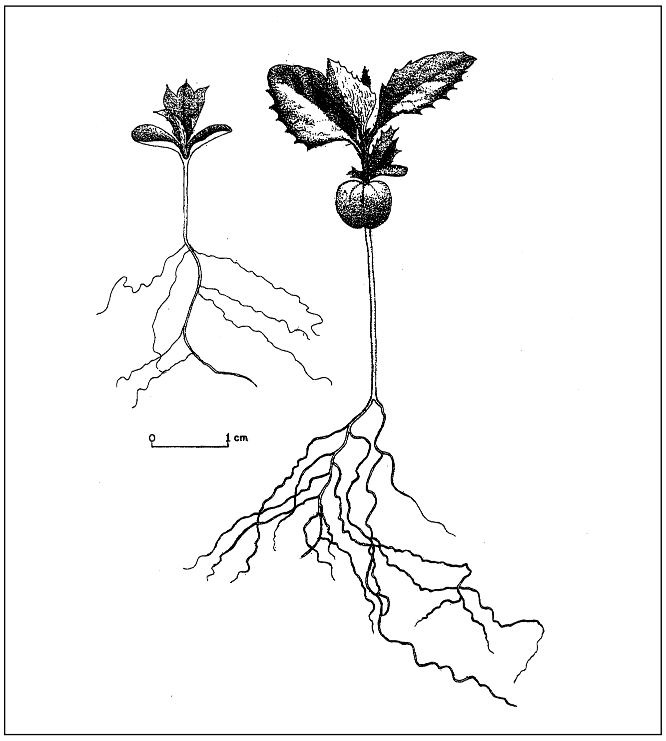


Figure 3—*Heteromeles arbutifolia*, Christmasberry: young seedling (**left**) and older seedling (**right**).



ture but did not give any data on viability retention under these conditions.

Germination and seed testing. Christmasberry seeds are reported to be nondormant at dispersal (Emery 1988; Keeley 1987; Magill 1974), whereas seeds that have been stored are rendered secondarily dormant and require 3 months of chilling at 3 to 5 °C in order to germinate. Under field conditions, Christmasberry seeds germinate within a few months of dispersal and do not form a persistent seedbank (Parker and Kelly 1989). Germination is epigeal (figure 3). Recruitment of new individuals is rarely observed. Although winter seedling emergence is a common occurrence, the seedlings almost invariably die, either from herbivory or summer drought (Parker and Kelly 1989). Because of the transient seed bank, there can be no recruitment after fire, and new recruitment is in fact restricted to chaparral stands that have been free of fire for at least 50 years (Keeley 1992). The seedlings are not very drought-tolerant and seem to need the shade and the deep litter that develops under adult shrub canopies in old stands in order to survive.

Recently harvested lots of Christmasberry seeds that are well-cleaned to remove unfilled seeds generally have high fill and high viability. Keeley (1987) reported germination of 99% at 23 °C. Such lots should be relatively easy to evaluate, either with a germination test or with tetrazolium staining. A 3-month chill at 5 °C followed by a germination test of 28 days at 20 or 25 °C should give maximum germination. Christmasberry seeds have no endosperm, and the embryo completely fills the seed cavity (figure 2). A procedure similar to that used for apple (*Malus* spp.) or bitterbrush (*Purshia* spp.) can be used for tetrazolium evaluation. The seeds should be soaked in water overnight, then clipped at the cotyledon end. The embryos can then be gently squeezed out, immersed in 1% tetrazolium chloride for 6 hours at room temperature, and examined for staining patterns. Older seedlots that have begun to lose viability and germinate sporadically will probably also have ambiguous tetrazolium staining patterns.

Field seeding and nursery practice. Christmasberry would probably be difficult to direct-seed in a wildland setting because of its establishment requirements (Keeley 1992). The seedlings require shady, moist conditions and deep litter, so they would have difficulty getting established on the open disturbances that characterize most wildland seeding projects. Christmasberry is easily propagated from seeds in a nursery setting, however. The seeds maybe planted in flats in sand or soil. If freshly harvested seeds are used, no pretreatment is necessary, and seedlings emerge over a period of 10 to 40 days (Magill 1974). Emergence of 73% has been reported in one case. The seeds may also be planted outdoors in nursery beds. Chilling before spring-planting is recommended (Magill 1974). Greever (1979) reported 100% emergence from March sowing in sand and that there was little difference in seedling size between December and March sowings by May. Propagation by grafting or cuttings is also practiced (Greever 1979; Magill 1974).

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H

Elaeagnaceae—Oleaster family

Hippophae rhamnoides L.

common seabuckthorn

Richard T. Busing and Paul E. Slabaugh

Dr. Busing is an ecologist at the USDI Geographical Survey, Corvallis, Oregon; Dr. Slabaugh (deceased) retired from the USDA Forest Service's Rocky Mountain Forest and Range Experiment Station

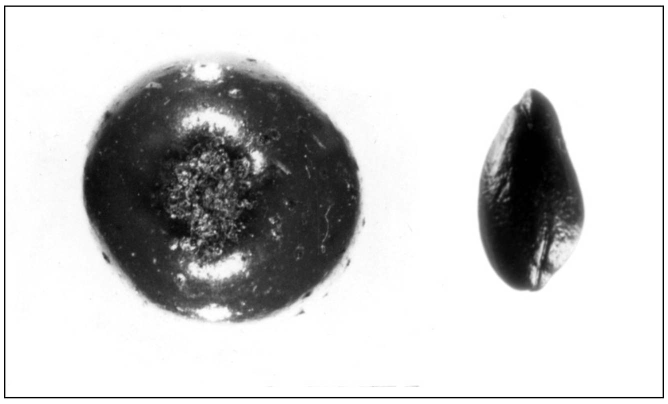
Other common names. Sandthorn, swallow-thorn.

Growth habit, occurrence, and use. Common seabuckthorn—*Hippophae rhamnoides* L.—is native to northwestern Europe through central Asia to the Altai Mountains, western and northern China, and the northern Himalayas. Of the 2 species in the genus, only common seabuckthorn is widely cultivated (Rehder 1940). A very hardy deciduous shrub or a small tree, common seabuckthorn is used primarily for ornamental purposes. In Europe and Asia, it is used to form hedges and, because of its nitrogen-fixing symbionts, serves to enrich and protect soils (Bogdon and Untaru 1967; Kao 1964; Stewart and Pearson 1967). A tendency to form thickets by root suckering limits its use in shelterbelts. In Asia, the plant has a variety of medicinal uses (Ma 1989). The berries, which are a rich source of vitamins (Stocker 1948; Valicek 1978; Zhmyrko and others 1978), have been used in making a cordial and jam in Siberia (Hansen 1931). The plant stems bear many sharp, stout thorns and provide protection, cover, and food for various birds and small rodents (Hansen 1931; Pearson and Rogers 1962).

Flowering and fruiting. The species is dioecious; its very small, yellowish, pistillate flowers appear in March or April before the leaves (Pearson and Rogers 1962; Slabaugh 1974). Orange-yellow, drupelike acidic fruits about the size of a pea (figure 1) (Rehder 1940) ripen in September or October (Hoag 1965; Hottes 1952) and frequently persist on the shrubs until the following March. Each fruit contains a bony, ovoid seed (figures 1 and 2). Seedcrops are borne annually.

Collection, extraction, and storage. Common seabuckthorn fruits are soft and cling tenaciously to the brittle twigs (Demenko and others 1983). Fruits may be picked from the bushes at any time between late fall and early spring. However, germination may vary with the time that seeds were extracted from the ripe fruits (Eliseev and Mishulina 1972). Seeds may be extracted by running the wet fruits through a macerator and floating off the pulp. Prompt cleaning and drying is advantageous because germination rate is very low for seeds left too long in the fruits (Eliseev and Mishulina 1977; Rohmeder 1942). From 45 kg (100 lb) of fruits, 4.5 to 14 kg (10 to 30 lb) of cleaned seeds may be extracted. Soundness of 85% and purity of 97% have been reported (Slabaugh 1974). The average number of cleaned seeds determined on 10 samples is 88,000/kg (40,000/lb), with a range of 55,000 to 130,000/kg (25,000 to 59,000/lb) (Slabaugh 1974). Smaller seeds, numbering 258,000 to 264,500/kg (117,000 to 120,000/lb), were reported in Romania (Enescu and Stegaroiu 1954). The seeds are orthodox and store easily at low moisture contents and temperatures. Dry seeds have been kept satisfactorily for 1 to 2 years at room temperature (Slabaugh 1974). Viabilty of 60% has been reported for seeds stored 4 to 5 years (Smirnova and Tikhomirova 1980).

Figure 1—*Hippophae rhamnoides*, common seabuckthorn: fruit and seed.



Germination. Internal dormancy in seeds of seabuckthorn can be broken by stratification in moist sand for 90 days at 2 to 5 °C (Cram and others 1960; Pearson and Rogers 1962). Stratification for 15 days is sufficient if seeds are sown in the autumn (Grover and others 1962). Germination tests may be run in 40 days on stratified seeds

Figure 2—*Hippophae rhamnoides*, common seabuckthorn: longitudinal section through a seed.

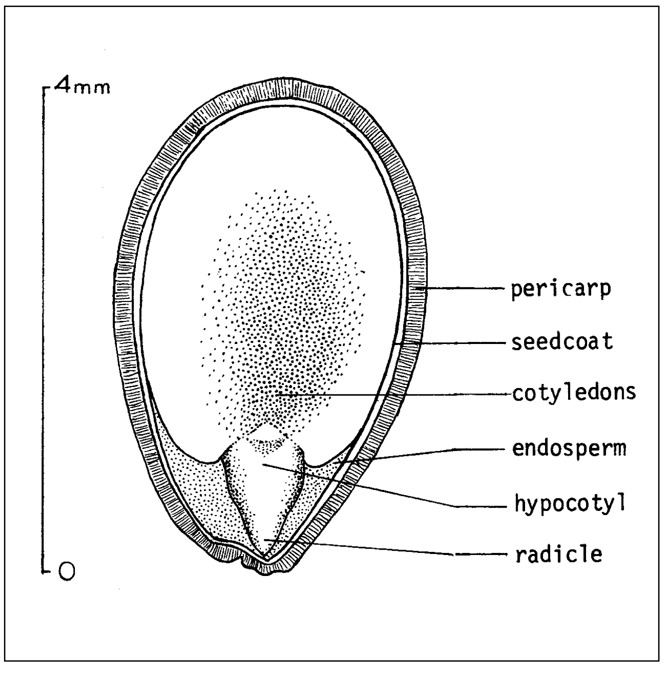
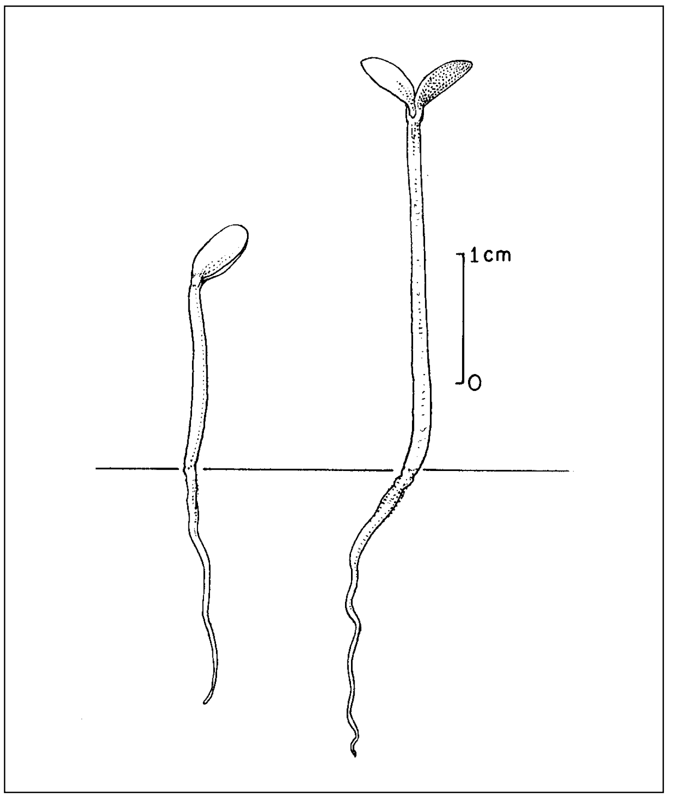


Figure 3—*Hippophae rhamnoides*, common seabuckthorn: seedling development at 1 and 7 days from germination.



in sand flats at diurnally alternating temperatures of 20 and 30 °C (Slabaugh 1974). Germination was increased slightly by exposure to light intensities up to 2,150 lumens/m^2 (Pearson and Rogers 1962). Soaking seeds in solutions of gibberellic acid, sulfuric acid, or other compounds, such as potassium iodide (KI), zinc sulfate ($ZnSO_4$), manganese sulfate ($MnSO_4$), or cobalt sulfate($CoSO_4$), may also increase germination (Avanzanto and others 1987; Eliseev and Mishulina 1972). Germination of untreated seeds ranged from only 6 to 60% after 60 days (Slabaugh 1974). Tests in Romania and England gave results of 75 to 85% and 95 to 100% (Enescu and Stegaroiu 1954; Pearson and Rogers 1962). Germination is epigeal (figure 3).

Nursery practice. Untreated seeds may be used for fall-sowing (Grover and others 1962), but stratified seeds are needed for spring-sowing (Cram and others 1960). Either broadcast or drill sowing is satisfactory if seeds are covered with about 6 mm ($^1/_4$ in) of soil. Shading during the early stages of germination is beneficial (Hansen 1927). This species can be propagated by layers, suckers, and root cuttings as well as by seeds (Avanzanto and others 1987; Papp 1982, Varga and Foldesi 1985). It grows best on moist, neutral to basic, sandy soils (Pearson and Rogers 1962).

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Rosaceae—Rose family

Holodiscus (K. Koch) Maxim.

ocean-spray

Nancy L. Shaw, Emerenciana G. Hurd, and Peter F. Stickney

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

Growth habit, occurrence, and use. *Holodiscus* is a taxonomically complex genus including about 6 species of western North America and northern South America (Hitchcock and others 1961; Ley 1943). The 2 generally recognized North American species (table 1)—creambush ocean-spray and gland ocean-spray—are deciduous, multi-stemmed shrubs with simple, alternate, deciduous, toothed to shallowly lobed, exstipulate leaves.

Creambush ocean-spray grows from 1 to 6 m in height, with slender, arching branches and grayish red exfoliating bark. It is a prolific root sprouter, capable of recovering from fire, grazing, or mechanical damage by resprouting from perennating buds in the root crown. Growing at elevations from sea level to 2,150 m, it is most abundant in coastal areas from British Columbia to southwestern California. It also occurs eastward to Montana in drier conifer types of the interior Pacific Northwest. A dominant shrub in a number of forested communities, creambush ocean-spray is also common in riparian areas and on rocky talus slopes (Halversen and others 1986; Topik and others 1986). Remnant stands are found on higher peaks of Great Basin mountain ranges (Hitchcock and others 1961; USDA FS 1937).

Gland ocean-spray is a low, intricately branched shrub that is 0.1 to 3 m tall (Harrington 1954). It differs from creambush ocean-spray in its more compact growth habit, leaves with decurrent petioles, and leaf lobes or teeth without secondary teeth. Gland ocean-spray grows east of the Cascade Mountains and the Sierra Nevada, from north central Oregon to Chihuahua, Mexico, at elevations ranging from 1,400 to 3,350 m (Harrington 1954; Mozingo 1987; USDA FS 1937). Although gland ocean-spray is found in a variety of plant communities, its most characteristic habitats are talus slopes, rock outcrops, slickrock plateaus, and dry, rocky desert areas.

Palatability and forage value of both ocean-spray species vary geographically but are generally low for livestock and big game. However, in the absence of more palatable shrubs, substantial quantities are browsed by deer (*Odocoileus* spp.) and by elk (*Cervus elaphus*) on low-elevation winter ranges. In some areas, ocean-sprays are important year-round (USDA FS 1937). Both shrubs may increase on summer ranges where other forage species are browsed preferentially (Ferguson 1983). Gland ocean-spray is browsed in summer by bighorn sheep (*Ovis canadensis*) and both species are browsed by rabbits (Sutton and Johnson 1974; Todd 1975; Van Dersal 1938).

Ocean-spray has considerable potential for revegetating a variety of disturbed areas. Populations capable of growing on dry, rocky, well-drained sites may be particularly useful

Table 1—*Holodiscus,* ocean spray: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
H. discolor **(Pursh) Maxim.** *Spiraea discolor* Pursh *Spiraea ariaefolia* Smith in Rees *Schizonotus discolor* Raf. *Sericotheca discolor* Rydb.	**creambush ocean-spray,** creambush, creambush rockspirea, hardhack, Indian arrow-wood, ocean-spray	S British Columbia, Washington, Oregon, W Montana, N Idaho, NE Nevada, & California
H. dumosus **(Nutt.) Heller** *Spiraea dumosa* Nutt. ex T. & G. *Spiraea discolor* var. *dumosa* Wats. *Schizonotus dumosus* Koehne. *Sericotheca dumosus* Rydb.	**gland ocean-spray,** bush ocean-spray, bush rockspirea, mountain-spray, rock-spirea, creambush	E & S Oregon, N central & S Idaho, NE California, Nevada, Utah, W & S Wyoming, Colorado, Arizona, New Mexico, & S to Chihuahua, Mexico

Sources: Archer (2000), Flessner and others (1991), Hitchcock and others (1971), Ley (1943), McMurray (1987b).

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(Stark 1966; Sutton and Johnson 1974). Ocean-spray has been recommended for use in nonintensive highway plantings, riparian areas, windbreaks, erosion control projects, wildlife habitat improvement projects, and conservation plantings (Antieau 1987; Atthowe 1993; Flessner and others 1992). Because of their growth habits, showy inflorescences, and fall coloration, both species are attractive ornamentals. Creambush ocean-spray was first cultivated in 1827 and gland ocean-spray in 1853 (Rehder 1940).

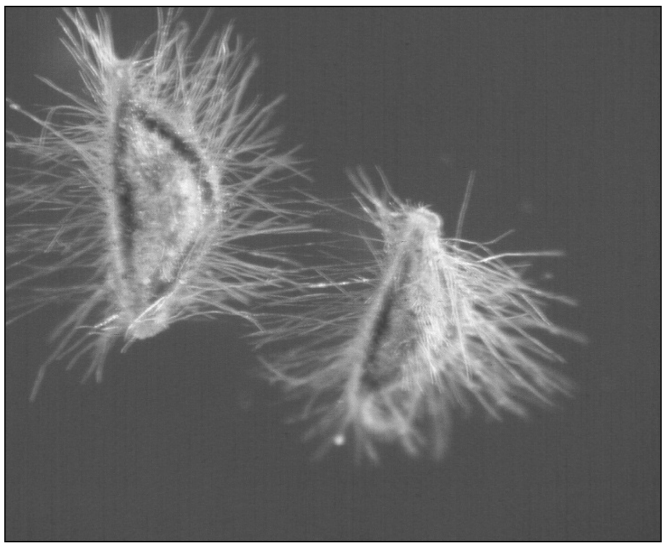
Native Americans made digging sticks and arrow shafts from the hard wood and straight branches of ocean-spray (Anderson and Holmgren 1969; Daubenmire 1970; Hopkins and Kovalchik 1983). Fruits of gland ocean-spray were eaten by Native Americans of the Great Basin, and pioneers made nails from its wood.

Both North American ocean-sprays are tetraploid, with 2X = n = 18 (Antieau 1986; Goldblatt 1979; McArthur and Sanderson 1985), and both exhibit considerable morphological variation. A genetic basis for variability in such characteristics as growth habit, growth rate, leaf morphology, and flower abundance in creambush ocean-spray is suggested by common garden studies (Flessner and others 1992).

Flowering and fruiting. Although the showy terminal panicles and floral buds of both species develop in early spring, flowering is delayed until late spring to mid-summer. Fruits ripen in late summer and are dispersed by wind and gravity through November (Hitchcock and others 1961; Stickney 1974) (table 2). The insect-pollinated flowers are small, creamy-white, perfect, and perigynous (Hitchcock and others 1961; McArthur 1984). The entire disk lining the hypanthium gives the genus its name (Greek: *holo* = whole and *diskos* = disk). Each flower produces 5 villous, light-yellow achenes that are about 2 mm long (figures 1 and 2). Seeds are broadly oblong and contain a thin endosperm and an embryo with ovate cotyledons (figure 2) (Ley 1943).

Collection, cleaning, and storage. Ocean-spray achenes are among the smallest of shrub fruits. Estimates of the number of cleaned achenes per weight exceed 11,000,000/kg (5,000,000/lb) for each species (King 1947; Link 1993). Achene collection is tedious, and supplies are rare and costly. In addition, the achenes are difficult to handle because of their pubescence and small size. Achenes are hand-stripped from inflorescences in late summer or autumn (table 2) (Monsen 1996). Large debris in air-dried collections can be removed with a fanning mill. Small lots may be cleaned by hand-rubbing and sieving (Link 1993).

Figure 1—*Holodiscus*, ocean spray: achenes of *H. discolor*, creambush ocean-spray (**left**) and *H. dumosus*, gland ocean-spray (**right**).



Sound achenes are identified by examining imbibed achenes through a dissecting microscope for the presence of an embryo. Using this method, King (1947) found that only 7% of ocean-spray achenes collected were sound. In creambush ocean-spray from British Columbia, viability was greater for achenes collected in October or November than for those collected in August or September (Marchant and Sherlock 1984).

Storage requirements for ocean-spray have not been examined. The achenes appear to be orthodox in storage behavior and can probably be stored for several years at low water contents and temperatures.

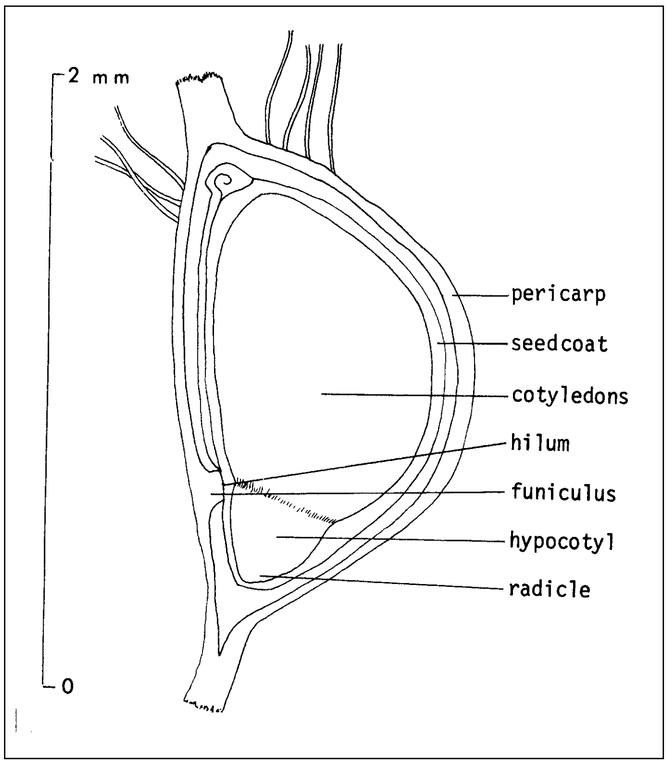
Germination. There are no official testing prescriptions for this genus. Germination of creambush ocean-spray seeds is enhanced by wet prechilling at 2 to 5 °C for 15 to 18 weeks (King 1947; Marchant and Sherlock 1984). King

Table 2—*Holodiscus*, ocean-spray: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening	Seed dispersal
H. discolor	California	May–Aug	—	—
	N Idaho	July	Aug	—
	N Idaho	July	Late July–early Sept	Aug–Nov
H. dumosus	Great Basin	June–Aug	—	—
	Utah	June–Aug	—	—
	—	—	Aug	Aug

Sources: Drew (1967), Jorgensen (2004), Mozingo (1987), Munz and Keck (1973), Orme and Leege (1980), Welsh and others (1987).

Figure 2—*Holodiscus discolor*, creambush ocean-spray: longitudinal section through an achene.



(1947) obtained 84% germination in 22 days when seeds were chilled for 18 weeks before incubation at 20 to 24 °C.

Germination of gland ocean-spray has received little study. Link (1993) reported that 16 weeks of wet chilling failed to release dormancy in this species. Effective treatments have not been reported.

Viability of ocean-spray seeds may be tested by tetrazolium chloride staining. After 3 hours of imbibition in water at room temperature, seeds are excised from the achene and the seedcoat is pricked or slit near the center of the seed. Seeds are then allowed to imbibe a 1% tetrazolium chloride for 4 hours at room temperature. Stained embryos may be read in place, as the seedcoat is very thin (Hurd 1996; King 1947). Staining should be evaluated as described by Peters (2002) for Rosaceae III genera.

Nursery practice. Ocean-sprays may be propagated as bareroot or container stock (Everett 1957). Achenes should be fall-sown or artificially prechilled and spring-sown in bareroot nurseries (Flessner and others 1992). Marchant and Sherlock (1984) obtained successful plantings only by planting freshly harvested achenes in fall. Cleaned achenes of both species can be drilled at reasonably uniform spacings within rows (Shaw and Monsen 2004). They may also be broadcast and covered by dragging a lightweight chain over the seedbed. Seedlings develop slowly and may be lifted as 1+0 or 2+0 stock, depending upon size specifications and growing conditions.

Container seedlings are propagated by planting several wet-prechilled achenes in each container and thinning or by planting germinants. Kruckeberg (1982) reported that ocean-spray can be propagated by fall-sowing achenes in boxes outdoors and covering them lightly with soil. Flessner and others (1992) planted wet prechilled (4 months at 4 °C) creambush ocean-spray achenes in shallow flats in a greenhouse. Seedlings emerged after 16 to 30 days of incubation at a minimum temperature of 21 °C. Developing seedlings were fertilized and treated with a fungicide as necessary. After 2 months they were transferred to larger containers in a lathhouse and held overwinter for planting as 1+0 stock.

Kruckeberg (1982) reported that creambush ocean-spray planting stock is easily obtained by potting wildlings, which are often abundant. Morgan and Neuenschwander (1988) observed high densities of creambush ocean-spray wildlings following severe burns, but Wright and others (1979) and Stickney (1996) concluded that the species exhibits poor seedling regeneration following fire in sagebrush (*Artemesia* spp.) and conifer communities of the intermountain and northern Rocky Mountain regions.

Ocean-spray can be grown from cuttings, but rooting of both species varies widely among clones, cutting types, and propagation techniques (Antieau 1987; Link 1993). Softwood cuttings may be treated with rooting hormones and propagated in a greenhouse with a mist system (Antieau 1987; Marchant and Sherlock 1984). Success with semi-hardwood cuttings is variable (Everett and others 1978; Kruckeberg 1982). Fall-harvested hardwood cuttings are cut to 15-cm (6-in) lengths and treated with 0.8% indole-3-butyric acid (IBA) powder and a fungicide (Macdonald 1986). Hardwood cuttings stored in straw-bale bins or cold frames will develop calluses (Macdonald 1986; Marchant and Sherlock 1984). When fall-planted, these cuttings root rapidly. Layers and suckers have also been propagated successfully (Kruckeberg 1982).

Field practice. Fresh achenes broadcast over a rough seedbed in fall are covered by natural soil sloughing (Shaw and Monsen 2004; Van Dersal 1938). Achenes may be mixed with seeds of other shrub species, but they should not be sown with more competitive grasses or forbs. Planting areas should be selected carefully to make the best use of seed supplies, as seeding results are often erratic. Native creambush ocean-spray seedlings develop slowly and are poor competitors (Wright and others 1979).

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Creambush ocean-spray can be established by transplanting. Youtie (1992) reported good survival of rooted cuttings on biscuit scablands in Oregon's Columbia River Gorge. Marchant and Sherlock (1984) found that planted seedlings grew slowly the first year. Low survival on western Montana roadcuts was attributed to poor soils and unhealthy planting stock (Hungerford 1984).

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H

Fabaceae—Pea family

Hymenaea courbaril L.

courbaril

J. A. Vozzo

Dr. Vozzo is a research plant physiologist at the USDA Forest Service's Southern Research Station Mississippi State, Mississippi

Other common names. jutaby, *cuapinol*, *algarrobo*.

Occurrence and growth habit. Courbaril—*Hymenaea courbaril* L.—is a large tree, about 45 m tall, relatively slow growing (about 1 m/year) with a well-formed clean trunk. It develops best on sandy, drained ridges and river banks (but not well in wetlands) and is normally found in open sites from southern Mexico, Central America, and the West Indies, to northern South America. It is found in a variety of soils, such as clay to sand, occurring predominantly in oxisols, with a pH range from 4.8 to 6.8. It grows best in areas with 1,900 to 2,150 mm of rainfall, and from sea level to about 900 m. It coppices well in cut-over areas except from large stumps and can also be propagated from cuttings. Courbaril is the most widespread of the 17 species in the genus *Hymenaea*; there is an African species and the remaining species are found in neotropical America. Courbaril readily forms forest associations in semi-deciduous, secondary, moist subtropics (Rzedowski 1981). It is also reported in nearly pure stands in Mexico (Weaver 1987).

Use. Courbaril's basic use is for timber. The wood is strong, hard, and tough; it is difficult to saw, machine, and carve but bends well after steaming. It is commercially useful for flooring, handles, sporting equipment, furniture, and railroad ties (Chudnoff 1984). Its heartwood has a specific gravity of about 0.70 g/cm^3. Although courbaril wood is resistant to white-rot fungi (less to brown-rot) and termites, it has little resistance to marine borers. It does not weather well and does require painting (Francis 1990; Longwood 1962). The tree has limited ornamental use for shade, parks, and streets because of its heavy legumes (pods) and the offensive odor of the broken legumes as seeds mature. Although it has a limited appeal, the seed pulp is a good dietetic source of sugar and high in fiber when eaten plain or toasted or drunk as a fermented beverage. It is also given to livestock. According to local folk medicine, a bark infusion is used as a laxative and the fruit pulp as an antidiarrheal (Liogier 1978).

Flowering and fruiting. Large trees with full, overhead light usually flower during spring and summer. Terminal racemes bear white flowers about 4 cm wide. Mature legumes (figure 1) measure 5 to 10 cm long, 2 to 3.5 cm wide, and 2.5 cm thick and fall during the following spring. The thick, hard legume does not open naturally, but protects 3 or 4 large seeds (figures 2 and 3) encased in a powdery, cream-colored pulp (Liogier 1978). Small animals—such as agouties (*Tayassu* spp.) and peccaries (*Dasyprocta* spp.)—open the legumes to eat the pulp and seeds. Legumes have a protective gum that delays rotting for several months, until the seeds begin to take up moisture for germination; otherwise the seeds would rot in their legumes (Jansen 1983).

Collection and storage. Seeds collected in Puerto Rico average about 253/kg (115/lb) (Francis 1990), whereas those collected in Brazil yield 475/kg (215/lb) (Pereira 1982). A single tree may produce 100 legumes per year but not necessarily each year. Because of the height of the trees, the legumes are usually picked manually from the ground, and seeds are obtained from fresh legumes that have fallen in spring (Jansen 1983). After-ripening causes an actual

Figure 1—*Hymenaea courbaril*, courbaril: legume.

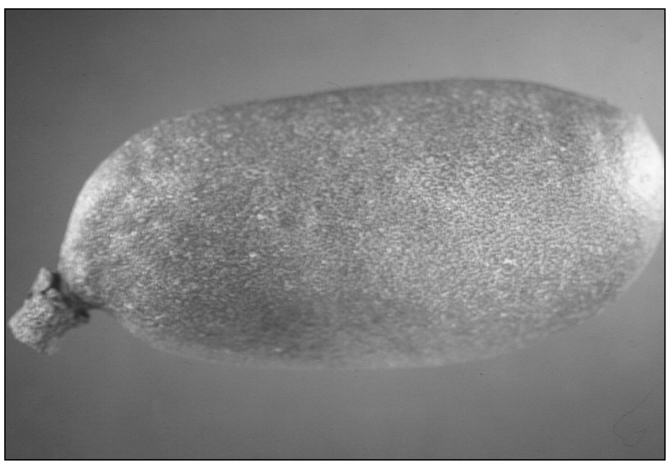

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Figure 2—*Hymenaea courbaril*, courbaril: seeds.

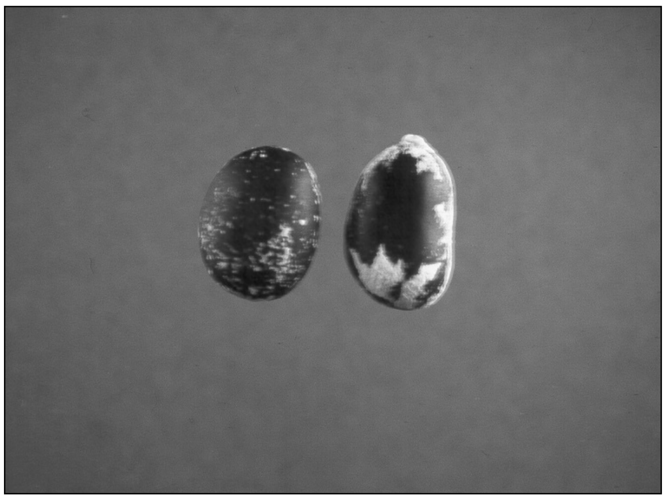
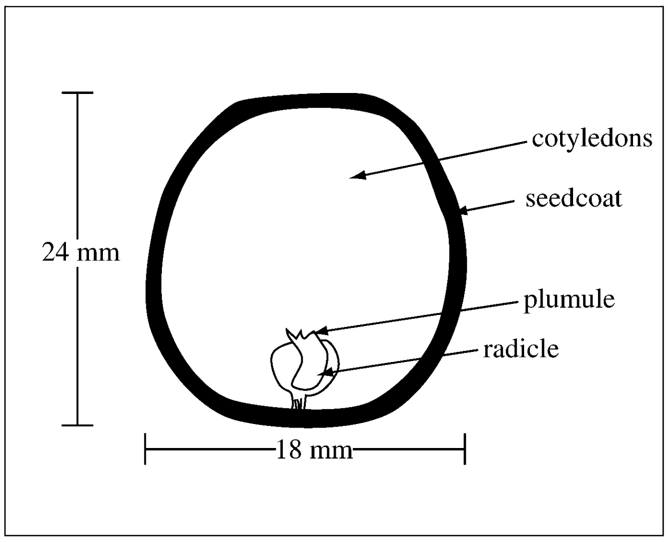


Figure 3—*Hymenaea courbaril*, courbaril: longitudinal section through a seed.



germination enhancement during the first 4 months after collection. This may also account for the long (9-month) period seeds remain in the legume on the tree before falling. Courbaril seeds are orthodox in storage behavior and store relatively well with acceptable germination for periods in excess of 1 year. However, the conditions for optimal storage changes with time. For the first year, sealed containers are preferable at ambient temperatures; after that, seeds should be refrigerated or kept in unsealed bags (Marrero 1943).

Germination. Simple scarification or an hour of soaking in sulfuric acid is necessary as a germination pretreatment (Marshall 1939). After imbibition, seeds may be germinated in potting mix for 14 to 21 days with up to 90% germination (Francis and Rodriquez 1993; Marrero 1949). Seeds can be germinated at ambient temperature in either potting mixture or sand placed in shallow trays or moistened filter or blotter paper in petri dishes.

Nursery practice. Container stock may be grown in either full sun or 50% shade. However, seedlings grown in full sun are ready for outplanting about 2 weeks earlier than seedlings grown in shade (Francis 1990; Pereira 1982). Although courbaril may be direct-seeded or underplanted, success is greater with containers unless seeds can be given greater protection. A large taproot with a well-developed fibrous net grows deeply and may or may not have associated nitrogen-fixing nodules (Allen and Allen 1981). Seeds may be infected by a bruchid beetle, *Pygiopachymerus* sp. (Decelle 1979); a weevil, *Rhinochenus* sp. (Jansen 1975); and an ant, *Atta* sp. (Jansen 1983).

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I

Aquifoliaceae—Holly family

Ilex L.

holly

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 hollies—genus *Ilex*—include almost 400 species of deciduous or evergreen shrubs and trees that occur in temperate and tropical regions of both hemispheres (Brown and Kirkman 1990). About 20 species are native to eastern North America. Of the 7 species included in this book (table 1), most are highly valued for ornamental plantings and all are good food sources for wildlife. More than a thousand cultivars of American holly have been selected for their ornamental features (Grelen 1990). This species also hybridizes with dahoon (*Ilex cassine* L.) to produce Topel holly (*I.* × *attenuata* Ashe) (Little 1979). The wood of American holly is also used in cabinetry and for construction of novelties and specialized wood products (Vines 1960).

Flowering and fruiting. The small, axillary, white or greenish white, dioecious flowers appear in the spring on the current season's growth (table 2). Holly fruits are rounded, berrylike drupes that range from 6 to 13 mm in diameter (figure 1). Each fruit contains 2 to 9 bony, flattened seeds that are botanically defined as nutlets, or pyrenes (figure 2). The fruits mature in the fall (table 2), turning from green to various shades of red, yellow, or black (table 3). The seeds contain a very small embryo in a fleshy endosperm (figure 3).

Collection, extraction, and storage. Ripe fruits may be picked by hand or flailed from the branches onto sheets spread on the ground. Seeds should be extracted by running the fruits through a macerator with water and floating or skimming off the pulp and empty seeds. For small seedlots, kitchen or laboratory blenders do a thorough job, although replacing the metal blades with plastic tubing propellers has been recommended to avoid damage to the seeds (Munson 1986). In another method, the fruits are fermented in warm water, then rubbed over a screen by hand to remove the pulp (Vines 1960). Seed yield data are summarized in table 4.

If the seeds are to be stratified immediately, drying is not necessary. If the seeds are to be stored, they should be dried to about 10% moisture content, placed in moisture-proof containers, and stored at temperatures near or below freezing. Viability of seeds after storage for more than 1 year has not been reported, but hollies are orthodox in storage behavior and should keep well at temperatures a few degrees above (or below) freezing. Storage foods in the embryo are primarily lipids and proteins (Hu and others 1979).

Table 1—*Ilex,* holly: nomenclature and occurrence

Scientific name & synonym	Common name(s)	Occurence
***I. aquifolium* L.**	**English holly**	Native to S Europe, N Africa, & W Asia to China; widely planted in SE & NW US
***I. decidua* Walt.**	**possumhaw,** deciduous holly, winterberry, swamp holly	Maryland to Florida, W to Texas, Missouri, & Illinois
***I. glabra* (L.) Gray** *I. monticola* Gray	**inkberry,** gallberry, smooth gallberry	Nova Scotia to Florida, W to Missouri & Texas
***I. montana* Torr. & Gray ex**	**Gray mountain holly,** mountain winterberry	New York to Florida, W to Louisiana
***I. opaca* Ait.**	**American holly,** holly, white holly	Massachusetts to Florida, W to Texas & Missouri
***I. verticillata* (L.) Gray**	**common winterberry,** winterberry, black alder	Newfoundland to Minnesota, S to Louisiana & Florida
***I. vomitoria* Ait.**	**yaupon,** cassena, Christmas-berry, evergreen holly	Virginia to central Florida, W to Texas & Oklahoma

Source: Little (1979).

I

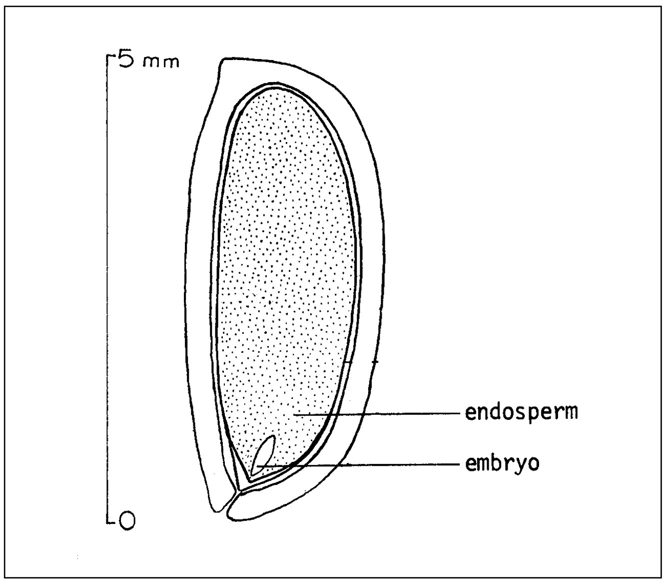
Table 2—*Ilex, holly:* phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening	Seed dispersal
I. aquifolium	—	May–June	Sept	Winter–spring
I. decidua	Gulf Coastal Plain	Mar–May	Fall	Winter–spring
I. glabra	—	Mar–June	Fall	Winter–spring
I. montana	Appalachian Mtns	May–June	Sept	—
I. opaca	—	Apr–June	Sept–Oct	Mar
I. verticillata	—	June–July	Sept–Oct	Fall–winter
I. vomitoria	Gulf Coastal Plain	Apr–May	Sept–Oct	Dec

Sources: Bonner (1974), Halls (1973), Little and Delisle (1962), Stupka (1964), Vines (1960).

Figure 1—*Ilex*, holly: fruits and leaves of *I. opaca,* American holly (**top**) and *I. vomitoria*, yaupon (**bottom**).

Figure 3—*Ilex montana*, mountain holly: longitudinal section of a nutlet.



Pregermination treatment. Holly seeds exhibit a deep dormancy that is caused partly by the hard endocarp surrounding the seedcoat (figure 3) and partly by an immature embryo. In nature, germination of American holly is commonly delayed for as long as 3 years (Bonner 1974). This condition suggests that alternating warm and cold moist treatments may be the best approach. Reasonable germination of American holly has been reported after 12 months of warm treatment that is followed by 3 months of

Figure 2—*Ilex*, holly: nutlets (pyrenes) of *I. aquifolium*, English holly (**top**); *I. montana*, mountain holly (**second**); *I. opaca*, American holly (**third**); *I. verticillata,* common winterberry (**fourth**); *I. vomitoria*, yaupon (**fifth**); and *I. glabra*, inkberry (**bottom**).

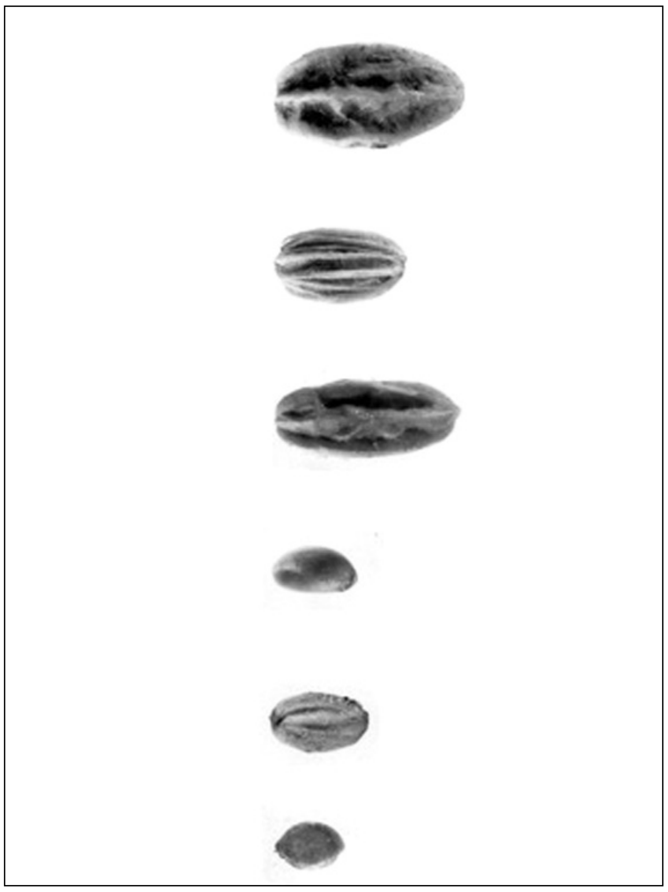


Table 3—*Ilex,* holly: height, seed-bearing age, and color of ripe fruit

Species	Height at maturity (m)	Year first cultivated	Minimum seed-bearing age (yrs)	Color of ripe fruit
I. aquifolium	15–24	Ancient times	5–12	Light red
I. decidua	6–9	—	—	Red, orange-red
I. glabra	4	1759	—	Black
I. montana	12	1870	—	Orange-red, rarely yellow
I. opaca	30	1744	5	Red, rarely orange or yellow
I. verticillata	8	1736	—	Red, orange or yellow
I. vomitoria	3–8	—	4–7	Red

Sources: Bonner (1974), Brown and Kirkman (1990), Grelen and Duvall (1966), Halls (1973), Little and Delisle (1962), Maisenhelder (1958), Rehder (1962), Vines (1960).

Table 4—*Ilex,* holly: seed yield data

	Cleaned seeds/weight				
	Range		Average		
Species	/kg	/lb	/kg	/lb	Samples
I. aquifolium	—	—	125,700	57,000	1
I. decidua	—	—	43,6000	19,800	1
I. glabra	—	—	63,900	29,000	1
I. montana	—	—	77,200	35,000	1
I. opaca	48,500–80,150	22,000–36,350	62,700	28,430	4
I. verticillata	88,200–284,450	40,000–129,000	202,860	92,000	4
I. vomitoria	—	—	83,350	37,800	1

Sources: Bonner (1974), Swingle (1939).

cold (Dirr and Heuser 1987). For common winterberry, which may have a more permeable endocarp than other hollies, some benefit may be obtained by stratifying seeds at alternating temperatures of 20 °C (night) and 30 °C (day) for 60 days, followed by 60 days at 5 °C (Giersbach and Crocker 1929).

Germination and viability tests. Because of the extremely slow germination of hollies, there is no satisfactory method for testing germination directly. Germination of 70 to 95% has been reported for inkberry in tests that ran over 300 days (Hughes 1964), and 33 to 56% for American holly in tests that ran 2 $^1/_2$ years (Barton and Thornton 1947). Test periods of this length are not practical, and indirect estimates of viability are commonly used in place of germination tests. Cutting tests give good estimates of viability for freshly collected seeds, but for most purposes, tetrazolium staining is best. Procedures recommended for English holly by the International Seed Testing Association (1993) should work well with other holly species. Seeds should be cut longitudinally through the seedcoat and into the endosperm, or cut transversely at distal or both ends into the endosperm, to allow entry of the tetrazolium solution. Incubation for 24 hours at 30 °C in a 1.0% solution should be sufficient for staining. All tissues, including the endosperm, should be fully stained in viable seeds.

Nursery practice. Holly seeds may be broadcast or sown in drills in fall or spring. Sowing immediately after collection has been recommended for American holly and inkberry (Afanasiev 1942; Hartmann and Kester 1968), but germination should not be expected until the second or even third spring (Bonner 1974). Seeds should be covered with 3 to 13 mm ($^1/_8$ to $^1/_2$ in) of soil, and fall-sown beds should be mulched (Bonner 1974; Muir 1965). In another recommended procedure, seeds are sown in a flat of moist medium that is then covered with a plastic bag and placed in a warm (15 to 27 °C) shaded room until seedlings start to emerge. When this occurs, the bag should be removed and the flat moved to a spot with normal germination conditions (Dirr and Heuser 1987). Half-shade is recommended for beds of English holly during the first 2 summers, and field planting should be with 2+2, 2+3, or 2+2+2 stock (Bonner 1974). Because of the extreme dormancy in holly seeds, most propagation is by rooted cuttings, especially for ornamental varieties and selections. All species do not root equally or with the same treatments, so a good manual on vegetative propagation should be consulted (Dirr and Heuser 1987). A considerable amount of research on embryo culture of several holly species has also taken place (Hu 1975, 1977).

I

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Juglandaceae—Walnut family

Juglans L.

walnut

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 walnuts include about 20 species of deciduous trees or large shrubs that occur in the temperate regions of North America, northwestern South America, northeastern Europe, and eastern Asia. Six are native to the United States, and 2 exotic species are also planted in this country (table 1). The wood of most species is used to some extent, and that of many species, primarily black walnut, is highly valued for furniture, cabinet work, gunstocks, and interior trim. The nuts provide food for humans as well as for wildlife, and ground shells are used as an abrasive grit for industrial cleaning. Numerous medicinal products and dyes have been made from extracts of walnut fruits (Krochmal and Krochmal 1982). English walnut is a major nut crop in many temperate regions around the world, including the United States. Of the 6 native species, black walnut is by far the most widely planted. Butternut, little walnut, and Hinds walnut have had limited utilization. Butternut is currently being killed throughout its range in North America by *Sirococcus clavigignenti–juglandacearum* Naiv. Kostichka & Kuntz, a fungus of unknown origin (Ostry and others 1994). Research is underway to identify and propagate resistant trees.

Geographic races. There is considerable genetic variation in the walnuts that are widely distributed. Three distinct geographic races of English walnut are recognized: Turkestani, Himalayan, and Central Asian—and many horti-

Table 1—*Juglans,* walnut: nomenclature and occurrence

Scientific name & synonym(s)	Common name(s)	Occurrence
***J. ailantifolia* Carriere** *J. sieboldiana* maxim.	**Japanese walnut,** Siebold walnut	Japan
***J. californica* S.Wats.**	**California walnut,** southern California walnut, black walnut	Coastal S California (Santa Barbara Co. to Orange Co.) California
***J. cinerea* L.** *Wallia cinerea* (L.) Alef.	**butternut,** oilnut, white walnut	New Brunswick to S Ontario & SE Minnesota, S to Arkansas,N Mississippi, N Georgia, & W South Carolina
***J. hindsii* (Jepson) Jepson ex R.E. Sm.** *J. californica* var. *hindsii* Jepson	**Hinds walnut,** northern California walnut, Hinds black walnut	Central California (Shasta Co. through Stanislaus Co.)
***J. major* (Torr.) Heller** *J. rupestris* var. *major* Torr. *J. microcarpa* var. *major* (Torr.) L. Benson *J. elaeopyren* Dode	**Arizona walnut,** Arizona black walnut, *nogal, nogal silvestre*	Central & SW Texas to SW New Mexico, Arizona, & mtns of northern Mexico
***J. microcarpa* Berl.** *J. rupestris* Englem. ex Torr.	**little walnut,** Texas walnut, river walnut, *nogal*, *nogalito, namboca,* Texas black walnut	W Oklahoma,W & S Texas & SE New Mexico, S to NE Mexico
***J. nigra* L.** *Wallia nigra* (L.) Alef.	**black walnut,** eastern black walnut, American walnut	W Vermont, S Ontario, & New York,W to S Minnesota & SE South Dakota; S to central Texas & NW Florida
***J. regia* L.**	**English walnut,** Persian walnut, Carpathian walnut	SE Europe to Himalayas & China

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

cultural varieties of English and Japanese walnuts have been developed (Brinkman 1974). Black walnut has demonstrated tremendous geographic variation in growth, wood, and fruiting characteristics (Bey 1970; Bresnan and others 1994; Rink and Kung 1995; Rink and Phelps 1989; Rink and others 1994; Williams and others 1985), and selected material has performed well (Beineke 1989; Hammitt 1989). Around 400 cultivars of this species alone have been released (Rink 1988; Williams 1990). Seed collection zones have also been recommended for black walnut (Deneke and others 1980).

Flowering and fruiting. Walnuts are monoecious. The greenish male flowers are slender catkins that develop from axillary buds on the previous year's outer nodes. They range in length from 5 to 7 cm on California walnut to 10 to 20 cm on Arizona walnut (Krochmal and Krochmal 1982; Sargent 1965). The small female flowers, usually 6 to 12 mm long, occur in short terminal spikes on the current year's shoots. The flowers appear with or shortly after the leaves in the spring (table 2). The ovoid, globose, or pear-shaped fruits ripen in the first year. The fruit is a nut enclosed in an indehiscent, thick husk that develops from a floral involucre (figure 1). The diameters range from 1 to 2 cm for little walnut to 5 to 8 cm for butternut (Krochmal and Krochmal 1982; Sargent 1965). The nut (figure 2) is incompletely 2- or 4-celled and has a bony, furrowed shell (figure 3). Available data on seeding habits of 8 species are listed in table 3.

Collection of fruits. Walnut fruits can be collected from the ground after natural dispersal in fall or early winter (table 2), or they may be dislodged from the trees by shak-

Figure 1—*Juglans*, walnut: nuts (enclosed in their husks) of *J. cinerea*, butternut (**left**) and *J. nigra*, black walnut (**right**).

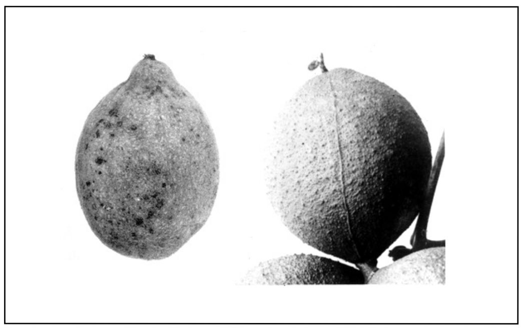


Figure 2—*Juglans*, walnut: nuts (with their husks removed) of *J. cinerea*, butternut (**top left**); *J. hindsii;* Hinds walnut (**top right**); *J. californica,* California walnut (**center left**); *J. nigra;* black walnut (**center right**); *J. microarpa*, little walnut (**bottom left**).

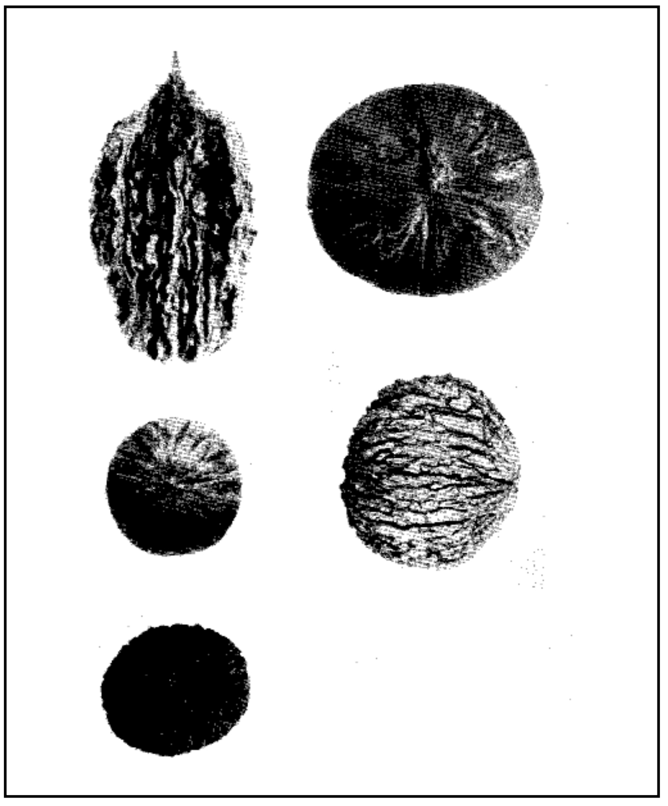
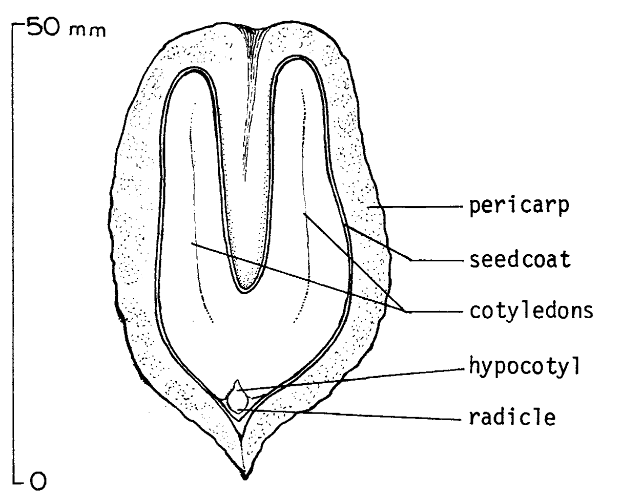


Table 2—*Juglans,* walnut: phenology of flowering and fruiting

Species	Flowering	Fruit ripening	Seed dispersal
*J. ailantifolia**	May–June	Aug–Oct	Oct
J. californica	Mar–Apr	Fall	Fall
J. cinerea	Apr–June	Sept–Oct	After leaf-fall
J. hindsii	Apr–May	Aug–Sept	Sept–Oct
J. major	Spring	Fall	Fall
J. microcarpa	Mar–Apr	Aug–Sept	Fall
J. nigra	Apr–June	Sept–Oct	After leaf-fall
J. regia	Mar–May	Sept–Nov	Fall

Sources: Brinkman (1974), Rink (1990), Vines (1960), Williams (1990), Wyman (1947).
* Dates are for Japan and Massachusetts.

Figure 3—*Juglans cinera*, butternut: longitudinal section through a seed.



ing branches or the whole tree with mechanical shakers. Collections should start promptly after the nuts are mature to prevent losses to rodents. Maturity is generally indicated by a darkening color of the fruit husk (table 3). Healthy butternut trees will yield up to .3 hl (.9 bu) each of clean nuts, and black walnut may produce 1 hl (2.9 bu) or more of fruit. Even though black walnut nut production is under strong genetic control (Jones 1993), environmental factors are very important. Nut production on pole-sized black walnuts was doubled in one trial by application of nitrogen and phosphorus at 4.5 and 2.3 kg (9.9 and 5.1 lb), respectively, per tree (Ponder 1976). Yield was 400 to 450 nuts/tree. Three hectoliters (8.4 bu) of black walnut and Hinds walnut fruits should yield about 1 hl (2.8 bu) of sound seeds (Brinkman 1974). Yield, size, and number of fruits per weight vary considerably among species (table 4).

Extraction and storage of seeds. Nuts are easy to extract when the husks are in an early stage of softening—that is, firm on the outside but slightly soft next to the nut. Black walnut nuts collected in the eastern United States are

Table 3—*Juglans,* walnut: height, seed-bearing age, seedcrops frequency, and fruit ripeness criteria

Species	Height at maturity (m)	Year first cultivated	Minimum seed-bearing age (yrs)	Years between large seedcrops	Fruit ripeness criteria: Preripe color	Ripe color
J. ailantifolia	20	1860	10	1–3	—	—
J. californica	12	1889	5-8	—	Light green	Dark brown
J. cinerea	30	1633	20	2–3	Greenish bronze	Greenish brown
J. hindsii	24	1878	9	—	Light yellow-green	Dark brown to black
J. major	15	1894	—	—	—	—
J. microcarpa	6	1868	20	—	—	—
J. nigra	46	1686	12	2–3	Light green	Yellowish green
J. regia	27	Long cultivated	8	—	Light yellowish green	Black

Source: Brinkman (1974).

Table 4—*Juglans,* walnut: cleaned seed and other yield data

Species	Place collected	Fruit wt/fruit vol kg/hl	Fruit wt/fruit vol lb/bu	Seed wt/fruit vol kg/h	Seed wt/fruit vol lb/bu	Cleaned seeds/weight Range /kg	Range /lb	Average /kg	Average /lb	Samples
J. ailantifolia	Japan	—	—	—	—	130–175	60–80	155	70	2
J. californica	California	—	—	—	—	65–165	30–75	110	50	2
J. cinerea	—	—	—	—	—	33–88	15–40	66	30	13
J. hindsii	Shasta Co., California	47	36	16	12.5	64–175	29–80	100	45	3
J. major	Coconino Co., Arizona	—	—	—	—	170–225	77–102	200	90	10
J. microcarpa	—	—	—	—	—	170–235	78–107	203	92	2
J. nigra	—	62	48	—	—	25–220	11–100	88	40	20+
J. regia	California	—	—	—	—	66–110	30–50	88	40	10+

often spread on the ground in the shade to allow husks to dry and deteriorate. If husks are allowed to dry too much, however, they become very hard and removal is difficult. In the slightly soft stage, husks can be removed by hand or by running the fruits through a macerator or a corn sheller. For commercial quantities of nuts, mechanical hullers are available. After complete husk removal, unfilled nuts can be separated from filled nuts by water floatation. Seeds enclosed in their husks will germinate, but most nurseries find it easier to control seedling density in the beds with cleaned seeds. Husking is necessary if seeds are to be treated with a fungicide.

Walnut nuts are basically orthodox in storage behavior (that is, capable of surviving desiccation), but their high lipid contents put them in the sub-orthodox category (Bonner 1990). Nuts of most species can be stored with or without their husks and are commonly stored without. If their moisture contents are reduced to around 10 to 15%, nuts can be stored at below-freezing temperatures. Long-term storage of walnuts is not common, however, and nuts are commonly stored at higher temperatures and moisture contents. Nuts of Japanese and little walnuts and butternut were successfully stored for several years at relative humidities of 80 to 90% and temperatures of 1 to 4 °C (Brinkman 1974). Cleaned black walnuts with a moisture content of 20 to 40% were stored successfully at 3 °C for a year in plastic bags (Williams 1971b), and nuts with 50% moisture in a screen container were buried in an outdoor pit for 4 years without significant loss in germination capacity (Williams 1971a).

Table 5—*Juglans*, walnut: stratification period, germination test conditions and results

Species	Cold stratification period* (days)	Germination test conditions† Daily light period (hr)	Temp (°C) Day	Temp (°C) Night	Days	Germination rate Amt (%)	Germination rate Days	Germination % Avg (%)	Germination % Samples	Purity (%)
J. ailantifolia‡	0	—	—	—	42	—	—	75	3	—
J. californica	156	—	—	—	30	—	—	58	3+	—
J. cinerea	90–120	8+	30	20	50–80	54	58	65	7	96
J. hindsii	156	—	30	20	30+	—	—	41	4	—
J. major	120–190	8+	30	20	49	10	28	64	5	—
J. microcarpa	190	—	30	20	30–60	68	14	46	7	94
J. nigra	90–120	8+	30	20	15–40	60	24	50	14+	87
J. regia	30–156	—	30	20	40	—	—	82	4	High

Source: Brinkman (1974).
* Stratification temperatures ranged from 1 to 5 °C.
† Test media were soil or sand.
‡ Seeds were soaked in water for 10 days before sowing.

Table 6—*Juglans*, walnut: nursery practice

Species	Stratification* Medium	Stratification* Days	Sowing season	Seedlings/area /m²	Seedlings/area /ft²	Sowing depth cm	Sowing depth in	Mulch Type	Mulch Depth cm	Mulch Depth in
J. californica	Peat	150	Spring	—	—	5	2	—	—	1
J. cinerea	Sand	90–120	Spring	—	—	2.5–5	1–2	Sawdust	2.5	1
	—	—	Fall	—	—	2.5–5	1–2	None	—	—
J. hindsii†	—	—	Fall	65–68	700–732	2.5	1	Vermiculite	2.5	1
J. major	Sand or peat	90–150	Spring	—	—	5	2	—	—	—
J. microcarpa	—	—	Fall	35–65	377–700	2.5–5	1–2	Sawdust	2.5	1
J. nigra	Sand	90–100	Spring	35–65	377–700	2.5–5	1–2	—	—	—
J. regia	Sand	30+	Spring	—	—	5	2	—	—	—

Sources: Brinkman (1974), Schultz and Thompson (1990), Williams and Hanks (1976).
* Outdoors during the winter or in a cold room at 1 to 5°C.
† Seeds were soaked in water at 88 °C for 1½ to 2 minutes before sowing.

Pregermination treatment. Seeds of most walnut species exhibit an embryo dormancy that can be broken by stratification at temperatures of 1 to 5 °C (table 5). For Japanese walnut, however, water soaking is adequate (Brinkman 1974). In practice, walnut seeds are either sown in the fall soon after collection or stratified over winter for spring-sowing. Large amounts are sometimes stratified in moist sand covered with at least 15 cm (6 in) of soil, sand, or mulch (Rink 1988). This process can be carried out in a hole in the ground or above ground with wooden sideboards to hold sand, nuts, soil, and mulch in place. Screening is nearly always necessary to exclude rodents, and a fungicide may be applied to prevent disease during stratification. Small lots of seeds may be stratified in plastic bags, moist peat, or sand at the same temperatures for 90 to 120 days. For Illinois sources, at least 100 days of cold stratification are required to overcome dormancy (Van Sambeek and others 1990).

Germination tests. There are no official seed testing prescriptions for walnuts. Germination of stratified nuts can be tested in flats of sand, peat, or soil (table 5). An alternating temperature regime of 20 °C for 16 hours and 30 °C for 8 hours is best; light is not necessary during testing. Nuts can also be tested in laboratory germinators on thick paper wadding, but their size often makes this impractical. Properly stratified seeds usually germinate within 4 weeks, but much variation among seedlots can be expected. Examples of test results are included in table 5. Indirect estimates of viability can also be made with radiographs, although exact predictions of viability are unlikely. If radiopaque agents are employed, cracked seedcoats and damaged tissues can be detected (Vozzo 1978). Moisture determinations can be made on walnuts by breaking open the nuts and drying the pieces for 17 hours at 103 °C (Bonner 1982). If the nuts are not broken, moisture may be trapped inside during drying, and the resulting percentage calculation will underestimate the moisture content.

Nursery practice. Research has demonstrated that a good black walnut seedling should have a top length of 38 to 50 cm (15 to 20 in), a stem diameter of 8 mm ($^1/_3$ in), and 8 to 10 permanent first-order lateral roots (Schultz and Thompson 1990). Unstratified nuts may be sown in the fall soon after collection, usually with the husks removed. It has been reported that husk removal will prevent predation by rodents (Nielson 1973), but subsequent tests have not supported this claim (Phares and others 1974). A hot-water soak of 1.5 to 2 minutes preceding fall-sowing of Hinds walnut has been helpful (Stuke 1960). To minimize alternate freezing and thawing overwinter, seedbeds should be mulched with sawdust, hay, or straw. The heavier mulches must be removed when germination begins in the spring. Stratified nuts must be used for spring-sowing; in the northeastern United States, spring-sown stratified black walnuts had more than double the germination of fall-sown unstratified seeds (DeHayes and Waite 1982). Although only 100 days of stratification may be required to overcome dormancy, additional time (up to 184 days) can increase the rate of emergence (Van Sambeek and others 1990). Some nurseries broadcast the nuts on tilled beds and press them into the soil with rollers, but a more common practice is to sow the nuts by hand in drill marks at a bed density of about 160 nuts/m^2 (15/ft^2). To produce the large seedlings that are necessary for successful outplanting of black walnut, bed densities of 35 to 65 seedlings/m^2 (3 to 6/ft^2) and root pruning in July (for the midwestern United States) to a depth of 15 cm (6 in) are recommended (Schultz and Thompson 1990). Nuts should be covered with 2.5 to 5 cm (1 to 2 in) of nursery soil (table 6); screening to exclude rodents is prudent, especially for fall-sown nuts.

Nuts of Hinds walnuts often are sown directly into growing beds, and the seedlings are then thinned to leave 20 cm (8 in) between plants in the row. A special technique is used in some nurseries: (a) unhulled nuts are air-dried to reduce moisture to about 50% and kept outdoors until January; (b) the partially dried nuts then are put into "sprout beds" containing as many as 3 layers of nuts with 2.5 cm (1 in) of sand below and 2.5 cm (1 in) of vermiculite above each layer; (c) about March 15, the beds are opened up and the sprouted nuts are hand-transferred to growing beds in rows spaced 1.5 m (5 ft) apart with the nuts 20 cm (8 in) apart in the row (Brinkman 1974). Black walnut can also be grown in containers (Van Sambeek 1988a).

Black walnut is susceptible to 2 serious root rot diseases in the nursery caused by *Phytophthora citricola* Sawada and *Cylindrocladium* spp. (Williams 1990). At one time, these diseases were controlled by chemical fumigation of seedbeds, but environmental concerns have eliminated these treatments. An alternative, but less effective, method is to treat the nuts with fungicides before sowing (Brinkman 1990). Because regulations for chemical applications change frequently, persons growing walnut seedlings should check with local state and federal extension agents for the latest information.

Vegetative propagation by cuttings is possible, but difficult (Farmer 1973). Most cultivars are budded or bench-grafted on seedling understock (Dirr and Heuser 1987; Van Sambeek 1988b). There has also been considerable research activity in embryo and tissue culture of walnuts (Long and others 1995; Van Sambeek and others 1990).

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Cupressaceae—Cypress family

Juniperus L.

juniper

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. There are about 50 species of junipers widely distributed throughout the temperate and subtropical regions of the Northern Hemisphere and south of the Equator in Africa. Most are evergreen shrubs and small trees. Thirteen species are native to the United States (Little 1979), and 11 of these are included in this book (table 1). Eastern redcedar is the most widespread juniper in the eastern United States, and Rocky Mountain juniper and Utah juniper are very common in the West. Common juniper is one of the most widespread tree species in the Northern Hemisphere, ranging from Asia to Europe and North America.

The close-grained, aromatic, and durable wood of the larger junipers was once used for furniture, interior paneling, novelties, posts, poles, fuel, and charcoal (Dealy 1990; Hemmerly 1970; Lawson 1990; Noble 1990; Wilhite 1990). The most important current uses are for firewood, furniture, paneling, and novelty products. Juniper "berries" are used for flavoring in cooking and in gin (the word "gin" is derived from the Dutch word for juniper, *jenever)*. Junipers are also valuable for watershed and windbreak plantings, wildlife habitat and food, and ornamental use (Dealy 1990; Johnsen and Alexander 1974; Lawson 1990; Noble 1990; Wilhite 1990). Their utility as ornamental plants has led to the selection and propagation of many horticultural varieties (Dirr and Heuser 1987; Vines 1960). Some junipers are sources for natural oil products. Cedar-wood oil is extracted from the heartwood and foliage of Ashe juniper and eastern redcedar to produce fragrance in soaps, sprays, disinfectants, and cleaning agents. Rocky Mountain juniper oils have the potential for these uses also (Adams 1987). Because of the encroachment of junipers onto range and pasture lands, particularly in the West, considerable effort has been directed toward their control (Burkhardt and Tisdale 1976; Jameson 1966; Johnsen 1962; McPherson and Wright 1990).

Genetic variation and hybridization. Junipers exhibit considerable natural variation in their growth habit and appearance, and studies have established marked differences in color, crown form, growth rate, and disease resistance in eastern redcedar (Henderson and others 1979; Minckler and Ryker 1959; Seidel and Watt 1969; Tauer and others 1987; Van Deusen 1979), Rocky Mountain juniper (Tauer and others 1987), and western juniper (Matthews 1945). Where ranges of the junipers overlap, natural hybridization abounds. This condition probably explains the large number of reported varieties of North American junipers (Dealey 1990; Fassett 1945; Hall 1952; Hall and others 1961; Lawson 1990; Noble 1990; Ross and Duncan 1949; Vines 1960; Wilhite 1990).

Flowering and fruiting. The small, inconspicuous flowers are borne in the spring (table 2) on the ends of short branchlets or along the branchlets. The flowers are dioecious or occasionally monoecious in oneseed juniper and some sources of western juniper (Dealy 1990; Johnsen and Alexander 1974). Pollen cones are yellow, terminal, and about 3 to 4 mm long; ovulate cones are composed of pointed scales, 3 to 8 in number, that fuse to form a fleshy cone 6 to 8 mm long (figure 1) (Brown and Kirkman 1990). The fleshy cones are commonly called berries. Cones are usually greenish in color when immature and change to a bluish black or reddish brown as they mature in the autumn (table 2). Most are covered with a conspicuous glaucous bloom. Cones of alligator, Utah, and common junipers require 2 years to reach full maturity, but those of common juniper may require 3 years in some parts of its range (Johnsen and Alexander 1974; Vines 1960). Cones of the other junipers mature in the fall of the first year (table 2). The outer skins of the cones may be thin and resinous, as in Virginia redcedar and Rocky Mountain and oneseed junipers, or dry and leathery or mealy, as in alligator and Utah junipers (Johnsen and Alexander 1974).

Table 1—*Juniperus,* juniper: nomenclature and occurrences

Scientific name & synonym(s)	Common name(s)	Occurrence
J. ashei **Buchh.** *J. sabinoides* (H.B.K.) Nees *J. mexicana* Spreng. *J. monticola* Martinez	**Ashe juniper,** mountain cedar, rock cedar, Mexican juniper	S Missouri, N Arkansas, NE & S Oklahoma, central & trans-Pecos Texas, & Mexico
J. californica **Carr.**	**California juniper,** desert white-cedar	SW Oregon, N California to Baja California & Mexico
J. communis **L.** *J. sibirica* Burgsd.	**common juniper,** dwarf juniper, prostrate juniper	Greenland, Newfoundland, & Labrador to NW Alaska, S in US from Washington, Montana, North Dakota, & Minnesota to California, Arizona, New Mexico, Georgia, & South Carolina; also in Europe & Asia
J. deppeana **Steud.** *J. mexicana* Schlecht. & Cham. *J. pachyphlaea* Torr. *J. deppeana* var. *pachyphlaea* (Torr.) Martinez	**alligator juniper,** checkered-bark juniper, western juniper (lumber)	Trans-Pecos Texas to W New Mexico & central Arizona; S to N & central Mexico
J. monosperma **(Engelm.) Sarg.** *J. occidentalis* var. *monosperma* Engelm. (Engelm.) Cory *J. mexicana* var. *monosperma*	**oneseed juniper,** cherrystone juniper, redberry juniper, west Texas juniper, *sabina*	Colorado, Utah, & Nevada S to SE Arizona, S New Mexico, central Texas, & Mexico
J. occidentalis **Hook**	**western juniper,** Sierra juniper	W Montana, Idaho, & Washington to Oregon, S California & W Nevada
J. osteosperma **(Torr.) Little** *J. californica* var. *utahensis* Engelm. *J. utahensis* (Engelm.) Lemmon	**Utah juniper,** bigberry juniper, western juniper (lumber), *sabina*	S Idaho & Nevada & SW Wyoming S to E & SE California, central Arizona, & W New Mexico
J. pinchotii **Sudworth** *J. monosperma* var. *pinchotii* (Sudworth) Van Melle *J. texensis* Van Melle	**Pinchot juniper,** redberry juniper	Central to NW & trans-Pecos Texas, SW Oklahoma & SE New Mexico
J. scopulorum **Sarg.** *J. scopulorum* var. *columnaris* Fassett	**Rocky Mountain juniper,** Rocky Mountain redcedar, redcedar, river juniper	NW to SE Alberta, E & S British Columbia, S to W North Dakota & Montana, Washington, E Oregon, Nevada, Colorado, South Dakota, Nebraska, to S Arizona, New Mexico, & trans-Pecos & NW Texas
J. virginiana **L.** *J. viriginiana* var. *crebra* Fern. & Grisc.	**eastern redcedar,** red juniper, *savin*	SW Maine, W to N New York, S Quebec, Ontario, Michigan, Wisconsin, Minnesota to SW North Dakota, to W Kansas, Oklahoma, to central Texas & E to Georgia
J. virginiana* var. *silicicola **(Small) J. Silboa** *J. silicicola* (Small) Bailey	**southern redcedar,** eastern redcedar	SE North & South Carolina & S & central Florida, W to S Mississippi & SE Texas

Sources: Johnsen and Alexander (1974), (1971, 1979).

There may be 1 to 4 brownish seeds per juniper cone (table 3). The seeds are rounded or angled, often with longitudinal pits (figure 2) and have thick, bony seedcoats (figure 3). Embedded within the fleshy, white- or cream-colored endosperm is a straight embryo with 2 to 6 cotyledons. Junipers begin bearing seeds when they are about 10 to 20 years old. Heavy seedcrops are irregular, but some seeds are produced almost every year. Large numbers of empty seeds are common in juniper crops, a likely result of poor pollination. Seeds disperse during the autumn, but some ripe cones of most species will persist on the trees through the winter. Seeds are naturally dispersed, usually by birds that eat the cones (Chavez-Ramirez and Slack 1994; Holthuijzen and others 1987; Livingston 1972).

Not much is known about the insects that infest seeds of junipers, or how much damage they do to seedcrops. Larvae of *Eurytoma juniperina* Marcovitch, a sawfly, have been found in seeds of Utah and western junipers and eastern redcedar. Larvae of *Periploca atrata* Hodges and another unnamed Cochylidae moth are known to feed on seeds of alligator and California junipers (Hedlin and others 1980).

Collection of cones. Juniper cones are usually collected in the fall by stripping them from the branches by hand directly into containers. Cones can also be collected by shaking or flailing the limbs to dislodge the cones onto netting or dropcloths on the ground. The larger fruits of alli-

Figure 1—*Juniperus,* juniper: strobili ("berries") of *J. ashei,* Ashe juniper (**top left**); *J. californica*, California juniper **(top center**); *J. deppeana*, alligator juniper (**top right**); *J. occidentalis*, western juniper (**middle left**); *J. pinchotii*, Pinchot juniper (**middle center**); *J. scopulorum*, Rocky Mountain juniper (**middle right**); and *J. virginiana* var. *silicicola*, southern juniper (**bottom left**); *J. virginiana*, eastern redcedar (**bottom right**).

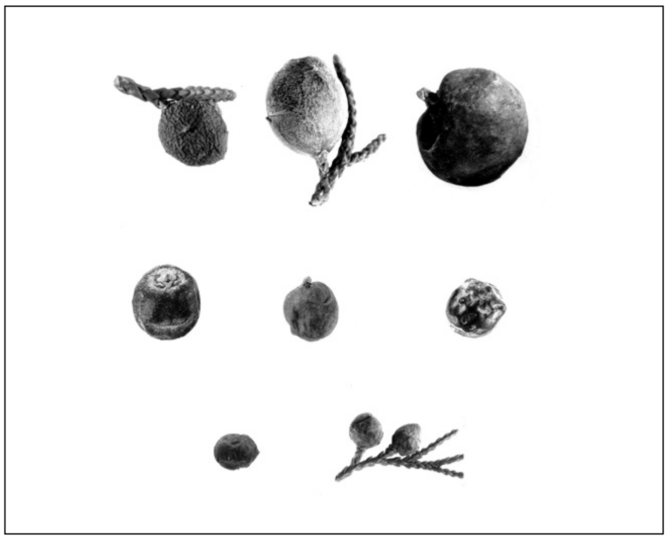
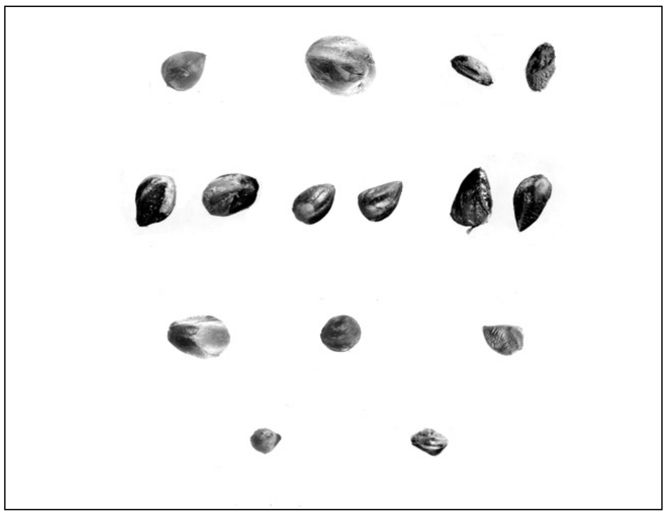


Figure 2—*Juniperus*, juniper: seeds of *J. ashei*, Ashe juniper (**top left**); *J. californica*, California juniper (**top center**); *J. communis*, common juniper (**top right**); *J. deppeana*, alligator juniper (**second row left**); *J. monosperma*, oneseed juniper (**second row middle**); *J. occidentalis*, western juniper (**second row right**); *J. osteosperma*, Utah juniper (**third row left**); *J. pinchotii*, Pinchot juniper (**third row middle**); *J. scopulorum*, Rocky Mountain juniper (**third row right**); and *J. virginiana* var. *silicicola*, southern juniper (**bottom left**); *J. virginiana*, eastern redcedar (**bottom right**).



gator and Utah junipers may be picked up from the ground after dispersal (Johnsen and Alexander 1974). Care should be taken to avoid collecting from plants with large numbers of green immature cones because they are difficult to separate from the mature ones. It is always wise to perform cutting tests on samples from each tree or group of trees to determine the percentage of filled seeds. The number of filled seeds can vary widely from tree to tree, as noted above, and collections can be adjusted to allow for this condition. Although collection can be delayed over much of the winter for some species, it is desirable to collect the fruits as soon as possible after ripening to reduce losses to wildlife. Freshly collected cones should be spread to avoid heating but should not be dried enough to make the fleshy covering tough and difficult to remove.

Table 2—*Juniperus,* juniper: phenology of flowering and fruiting

Species	Location	Flowering	Fruit ripening	Seed dispersal
J. ashei	—	Jan–Apr	Sept–Nov	Fall–winter
J. communis	—	Apr–May	Aug–Oct	Persists for 2 yrs (2nd–3rd yr)
J. deppeana	—	Feb–Mar	Aug–Oct	Persists for 2 seasons (2nd yr)
J. monosperma	Arizona	Mar–Apr	Aug–Sept	Oct–Nov (persists 1–2 yrs)
J. occidentalis	Oregon	Mid-Apr–mid-May	Mid-Sept	Persists for 2 yrs
J. osteosperma	Arizona	Mar–Apr	Sept (2nd year)	Persists for 2 yrs
J. pinchotii	Texas	Spring	Oct–Nov	Year-round
J. scopulorum	—	Mid–Apr–mid-June	Mid-Sept–mid-Dec	October (persists 2–3 yrs)
J. virginiana	Nebraska	Mid–Mar–mid-May	Sept–Nov	Feb–Mar (1st yr)
J. virginiana var. *silicicola*	South Carolina	Jan–Feb	Oct–Nov	—

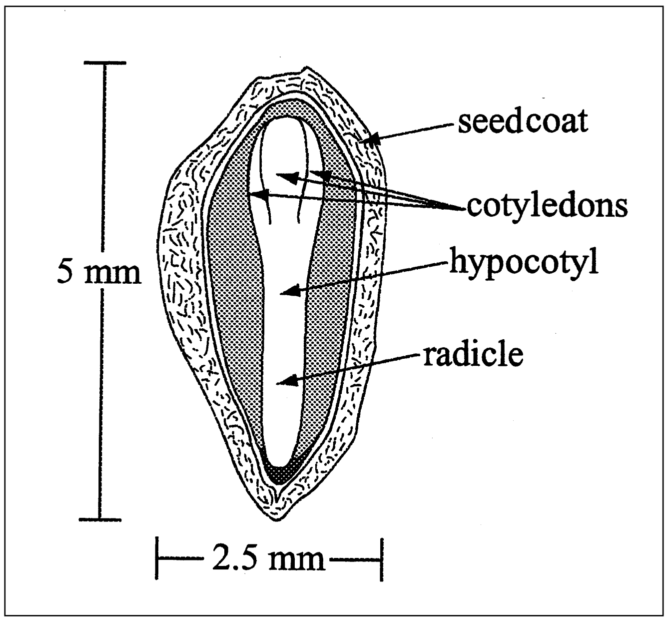
Sources: Johnsen and Alexander (1974), Rehder (1956), Vines (1960).

Table 3—*Juniperus,* juniper: height, seedcrop frequency, and fruit color

Species	Height at maturity (m)	Year first cultivated	Seeds/ cone	Years between large seedcrops	Fruit ripeness criteria Preripe color	Ripe color
J. ashei	3–6	1925	1–2	—	Green	Deep blue
J. californica	1–5	—	1–2	—	Bluish w/dense bloom	Reddish brown
J. communis	1–15	1560	1–3	Irregular	Red	Bluish to black, glaucous
J. deppeana	3–20	1873	2–4	—	Green	Bluish to reddish brown, glaucous
J. monosperma	3–8	1900	1–2	2–5	Green with waxy bloom	Copper to dark blue with white waxy bloom
J. occidentalis	5–9	1840	2–3	—	Green-blue	Bluish black, glaucous
J. osteosperma	5–12	1900	1–2	2	Green glaucous	Reddish brown,
J. pinchotii	1–5	—	1	—	Green with light bloom	Copper to red to reddish brown
J. scopulorum	6–15	1936	1–2	2–5	Green with	Blue w/white waxy bloom
J. virginiana	9–30	1664	1–2	2–3	Green	Blue
J. virginiana var. *silicicola*	7	—	1–2	—	Green	Dark blue

Sources: Johnson and Alexander (1974), Sargent (1965), Vines (1960).

Figure 3—*Juniperus scopulorum*, Rocky Mountain juniper: longitudinal section through a seed.



Extraction and storage of seeds. Twigs, leaves, and other debris should be removed by winnowing, screening, or aspiration. Seeds can be easily extracted from the pulpy cones by maceration with water. Small seedlots can be cleaned with laboratory or kitchen blenders, and large lots can be cleaned in larger macerators. Full seeds should sink, and pulp and empty seeds can be floated off the top of the water (Johnsen 1959; Johnsen and Alexander 1974). For extraction of Rocky Mountain juniper and eastern redcedar seeds, a cone volume to water volume ratio of 1:2.5 is recommended. The pulp residue can then be removed from the filled seeds by adding a little liquid detergent to warm water and agitating for about 5 minutes (Van Haverbeke and Barnhart 1978). Dried fruits should be soaked in water for several hours before macerating. After the seeds have been separated from the pulp and cleaned, they can be prepared for stratification or dried for storage. Intact cones can be stored also, but this is not usually done. Seed yields and weights are listed in table 4.

Juniper seeds are orthodox in storage behavior. They should be air-dried to a moisture content of about 10% and stored at temperatures of 5 to 18 °C (Johnsen and Alexander 1974; Jones 1962; Stoeckler and Slabaugh 1965). There have been no long-term studies to compare different storage temperatures and moisture contents for juniper, but results are available from several sources. Seeds of Ashe juniper stored in a bag at about 5 °C and high humidity retained about half their original viability after 4 years, and seeds of Rocky Mountain juniper stored in sealed containers at 12 to 16 °C (both in dried cones and as cleaned seeds) showed about 30% germination after 3 $^{1}/_{2}$ years (Johnsen and Alexander 1974). The seeds of alligator, oneseed, and Utah junipers stored dry in sealed bags or jars at room temperature for 45, 21, and 9 years, respectively, yielded germination of 17, 51, and 16% (Johnsen 1959).

Pregermination treatments and germination tests. Juniper seeds germinate very slowly due to conditions of deep dormancy. Their dormancy appears to result from internal embryo dormancy, seed coat dormancy, germination inhibitors in the pulp of the cones, or a combination of all

Table 4—*Juniperus,* juniper: seed yield data

Species	Place collected	Cleaned seeds/weight Range /kg	/lb	Average /kg	/lb	Samples
J. ashei	—	—	—	22,270	10,100	1
J. communis	—	56,120–120,170	25,450–54,500	80,480	36,500	8
J. deppeana	Arizona	19,840–34,400	9,000–15,600	28,270	12,820	5
J. monosperma	Arizona & New Mexico	33,650–44,100	15,260–20,000	40,350	18,300	10
J. occidentalis	Oregon	17,640–34,970	8,000–15,860	27,120	12,300	—
J. osteosperma	Arizona	7,940–15,660	3,600–7,100	10,910	4,950	15
J. pinchotii	Sonora & Texas	21,280–30,650	9,650–13,900	24,230	10,990	2
J. scopulorum	Arizona	39,360–92,830	17,850–42,100	59,760	27,100	36
J. virginiana	Great Plains	81,580–121,270	37,000–55,000	96,140	43,600	34

Sources: Johnsen and Alexander (1974), Stoeckler and Slabaugh (1965), Vines (1960).

three (Johnsen and Alexander 1974). There is wide variation among species in degree of dormancy. The least dormant may be eastern and southern redcedars, whereas Rocky Mountain juniper is among the most dormant (Rietveld 1989). There is also considerable variation among sources and crop years; some seedlots from alligator and oneseed junipers germinated without any stratification (Johnsen 1959; Meagher 1943; Riffle and Springfield 1968).

The most common treatment for overcoming dormancy is long periods of moist stratification at 3 to 5 °C. Periods of 30 to 180 days have been used for seeds of Ashe, alligator, and oneseed junipers and eastern redcedar (Barton 1951; Benson 1976; Johnsen and Alexander 1974; Taylor 1941). Early reports suggested freezing juniper seeds during stratification, but this method has generally been unsuccessful (Johnsen and Alexander 1974). Seeds of common, Utah, and Rocky Mountain junipers, eastern redcedar, and possibly western juniper often respond positively to warm stratification at room temperature (around 25 °C) or alternating temperatures of 20 °C (night) and 30 °C (day) for 45 to 240 days, followed by cold stratification for similar periods (Johnsen and Alexander 1974; Rietveld 1989; Van Haverbeke and Comer 1985). Young and others (1988), however, reported no response by western and Utah junipers to the 2-temperature pretreatment. The best treatment for eastern redcedar was to first soak the seeds for 96 hours in a 10,000 ppm solution of citric acid, followed by warm stratification for 6 weeks and cold stratification for 10 weeks (Van Haverbeke and Comer 1985). The use of citric acid was suggested by Cotrufo (1963), and although the nature of the stimulation is unknown, some seedlots respond with faster germination rates. Faster germination has also been reported for seeds of Pinchot and Rocky Mountain junipers and eastern redcedar that were soaked in concentrated sulfuric acid for periods of 35 to 120 minutes (Djavanshir and Fechner 1976; Johnsen and Alexander 1974), although stimulation for the latter 2 species occurred only when the carbonized layer was removed from the surface of the seeds (Djavanshir and Fechner 1976). Washing seeds of oneseed juniper in running water for 48 hours, followed by 30 minutes in 30% hydrogen peroxide, stimulated germination to 79% from 47% for untreated controls (Riffle and Springfield 1968). Another promising method reported for western and Utah junipers is 12 weeks of soaking in aerated water at 5 °C; germination percentages of around 50% were recorded for both species. If gibberellin (GA_3 at 0.289 mmol/liter) was added to the aerated solution, germination increased to 84% for western juniper and to 64% for Utah juniper (Young and others 1988).

Prescriptions for official germination tests have been established for 3 species: common juniper, eastern redcedar, and Rocky mountain juniper (ISTA 1993). Tetrazolium staining is the recommended method for these species, but alternative stratification directions are also suggested. Common juniper should be stratified for 90 days at 3 to 5 °C, whereas eastern redcedar and Rocky Mountain juniper seeds should receive 60 days at 20 °C, followed by 45 and 40 days, respectively, at 3 to 5 °C. Recommended germination temperatures are 20 °C for common juniper and 15 °C for eastern redcedar and Rocky Mountain juniper. Germination of Pinchot juniper is reported to be best at 18 °C (Smith and others 1975), but no data exist for the other junipers. There is obviously much to learn about stimulation of germination for the junipers, and more research is called for. Germination capacities for various pretreatments and test conditions are given in table 5. Germination is epigeal (figure 4).

Nursury practices. Juniper seeds are usually sown in the late summer or fall, but may be sown in the spring or early summer. All seeds should usually be stratified, no matter when they are sown, but untreated seeds can be used in some circumstances. Untreated fresh seeds may be sown in the fall within a week after collection and extraction if they are not dried (Meines 1965). Stratified seeds sown in the spring should be in the ground early enough to ensure complete germination before the air temperatures go higher than 21 °C. If stratification is successful, germination should begin 6 to 10 days after sowing and be completed in 4 to 5 weeks (Johnsen and Alexander 1974; Stoeckler and Slabaugh 1965).

Juniper seeds are usually drilled in rows 15 to 20 cm (6 to 8 in) apart and covered with about 6 mm (1/4 in) of firmed soil or sand (Stoeckler and Slabaugh 1965). The seeds are occasionally broadcast by hand onto the seedbed and covered with sand. The beds should be mulched with straw, sawdust, burlap, or plastic film to prevent winter drying, alternate freezing and thawing, and premature germination in the spring. The mulch normally must be held in place to prevent blowing (Stoeckler and Slabaugh 1965). The seedbeds should be kept moist, and burlap or plastic film mulches should be removed as soon as germination begins. Light shade should then be provided by slat-wire snow fence or plastic screening materials; young seedlings of eastern redcedar and Ashe, oneseed, and Rocky Mountain junipers should be shaded throughout the first growing season. Seedlings of alligator juniper (figure 4) should be shaded only during the germination period (Johnsen and Alexander 1974). Burlap may be used over snow-fence shade structures to conserve moisture and to protect against early spring freezing (Stoeckler and Slabaugh 1965). During the autumn,

Figure 4—*Juniperus deppeana*, alligator juniper: seedling development at 2, 17, 43, and 96 days after germination

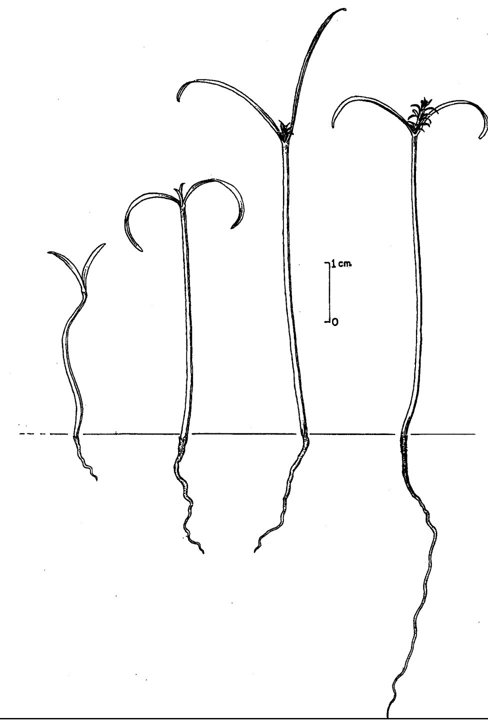


Table 5—*Juniperus*, juniper: germination test conditions and results

Species	Stratification period (days) Warm*	Cold†	Daily light (hr)	Medium	Temp (°C) Day	Night	Germination rate Days	%	Germination percentage Days	%	Samples
J. ashei	0	120	—	Sand	86	68	60	30	10	33	1
	0	120	—	Sand	50	50	60	27	29	36	1
J. communis	60–90	90+	8	Paper, sand	86	68	20–30	—	—	7–75	10+
J. deppeana	0	0	—	Paper, sand	86	68	40	—	—	16-30	2
	0	30–60	—	Sand, peat	86	68	30–40	—	—	45	1
J. monosperma	0	0	—	Sand, peat, soil	86	68	30–70	—	—	20–75	34
J. osteosperma	120	120	—	Sand, soil	86	68	70	—	—	8–49	8
J. pinchotii‡	0	0	8	Perlite	60	60	36+	—	—	63	4
	30	60	8	Perlite	60	60	—	—	—	53	4
J. scopulorum	120	120	—	Paper, sand	86	68	20–30	5–31	8–15	22	7
J. virginiana	0	30–120	—	Paper, sand	50	77	20–30	6–74	9–24	76	16
	4§	90	0	Perlite	58	58	60	84	30	87	3
	0	45	—	Kimpak	60	60	66	70	43	78	2

Sources: Cotrufo (1963), Johnsen (1959), Johnsen and Alexander (1974), Meagher (1943), Riffle and Springfield (1968).
* 30 to 20 °C alternated diurnally.
† 5 °C.
‡ Seeds soaked in sulfuric acid 45 minutes.
§ Seeds soaked in 1% citric acid for 4 days.

the seedlings may change color due to freezing weather, reduced watering, or increased light intensity resulting from removal of the half-shades. Seedlings of eastern redcedar change from green to purple, most markedly with the 1+0 seedlings. The normal green color returns the next spring.

In the West, juniper seedlings are usually transplanted in the nursery after the first or second year. Early lifting in the spring gives the best survival. Roots must be kept moist during lifting, and the seedlings can be stored as long as a week before transplanting with little damage if kept cool and moist (Afanasiev and others 1959). Spacing in the transplant bed ranges from 15 by 2.5 cm (6 by 1 in) to 20 by 5 cm (8 by 2 in) for eastern redcedar and Rocky Mountain juniper (Johnsen and Alexander 1974; Stoeckler and Slabaugh 1965). Undercutting of third-year transplants of Rocky Mountain juniper seems to stimulate strong lateral root development (Stoeckler and Slabaugh 1965).

The most serious nursery disease that affects junipers is the cedar blight, which is caused by *Phomopsis juniperovora* Hahn (Otta and others 1980; Peterson 1973; Stoeckler and Slabaugh 1965). Good sanitation practices in the nursery and chemical control measures are needed to keep this disease in check. Once established in a nursery site, it is very difficult to eradicate (Stoeckler and Slabaugh 1965). Other diseases that cause problems for junipers are cercospora blight, caused by *Cercospora sequoiae* Ellis & Everh. var. *juniperi* Ellis & Everh. (Peterson 1977; Peterson and Wysong 1968) and cedar apple rust, caused by *Gymnosporangium juniperi-virginianae* Schwein. (Stoeckler and Slabaugh 1965). Application regulations and chemical recommendations change frequently, so local extension experts should be consulted for the current chemical control measures for these diseases in the nursery.

Other nursery pests that affect junipers are nematodes, grubs, and red spiders—*Pentamerismus erythrens* Ewing. Foliage may be damaged by winter injury and drying out, even in second-year beds and transplant beds. The plants usually recover during the spring. Small juniper seedlings are also subject to frost heaving, which can be reduced by heavy mulching or overhead sprinklers (Stoeckler and Slabaugh 1965).

Many junipers can be propagated vegetatively with cuttings (Dirr and Heuser 1987). There is evidence of wide variation in rooting ability among populations of common juniper (Houle and Babeux 1994). Rooting success as high as 82% has been reported for Rocky Mountain juniper (Edson and others 1996). Treatment of the 12-cm-long (5-in-long) cuttings with 1.6 or 3.0% indole-butyric acid (IBA) accelerated rooting by several months and increased overall succes by up to 36%. Two years after transplanting to containers, 92% of the stecklings survived.

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