United States Department of Agriculture

Field Guide for Managing Dyer's Woad in the Southwest

Forest Service

Southwestern Region

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Dyer's wood (*Isatis tinctoria* L.)

Mustard family (Brassicaceae)

Dyer's woad is listed as a noxious weed in both Arizona and New Mexico. This field guide serves as the U.S. Forest Service's recommendations for management of dyer's woad in forests, woodlands, and rangelands associated with its Southwestern Region. The Southwestern Region covers Arizona and New Mexico, which together have 11 national forests. The Region also includes four national grasslands located in northeastern New Mexico, western Oklahoma, and the Texas panhandle.

Description

Dyer's woad (synonyms: Asp-of-Jerusalem, glastum) is a member of the Mustard family and grows as a winter annual, biennial, or short-lived perennial. It is native to southeastern Russia and has historically been cultivated for use as a blue dye. It is currently being studied for its anti-cancer properties and potential as a less toxic alternative to wood preservative and inkjet printer fluid. Dyer's woad was accidentally introduced to the western United States as a contaminant in alfalfa seed during the early 1900s and has since proliferated throughout the arid West. It has been observed in Santa Fe and Sandoval Counties in New Mexico and Coconino County in Arizona.

Growth Characteristics

- Grows as a winter annual, biennial or short-lived perennial herbaceous plant depending on local environmental conditions; 2 to 4 feet tall.
- Produces a taproot (30 inches long) and lateral roots found mostly in the upper 12 inches of soil.
- Has fine haired, basal leaves in the rosette stage that are twice as long as they are wide with a pale mid-vein; grows an erect upright woody stem (20 to 35 inches) during bolting.
- Stems leaves are 1 to 4 inches long, grayish-green, narrow, alternate, basally lobed; clasping stem.
- Yellow, 4-petalled flowers occur mostly in April to July in flat-topped bunches at branch tips.
- Fruits are a primary distinguishing feature. Thin, flat pods are initially green, turning black at maturity; the persistent, samara-like fruits hang from slender, short pedicels.
- Reproduces via seed; each pod produces one seed. However, each plant produces an average of 300 to 500 seeds; under certain conditions a single plant may produce 10,000 seeds.

Ecology

Impacts/threats

Dyer's woad develops dense, monotypic stands that crowd out native species. Established infestations reduce forage available for cattle and horses, degrade wildlife habitat, lower flora and fauna species diversity, and decrease land value. Its dominant presence increases the potential for soil erosion.

Location

This weed is common along roadsides and railway rights-of-way; upon dry, rocky foothills and hillsides; within both disturbed and undisturbed pastures and rangelands. It is a serious problem especially in intermountain sagebrush communities in Utah, Nevada, Wyoming, Montana, and California.

Spread

Seed is easily dispersed by animals, human activity, and water. Seed is spread long distances as a contaminant in alfalfa hay or seed, and by adhering to surfaces and undercarriages of vehicles and road maintenance equipment.

Invasive Features

Dyer's woad is an aggressive, dry-land invader due to its prolific seed production, early emergence, and deep taproot. Initial invasion may occur in a disturbed area; however, it can rapidly expand into undisturbed rangeland and wooded areas. Dyer's woad produces a water-soluble chemical that inhibits germination of other plants and can delay its own germination until favorable precipitation levels are available.

HOARY CRESS
(*Cardaria draba*)

Description: Hoary cress, also referred to as heart-podded hoary cress, perennial pepper-grass and whitetop, is a member of the Brassicaceae or mustard family. Hoary cress is a deep-rooted perennial forb that can grow up to 2 feet tall. Stems of the plant are erect or procumbent, branching above, glabrous or slightly to densely pubescent below, and appear gray in color. Hoary cress has both basal and stem leaves. Basal leaves have scattered to dense pubescence, irregularly toothed to entire and taper to a short stalk that attaches to the crown of the plant near the ground. Middle and upper stem leaves are sparsely pubescent to glabrous, obovate, elliptic-oblong, or lanceolate, irregularly toothed to entire, and grayish-green in color. Upper leaves have two lobes that clasp the stem. Flowers of the plant are white, four-petaled, and borne on slender stalks. Fruits of the plant are a mature silicle or pod that is shaped like an inverted heart and usually contains two seeds. Seeds are oval or round at one end, narrow to a blunt point at the other, and reddish-brown in color.

Hoary cress

Plant Images:

Rosettes

Flowers

Infestation

Distribution and Habitat: Hoary cress is considered naturalized throughout Europe and other continents. The plant can occur in a variety of soil conditions with moderate moisture and typically the plant is abundant on alkaline soils that are wet during late spring. Hoary cress can be found in grainfields, hayfields, croplands, pastures, waste sites, feed lots, and along roadside and irrigation ditches.

Life History/Ecology: Hoary cress is a herbaceous, deep-rooted perennial that reproduces vegetatively and by seed production. Seedlings of the plant begin to germinate and establish a root system that consists of vertical and lateral roots in the spring and fall. Both the vertical and lateral roots can produce adventitious buds that develop into rhizomes and new shoots. Seedlings that are produced in the fall overwinter as rosettes. Plants begin to emerge the following spring, flower from May to June, and begin producing seeds by July. A single plant can produce between 1,200 to 4,800 seeds each year, with a single flowering stem capable of producing as many as 850 seeds. Seeds can remain viable in the soil for approximately three years.

Hoary cress contains glucosinolates that may have allelopathic potential.

History of Introduction: Hoary cress is native to the Balkan Peninsula, Armenia, Turkey, Israel, Syria, Iraq and Iran. The plant is widely introduced and naturalized throughout Europe and all other continents. Hoary cress was first introduced to the United States in Long Island, New York, in 1862, through ship ballast or contaminated alfalfa. In North Dakota, hoary cress has had scattered occurrences and has been found in 27 counties across the state including: Foster, Stark, Slope, Billings, Adams, McKenzie, Dickey, Dunn, Grant, Williams, Mountrail, Golden Valley, Hettinger, Adams, Morton, Bottineau, Sheridan, Wells, Cavalier, Grand Forks, Trial, Cass, Barnes, Ransom, and Richland.

Effects of Invasion: Hoary cress is an aggressive plant that can form dense monocultures on disturbed habitats. Disturbances such as grazing, cultivation, and especially irrigation can promote the colonization and spread of the plant. Hoary cress can displace native plant species, thereby reducing bio-diversity and forage production. Wildlife habitat is also negatively affected by the plant.

Control:
Management objectives for hoary cress control should involve containing and controlling known infestations and preventing infestations from spreading to new areas. Initial establishment of hoary cress is frequently by seed, therefore control methods should be conducted during the seedling or rosette growth stage of the plant prior to seed production. Seeds of hoary cress can remain viable in the soil for approximately three years, therefore infestations should be monitored to prevent re-establishment. However, hoary cress can also regenerate from an extensive root system. As a result, control methods should be combined into an integrated management system for the best long-term control of the plant.

Mechanical - Digging can provide control for small infestations of hoary cress if the entire root system is removed. Digging should be conducted to completely remove the plant within 10 days of emergence throughout the growing season for two to four years to be successful. Hand pulling generally is not effective because the root system may not be entirely removed. Cultivation is the major factor for the spread of the plant because root fragments that are left behind can produce new plants. Cultivation can eradicate plants if cultivations are repeated frequently throughout the growing season for a period of two to four years. Mowing has had variable results. In some studies, hoary cress was able to survive repeated removal of top-growth for at least one growing season without a loss in plant vigor. After two consecutive years of mowing, a noticeable decline in plant vigor was observed. Other studies suggest, mowing can reduce biomass, seed production, and shoots produced. Plants that were mowed during flowering produced fewer viable seeds than plants that were mowed during bolting. However, mowing does not provide long-term control and should be combined with other control methods to be more effective. Burning may enhance the growth of hoary cress as it re-sprouts from rhizomes or seed production. Little information is available on prescribed burning for hoary cress control. Further research is needed in this area.

Chemical - Herbicides can be used to control hoary cress, but success can be difficult. Metsulfuron, chlorsulfuron, MCPA, DCPA, dicamba, glyphosate, and 2,4-D have been used to control the plant. However, timing of herbicide application is important and herbicide re-treatment may be needed to provide the hoary cress control desired in a long-term management plan. Most studies recommend that herbicides should be applied at the bud or flowering stage when herbicides are translocated with carbon into the roots and rhizomes of the plants.

Contact your local county extension agent for recommended use rates, locations, and timing.

Biological - No biological control agents are available for hoary cress. Sheep will graze hoary cress in the early growth stages, but some reports state that cattle may produce tainted milk as a result of consuming the plant.

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Hoary cress photograph courtesy of Colorado State University Cooperative Extension - Natural Resources.

Rosettes photograph courtesy of T. Breitenfeldt, Montana War on Weeds, (mtwow.org).

Flowers and infestation photographs courtesy of JC Schou, Biopix.dk.

Managing Canada Thistle

Canada thistle (*Cirsium arvense*) is a perennial that has plagued farmers in America since European settlement, and is a Noxious Weed in Pennsylvania. It is adapted to a wide range of soil conditions, and spreads vigorously by wind-borne seeds and by way of its extensive, creeping root system.

Not Your Average Thistle

The key to Canada thistle's weediness is its root system. The roots of Canada thistle spread aggressively, and can increase the width of a thistle patch 6 to 10 feet in a season. As the root system spreads, it gives rise to new shoots. If left unchecked, a single Canada thistle plant eventually turns into a patch containing thousands of stems.

Although thistle may serve as a food source for some insects and provide seed to some bird species, it has a negative impact on wildlife habitat quality in your CREP planting. Canada thistle grows in dense patches and reduces the vigor and establishment of grassland plantings and riparian buffers that are planted to improve wildlife habitat.

The plants you are most likely to confuse Canada thistle with are other thistles. The common, weedy thistles in PA include bull thistle (*Cirsium vulgare*), musk thistle (*Carduus nutans*), and plumeless thistle (*Carduus acanthoides*). All these thistles grow erect, have spiny foliage, and bear prominent pink flowers that produce seed attached to downy

Figure 1. A flowering stem of Canada thistle showing flowers ranging from the pea-like bud stage to nearly ready to disperse ripened seed. The stems of Canada thistle are smooth, while the other common weedy thistles in Pennsylvania have spiny 'wings' on their stems.

Figure 2. A 'patch' of Canada thistle emerging in the spring. A patch is often one plant, with hundreds or thousands of stems arising from a shared root system.

'umbrellas' that carry them on the wind, much like dandelion seed.

Bull, musk, and plumeless thistles are biennials. They have a single, strongly-taprooted crown, and reproduce only by seed. You can distinguish Canada thistle from the biennial thistles because it has small flowers (less than 1 inch) and smooth stems between the leaves (Figure 1). The biennial thistles all have spiny 'wings' - tissue that looks like a continuation of the leaf - along their stems. Another distinguishing feature is that well-established Canada thistle grows in distinct patches (Figure 2) that are easily seen early in the spring as the thistle is emerging.

The typical growth pattern for Canada thistle begins with emergence of the new shoots in the first few weeks of spring. This first flush of growth enters the flower bud stage in late May to mid-June when the plants are 3 to 4 feet tall. The scaly flower heads are the size of a large pea. The heads open showing pink flowers up to 1 inch in diameter, then close after fertilization to shelter the ripening seed. When the seed is ripe, the flower opens again and releases the 'summer snow' that carries the seed away.

Canada Thistle Control Measures

To eliminate Canada thistle you must injure and exhaust its root system, and do it repeatedly. A successful control program requires multiple seasons, and multiple treatments within a season (Table 1).

A well-established groundcover, particularly a grassland

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planting, greatly aids your control efforts by competing with the thistle as you suppress it.

The most important opportunity for control is the fall when thistle is recharging its root system for the next growing season. Fall is the ideal time to maximize injury to the thistle's root system because systemic herbicides move through plants with the sugars being sent to the roots. As the thistle is stocking up its root reserves for the winter, it will send fall-applied herbicides to where they can do the most damage. Product selection is more important in the fall as only a few herbicides available for use in CREP plantings are truly effective Canada thistle control products (Table 1).

Late spring, when thistle is at the bud-to-early-bloom stage is the second important opportunity for control. Much of the energy to produce the spring flush of growth comes from stored reserves in the root system, causing a seasonal-low of stored energy at bloom stage. This is an ideal time to eliminate the top growth and force the plant to use its scarce reserves to regrow.

An herbicide application at bloom stage will serve as a 'chemical mowing'. The choice of herbicide treatment in the spring is not as critical as it will be in the fall. The spring application acts somewhat like a burndown treatment, eliminating the top-growth, but injury to the root system is limited. Well-established Canada thistle will eventually regrow after a spring application, regardless of the treatment. What is important is that the treatment effectively eliminates the existing top growth.

In grassland plantings, there are many inexpensive herbicide products that will selectively eliminate the aboveground thistle growth and leave grasses intact. In tree plantings, spot treatments using *glyphosate* reduce the risk of injuring the trees with broadleaf herbicides through root absorption.

An alternative to a late-spring herbicide treatment is a mowing timed for bud to early-bloom stage. This mowing should be as low to the ground as practical. After the grassland cover or riparian buffers are established, only spot mowing can be allowed by the FSA County Committee - and only approved on an annual basis.

After seed set, Canada thistle produces a second flush of growth. Some of it comes from buds on the spring stems, and a lot of it comes as new shoots from the root system. Instead of growing tall and flowering, the second flush of growth produces just enough foliage to 'recharge' the root system. This is the target of the critical fall herbicide application.

There is no 'silver bullet' for Canada thistle control. Once you accept that you need multiple treatments for multiple seasons, you will find it is a species you can successfully manage.

Table 1. Managing Canada thistle requires treatment in the spring to prevent seed set and eliminate the first flush of growth, ***and*** *in the fall to maximize injury to the root system. Choose one spring treatment and one fall treatment. The spring treatment is applied at bud to early-bloom stage. Herbicide choice is less critical in the spring because no treatment will prevent regrowth. The spring treatments listed below are just a few examples - any herbicide treatment that will kill the top growth is useful. The fall herbicide treatment maximizes injury to the root system, so only products known for their activity against Canada thistle are recommended.*

timing	treatment	product rate (oz/ac)	comments
late spring	Roundup Pro	64	Roundup Pro is just one of many *glyphosate* products. A spot treatment with *glyphosate* is the recommended herbicide alternative in tree plantings because there is no soil activity that could lead to herbicide injury through root absorption.
late spring	broadleaf herbicide	varies	In grassland plantings, there are many relatively inexpensive products that will provide burn-down of Canada thistle. Examples include 'Weedmaster' and 'KambaMaster' (*dicamba + 2,4-D*),
late spring	mowing	- -	If mowing once, mow at bud to early bloom stage to maximize root system depletion. Spot mowing may be necessary in grassland plantings.
fall	Milestone	6	Milestone (*aminopyralid*) is very active against thistles and legumes. This treatment will not injure established grasses, but should not be used in close proximity to desirable trees.
fall	Forefront R&P	32	Forefront is a mixture of *aminopyralid* plus *2,4-D*, and provides a broader spectrum of control if other broadleaf weeds are present. This treatment will not injure established grasses, but should not be used in close proximity to desirable trees.
fall	Telar	2	At lower rates, Telar XP (*chlorsulfuron*) is safe to grasses, but this rate will cause significant injury to most grasses.
fall	Roundup Pro	128	Roundup Pro (*glyphosate*) is non-selective, and this rate will severely injure all contacted vegetation. This is the best option - as a spot treatment - for use in hardwood plantings and riparian forest buffers because *glyphosate* has no soil activity.
fall	Vanquish	48	Vanquish is a less-volatile formulation of *dicamba*, the active ingredient in the 'Banvel' products. This treatment will not injure established grasses, but should not be used in close proximity to desirable trees.

Cooperative Extension

Bringing the University to You

Fact Sheet-02-57

Managing Scotch Thistle

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Scotch thistle (*Onopordum acanthium*) is a native of Europe and Asia. It was introduced into the United States in the late 19th Century as an ornamental; it has since escaped cultivation. Scotch thistle is also known by two other common names: cotton thistle and woolly thistle. This is because the leaves and upper stems are covered with thick cottony hairs.

Scotch thistle is an invasive weed that infests disturbed and neglected lands. It prefers sites near ditch banks and rivers, but also infests pastureland, crops, rangeland and roadsides. Its leaves are armed with sharp spines, making access to areas infested with scotch thistle difficult.

Historically, scotch thistle has been used to treat cancers, ulcers, and to slow the discharge of mucous membranes. Its thick hairs were used to stuff pillows and the oil from its seeds was used for cooking and burning. Although it had significant historical uses, scotch thistle is no longer used for these purposes and has become a troublesome weed for farmers, ranchers and land managers.

Description and Habitat

Scotch thistle is a biennial that flowers in the summer. The first year, scotch thistle grows very spiny leaves in a large rosette (a plant with leaves radiating from the crown (center) close to the ground and without flower stalks) that can be 12 inches or more wide (Fig. 1). Flowering shoots are produced the second season. The plants grow eight to 12 feet tall, up to five feet wide, and are multi-branched. The shoots and leaves are covered with thick hairs giving the plant a distinct bluish-green look. The leaves grow down the winged stems. The oblong leaves on the plant can be up to two feet long and a foot wide. Their lobes end in a very sharp yellow, green or white spine. The stems of scotch thistle become coarse and the leaves become more rectangular with age. The vibrant purple flower grows at the end of leafy stalks, as a single flower or as a cluster of flowers (Fig. 2). The flowers are an inch in diameter or larger. In dry years, when the plant is short, it can still flower and create as many seeds as a full-sized plant. Plants grow 70 to over 300 flower heads, and each flower produces 100 to 200 seeds.

Scotch thistle reproduces only by seed. It has an egg-shaped (obovate) seed that is dark brown

Figure 1. A scotch thistle plant in flower with a second season flower head (upper insert) and a first season rosette (lower insert).

or black with a bristle-like pappus (hairs) at one end. Seeds germinate and plants thrive in wide-open, disturbed, moist areas such as ditches, gullies, and roadsides. A water-soluble germination inhibitor in the seed coat must be leached away to allow the seed to germinate, thus the need for moisture.

Although scotch thistle prefers disturbed areas with high soil moisture, drier areas do not limit its invasive nature. It commonly invades overgrazed lands, rangeland, pastures, roadsides and construction sites.

Figure 2. Scotch thistle flower head.

Associated Impacts

Scotch thistle is present in all of Nevada's 17 counties. It drastically reduces productive rangeland by out competing desirable forage species. It can be so thick that it becomes an impenetrable, thorny barrier for ranchers, cattle, wildlife and recreationists.

Although, scotch thistle is considered a biennial weed, it can behave as an annual or a short-lived perennial. All of these variations contribute to the persistence of the plant. It is not bound by strict photoperiods (daylight) or temperature requirements for growth and flowering. Its flexibility in flowering is responsible for its success in so many different climates and growing conditions.

Wind, water, wildlife, livestock and human activities disperse the seeds. Most of the seeds fall close to the plant. The seeds remain viable in the soil for up to seven years. The seed must be completely embedded in the soil to germinate. Scotch thistle seeds will germinate and seedlings will grow in nutrient-deficient soils. Moisture and temperature determine the plant's success, not an abundance of soil nutrients. This gives scotch thistle an advantage over desirable plant seedlings that attempt to compete in nutrient-deficient soils and allows it to thrive in overgrazed pastures and rangeland.

In addition to lost rangeland, scotch thistle is responsible for lost wildlife habitats and recreational areas. Wildlife forage is reduced by the presence of scotch thistle. Campsites and trails can become inaccessible and no longer enjoyable when infested with scotch thistle. Access to trails, stream banks and fishing areas can be completely cut off by scotch thistle.

Control and Management

Because scotch thistle reproduces by seed, it is one of the few invasive weeds that can be controlled by mechanical, chemical and cultural methods. A persistent combination of these methods will yield the best results. Keep in mind that scotch thistle has the ability to germinate nearly year round. This adds to the difficulties associated with control and the timing of herbicide applications. A combination of control methods is recommended.

- **Prevention**

The best and most cost effective method for weed control is prevention. This stage is often overlooked until costlier methods of control are required. By monitoring your land and destroying single plants or new infestations, great expense can be avoided. Cooperative effort among land managers is recommended to successfully prevent weed infestations among adjacent landowners. If a small infestation is found and eradicated immediately, before seeds are produced, it will reduce the chance of further infestation on your land and your neighbor's.

- **Mechanical/ Physical Controls**

Mechanical and physical control is very effective if completed before scotch thistle goes to seed. Mechanical control is effective because scotch thistle does not reproduce vegetatively. Severing the roots of the rosette or the plant kills it. Small infestations can be pulled by hand. This should be done with caution while wearing heavy gloves, a long-sleeved shirt and pant, and eye protection because scotch thistle has stout spines.

Most mechanical methods, such as tilling, are not appropriate for rangeland and waterways. It is very important to keep scotch thistle out of these areas.

Mowing makes the stand more uniform, which makes herbicide applications more effective, but mowing does not kill scotch thistle. Mowing before seed dispersal will limit the amount of seed available for germination. However, plants are able to produce seed even after they have been mowed. Consequently, mowing is not recommended unless used with a follow-up herbicide application or tillage.

- **Biological/Cultural Controls**

Currently, there are no insect biological control agents for scotch thistle in the United States.

Sheep and cattle will not graze scotch thistle. Goats will, but only in its early rosette stage. After it has developed a coarse stem and stout spines, goats refuse to eat it.

An infestation of scotch thistle may be reduced or eliminated with the planting of competitive grasses. Revegetating an area with competitive grasses following treatment helps prevent the invasion and establishment of new scotch thistle plants. Desirable forage that emerges during the growing season should be managed to increase its competitiveness. Not only does this help reduce the possibility of reinfestation by scotch thistle, the increased forage provides increased protection from soil erosion.

As part of a good grazing plan, the establishment of desirable forages is integral to a weed management program. By monitoring for scotch thistle, not overgrazing pastures, and establishing desirable forage, scotch thistle's threat can be reduced.

- **Chemical Control**

Various chemicals control scotch thistle. The growing stages, environmental conditions, stand size, density, location, and the product's cost are all factors to consider in selecting the correct herbicide for the job. A combination of chemical treatments may be necessary to achieve the desired level of control. Always check with your state or county weed specialist before purchasing and applying herbicides. *The label on each product must be read, understood and followed correctly. It's the law!*

Applying herbicides to scotch thistle rosettes is very effective. In this stage, applying products that contain clopyralid, dicamba, MCPA, picloram or 2, 4-D will successfully kill scotch thistle. It is effective to spray the rosettes in the spring or fall, but it is more effective in the fall. All live plants that escaped the spring application will be seedlings or rosettes and ready to be sprayed later in summer or fall. Do not let them go to seed. Table 1 (back page) is a list of chemicals and the suggested application rates to use on scotch thistle. Remember to carefully follow state or county restrictions in addition to the label directions. Failure to do so makes the applicator liable for any damages created by the chemical.

Summary

Scotch thistle is responsive to mechanical, cultural and chemical control methods. A combination of treatments is recommended, followed by a sound revegetation program. This will provide satisfactory management of scotch thistle. Retreatments of the area may be necessary for four to six years or until the seeds in the soil are exhausted. Revegetation along with an active control program will ensure healthy pastures, rangeland, cropland and recreation areas for years.

Additional Resources:

1) Ball, D., P.J.S. Hutchinson, T.L. Miller, D.W. Morishita, R. Parker, R.D. William, and J.P. Yenish. 2001. *Pacific Northwest Weed Management Handbook*. Oregon State University, Corvallis, OR. pp. 408.
2) Sheley, R.L. and J.K Petroff. 1999. *Biology and Management of Noxious Rangeland Weeds.* Oregon State University Press. Corvallis, OR. pp. 202-216.
3) Bussan, A.J., S.A. Dewey, T.D. Whitson and M.A. Trainor. 2001. *Weed Management Handbook.* Montana State University, Bozeman, MT. pp. 294.

Editing by Sue Strom

Table1. Recommended herbicides and application rates to control scotch thistle.[1]

Common Name **Herbicide Name**	**Rate (*ae or ai/A*)[2] or product/A**	**Timing**	**Remarks and Cautions**
2, 4-D Many Products	*1.5 - 2 lb*	Apply to seedlings, rosettes in fall & before flower stalk elongates in spring.	Do not allow spray to drift onto sensitive crops.
chlorsulfuron[3] Telar (75DF)	*0.75 oz* 1 oz. product	Apply to young actively growing plants.	Agitate mixture, use 0.25% nonionic surfactant; do not treat frozen ground, dry soils & sandy soils without rain, & avoid sensitive crops.
clopyralid[3] Transline (3EC)	*0.09 - 0.375 lb* 0.25 – 1 pt product	Apply up to thistle bud stage.	There are labeled crop, grazing & hay restrictions; soil residuals may damage crops up to 4 years.
clopyralid[3] *+ 2,4-D amine* Curtail (2.38EC)	1 - 5 qt product	Apply to actively growing thistle up to bud stage.	There are labeled crop, grazing & hay restrictions; soil residuals may damage crops up to 4 years.
dicamba Banvel, Clarity	*0.5 - 1 lb*	Apply to seedlings, rosettes in fall & before flower stalk elongates in spring.	Soil residual may affect crops for 12 to 18 months, grasses tolerate these rates.
metsulfuron[3] Escort (60DF)	*0.6 oz* 1 oz product	Apply post emergent to actively growing plants.	Non-cropland use only, use 0.025% v/v nonionic or silicone surfactant, use mixture within 24 hours, do not apply to fescue or creeping meadow foxtail.
picloram Tordon 22K (2EC)	*0.25 lb*	Apply in fall before bolting; follow up applications will be necessary to control seedlings & escaped plants.	**Restricted Use Herbicide**; Avoid water & sensitive plants; soil residuals may last over one year.
triclopyr + clopyralid[3] Redeem R&P	1.5 - 2 pt product	Apply to actively growing thistle from rosette to early bolting.	Apply no more than 4 pts per year; avoid drift; observe labeled over-seeding & reseeding restrictions.

1. Application rates adapted from the Pacific Northwest Weed Management Handbook and the Montana, Utah, Wyoming 2001 – 2002 Weed Management Handbook.
2. Acid equivalent or active ingredient per acre = ae or ai/A.
3. Caution: These products are persistent in alkaline (high pH) soils and may affect crops sown in subsequent years where they have been applied.

Information herein is offered with no discrimination. Listing a product does not imply endorsement by the authors, University of Nevada Cooperative Extension (UNCE) or its personnel. Likewise criticism of products or equipment not listed is neither implied nor intended. UNCE and its authorized agents do not assume liability for suggested use(s) of chemical or other pest control measures recommended herein. Pesticides must be applied according to the label directions to be lawfully and effectively applied.

Centaurea solstitialis L.

Yellow starthistle

Family: Asteraceae
Range: Most contiguous states, except a few southern and northeastern states.
Habitat: Open disturbed sites, open hillsides, grassland, rangeland, open woodlands, fields, pastures, roadsides, waste places. May also inhabit cultivated fields. Does not tolerate low light areas or shading.
Origin: Southern Europe. Accidentally introduced as a seed contaminant in alfalfa. It has spread rapidly since its introduction into California in the mid-1800s.
Impacts: Plants are highly competitive and typically develop dense, impenetrable stands that displace desirable vegetation in natural areas, rangelands, roadsides and other places. Yellow starthistle is considered one of the most serious rangeland weeds in the western U.S. Yellow starthistle is sometimes problematic in grain fields, where the seeds can contaminate the grain harvest and lower its quality and value. Yellow starthistle contains an unidentified compound that causes nigropallidal encephalomalacia or chewing disease in horses.
Western states listed as Noxious Weed: Arizona, California, Colorado, Idaho, Montana, New Mexico, Nevada, North Dakota, Oregon, South Dakota, Utah, Washington
California Invasive Plant Council (Cal-IPC) Inventory: High Invasiveness

Yellow starthistle is a simple to bushy winter annual, occasionally biennial, with spiny yellow-flowered heads and stiff wiry stems to 6 ft tall. Plants form a basal rosette of leaves until mid-spring. Stem leaves are alternate and mature foliage is grayish- to bluish-green, densely covered with fine white cottony hairs. Its leaf bases form wings along the stems. Rosette leaves typically wither by flowering time. The taproot can extend deep into the soil (> 6 ft) allowing plants to utilize deep soil moisture not available to other annual species, particularly grasses.

The flowerheads are solitary on stem tips, and consist of numerous yellow disk flowers. The phyllaries are densely to sparsely covered with cottony hairs or with patches of hairs at the bases of the spines. The central spine of the main phyllaries is 10 to 25 mm long, stiff, yellowish to straw-colored throughout. Yellow starthistle reproduces only by seed and develops two types of achenes. The outer ring of achenes is a dull dark brown, often speckled with tan, lacking pappus bristles, and often remaining in heads. The inner achenes are glossy, gray or tan to mottled cream-colored and tan, with slender white pappus bristles 2 to 5 mm long. Most seeds fall near the parent plant. Some seed is viable 8 days after flower initiation. Large flushes of seeds typically germinate after the first fall rains, but smaller germination flushes can occur during winter and early spring. Seeds can survive for up to about 10 years in the field under certain environmental conditions, but it appears that few seeds survive beyond 4 years.

NON-CHEMICAL CONTROL

Mechanical (pulling, cutting, disking)	Hand removal, mowing, or cultivation, when used to prevent seed production over 2 to 3 years or more (the soil life of the seeds), can reduce or eliminate an infestation. Manual removal of yellow starthistle is most effective with small patches or in maintenance programs where plants are sporadically located in the grassland system. This usually occurs with a new infestation or in the third year or later in a long-term management program. These methods can also be important in steep or uneven terrain where other mechanical tools (e.g., mowing) are impossible to use. To ensure that

	plants do not recover it is important to detach all above-ground stem material. Leaving even a 2-inch piece of the stem can result in recovery if leaves and buds are still attached to the base of the plant. The best timing for manual removal is after plants have bolted but before they produce viable seed (i.e. early flowering). At this time, plants are easy to recognize, and some or most of the lower leaves have senesced. If hand removal is conducted after plants begin to produce seeds, it may be necessary to put pulled plants in bags and remove them from the site. Hand removal is particularly easy in areas with competing vegetation. Under this condition, yellow starthistle will develop a more erect slender stem with few basal leaves. These plants are relatively brittle and easy to remove. In addition, they usually lack leaves at the base and, consequently, rarely recover even when a portion of the stem is left intact. Hand removal options for yellow starthistle typically include hand pulling, hoeing, or string trimming. Systematic surveys and repeated removal should be conducted every 2 to 4 weeks throughout the growing season. Mowing is most effective when 2 to 5% of the total population of seedheads is in bloom. Mowing too early can result in higher seed production. Plants should be cut below the height of the lowest branches. It will require multiple years of continuous mowing to successfully manage yellow starthistle. Mowing is best used in an integrated approach. Since it is a late season management tool, it is best employed in the later years of a long-term management program or in a lightly infested area. Mowing is not feasible in many locations due to rocks and steep terrain. Mowing is not always successful and can decrease the reproductive efforts of insect biocontrol agents, injure late growing native forb species, and reduce fall and winter forage for wildlife and livestock. The success of mowing depends on proper timing and the growth form of the plant. Mowing too early (before seedheads reach spiny stage) or too late (after seed set) will usually increase the yellow starthistle problem. Mowing too early in the season can remove competitive grass cover and promote vigorous yellow starthistle regrowth. If done too late, mowing scatters yellow starthistle seed. Best results were obtained by mowing once at the early flowering stage, and again 4 to 6 weeks later to cut regrowth during the floral bud stage. A dense spring canopy of desirable vegetation optimizes yellow starthistle control. Yellow starthistle plants with an erect, high-branching growth form are effectively controlled by a single mowing at the early flowering stage, while sprawling low-branching plants cannot be controlled even with repeated mowing. Despite its limitations, mowing conducted at the early flowering stage, before viable seed production, can be very effective for yellow starthistle control. Anecdotal information also indicates that mowing the standing skeletons in fall, before the first rains, can form a mulch that blocks light and suppresses subsequent germination of yellow starthistle. A flail mower is considered best. The yellow starthistle litter layer may be less suppressive to grass germination, as it is not as light dependent as yellow starthistle. Tillage is effective, and is occasionally used on roadsides. It is also often used in agricultural lands, which is probably why yellow starthistle is not a significant cropland weed. In wildlands and rangelands, tillage is usually not appropriate because it can damage important desirable species, increase erosion, alter soil structure, and expose the soil for rapid reinfestation if subsequent rainfall occurs. Any tillage operation that severs the roots below the soil surface can effectively control yellow starthistle. Early summer tillage, before viable seeds are set, and repeated tillage following rainfall/germination events will rapidly deplete the yellow starthistle seed bank, but may also have the same effect on the seed bank of desirable species.
Cultural	High-intensity short-duration grazing by sheep, goats, or cattle should be implemented during the period when yellow starthistle plants have bolted to just before they produce spiny heads. Cattle and sheep avoid yellow starthistle once the buds produce spines, whereas goats continue to browse plants even in the flowering stage. For this reason, goats have become a more popular method for controlling yellow starthistle in relatively small infestations. Grazing the weed during the bolting stage can provide palatable high protein forage (8 to 14%). This can be particularly useful in late spring and early summer when other annual species have senesced. Grazing alone will not provide long-term management or eradication of yellow starthistle, but can be a valuable tool in an integrated management program. This prescription must be continued for at least 3 years in a severe infestation to reduce the yellow starthistle seed bank. Prescribed burns can provide control if conducted at the proper timing. Burning should be timed to coincide with the very early yellow starthistle flowering stage. At this time yellow starthistle has yet to produce viable seed, whereas seeds of most desirable species have dispersed and grasses have dried to provide adequate fuel. Fire has little if any impact on seeds in the soil. Burning at other times may enhance yellow starthistle survival by removing the thatch and encouraging seed germination in fall. The ability to use repeated burning depends on climatic and environmental conditions. In areas where

	resources are ample and total plant biomass is abundant, 2 or 3 consecutive years of burning may be practical. However, in other situations, fuel loads may not be sufficient to allow multiple year burns. Consequently, prescribed burning may be more appropriate as part of an integrated approach. Air quality issues can be significant when burns are conducted adjacent to urban areas. A major risk of prescribed burning is the potential of fire escapes. This risk is greatest when burns are conducted during the summer months. In some areas, burning can lead to rapid invasion by other undesirable species with wind-dispersed seeds, particularly members of the sunflower family. In addition to summer burning, yellow starthistle seedlings have been controlled using winter or early spring flaming. This technique is somewhat nonselective, and control of yellow starthistle is inconsistent. When spring drought follows a flaming treatment, control of yellow starthistle can be excellent. In contrast, a wet spring can lead to complete failure and increased yellow starthistle infestation, particularly since competing species may be dramatically suppressed.
Biological	Six insects have become established for the control of yellow starthistle in the western United States. These include three species of weevils (seed-head weevil [*Bangasternus orientalis*], flower weevil [*Larinus curtus*], and the hairy weevil [*Eustenopus villosus*]), and three species of flies (seed-head fly [*Urophora sirunaseva*], peacock fly [*Chaetorellia australis*], and the false peacock fly [*Chaetorellia succinea*]). All six insects attack the flower heads of yellow starthistle and produce larvae that develop and feed within the seedhead. Of these, only four have become well established. Of these, only two, *Eustenopus villosus* and *Chaetorellia succinea*, have any significant impact on reproduction. The combination of these two insects reduces seed production by 43 to 76%. Although this level of suppression is not sufficient to provide long-term yellow starthistle management, the use of biological control agents can be an important component of an integrated management approach. A more successful biological control program will likely require the introduction of plant pathogens or other insects which attack roots, stems, or foliage. A new potential biological control agent is a root-feeding weevil, *Ceratapion basicorne*, that has shown promise under greenhouse conditions. It has yet to be approved, but is expected to be released in the next couple of years. The most widely studied pathogen for yellow starthistle control is the Mediterranean rust fungus *Puccinia jaceae*. It can attack the leaves and stem of yellow starthistle, causing enough stress to reduce flowerhead and seed production. Although it has been released it does not seem to have much impact on yellow starthistle populations.

CHEMICAL CONTROL

The following specific use information is based on published papers and reports by researchers and land managers. Other trade names may be available, and other compounds also are labeled for this weed. Directions for use may vary between brands; see label before use. Herbicides are listed by mode of action and then alphabetically. The order of herbicide listing is not reflective of the order of efficacy or preference.

GROWTH REGULATORS	
2,4-D Several names	**Rate:** 1 to 1.5 pt product/acre (0.48 to 0.72 lb a.e./acre) for small rosettes, 2 to 4 pt product/acre (0.95 to 1.9 lb a.e./acre) for larger plants up to bolting **Timing:** Postemergence from rosette to beginning of bolting, but before flowering. **Remarks:** 2,4-D controls larger plants well, but is not considered as effective as other growth regulator herbicides for season-long control. It is broadleaf-selective and may injure other non-target species, particularly crop plants. 2,4-D has no soil activity. Do not apply ester formulation when outside temperatures exceed 80°F. Amine forms are as effective as ester forms for small rosettes, and amine forms reduce the chance of off-target movement from volatility.
Aminocyclopyrachlor + chlorsulfuron *Perspective*	**Rate:** 3 to 5 oz product (*Perspective*)/acre **Timing:** Postemergence and preemergence. Postemergence applications are most effective when applied to plants from the seedling to the mid-rosette stage. **Remarks:** Aminocyclopyrachlor gives control of yellow starthistle similar to aminopyralid. *Perspective* provides broad-spectrum control of many broadleaf species. Although generally safe to grasses, it may suppress or injure certain annual and perennial grass species. Do not treat in the root zone of desirable trees and shrubs. Do not apply more than 11 oz product/acre per year. At this high rate, cool-season grasses will be damaged, including bluebunch wheatgrass. Not yet labeled for grazing lands. Add an adjuvant to the spray solution. This product is not approved for

	use in California and some counties of Colorado (San Luis Valley).
Aminopyralid *Milestone*	**Rate:** 3 to 5 oz product/acre (0.75 to 1.25 oz a.e./acre). Use higher rates when weeds are larger. **Timing:** Postemergence and preemergence. Postemergence applications are most effective when applied to plants from the seedling to the mid-rosette stage. Earlier applications (i.e., in fall) may not provide full-season control, and later applications (bolting to early spiny stage) will require higher rates. **Remarks:** Aminopyralid is one of the most effective herbicides for the control of yellow starthistle. It is safe on grasses, although preemergence application at high rates can greatly suppress invasive annual grasses, such as medusahead. Aminopyralid has a longer residual and higher activity than clopyralid. Other members of the Asteraceae and Fabaceae are very sensitive to aminopyralid. For postemergence applications, a non-ionic surfactant (0.25 to 0.5% v/v spray solution) enhances control under adverse environmental conditions; however, this is not normally necessary. Other premix formulations of aminopyralid can also be used for yellow starthistle control. These include *Opensight* (aminopyralid + metsulfuron; 1.5 to 2 oz product/acre) and *Forefront HL* (aminopyralid + 2,4-D; 2 to 2.6 pt product/acre), both applied at the rosette to bolting stages.
Clopyralid *Transline*	**Rate:** 0.25 to 0.67 pt product/acre (1.5 to 4 oz a.e./acre). Seedlings and rosettes can be treated at the lower rate, but bolted plants should be treated at higher rates. **Timing:** Postemergence and preemergence. For postemergence application, apply to plants from seedling to mid-bolting stage. However, since clopyralid has a shorter soil residual compared to aminopyralid, optimal timing is at the later rosette stages, but before bolting. Earlier applications (i.e., in fall) may not provide full-season control, and later applications (bolting to early spiny stage) will require higher rates and may not give sufficient control. **Remarks:** Clopyralid gives excellent control of yellow starthistle. While it is very safe on grasses, it will injure many members of the Asteraceae, particularly thistles, and can also injure legumes, including clovers. Most other broadleaf species and all grasses are not injured. When clopyralid is used to control seedlings a surfactant is not necessary. However, when treating older plants or plants exposed to moderate levels of drought stress, surfactants can enhance the activity of the herbicide.
Clopyralid + 2,4-D *Curtail*	**Rate:** 2 to 4 qt *Curtail*/acre **Timing:** Same as for clopyralid. **Remarks:** Add a non-ionic surfactant.
Dicamba *Banvel*, *Clarity*	**Rate:** 0.5 pt product/acre (0.25 lb a.e./acre) for seedlings, 1 to 1.5 pt product/acre (0.5 to 0.75 lb a.e./acre) for larger plants up to bolting. **Timing:** Postemergence to plants from rosette to beginning of bolting. **Remarks:** Dicamba is a broadleaf-selective herbicide often combined with other active ingredients. It is not typically used alone to control yellow starthistle. Dicamba is available mixed with diflufenzopyr in a formulation called *Overdrive*. This has been reported to be effective on yellow starthistle. Diflufenzopyr is an auxin transport inhibitor which causes dicamba to accumulate in shoot and root meristems, increasing its activity. *Overdrive* is applied postemergence at 4 to 8 oz product/acre to rapidly growing plants. Higher rates should be used on large annuals. Add a non-ionic surfactant to the treatment solution at 0.25% v/v or a methylated seed oil at 1% v/v solution.
Picloram *Tordon 22K*	**Rate:** 1 to 1.5 pt product/acre (4 to 6 oz a.e./acre) **Timing:** Postemergence and preemergence. Postemergence applications should be made to plants from rosette to bud formation stage. Apply when there is adequate soil moisture and weeds are growing rapidly. **Remarks:** Picloram acts much like aminopyralid, aminocyclopyrachlor, and clopyralid, but gives a broader spectrum of control and has much longer soil residual activity. It can provide about 2 to 3 years of control. Most broadleaf plants are susceptible. Although well-developed grasses are not usually injured by labeled use rates, some applicators have noted that young grass seedlings with fewer than four leaves may be killed. Do not apply near trees. *Tordon 22K* is a federally restricted use pesticide. Picloram is not registered for use in California.

Triclopyr *Garlon 3A, Garlon 4 Ultra*	**Rate:** 1 pt *Garlon 4 Ultra* or 1.33 pt *Garlon 3A*/acre (0.5 lb a.e./acre) for seedlings, up to 3 pt *Garlon 4 Ultra* or 4 pt *Garlon 3A*/acre (1.5 lb a.e./acre) for larger plants. **Timing:** Postemergence from seedling to bolting stage. **Remarks:** Triclopyr has little to no residual activity. It is broadleaf-selective and typically does not harm grasses. *Garlon 4 Ultra* is formulated as a low volatile ester. However, in warm temperatures, spraying onto hard surfaces such as rocks or pavement can increase the risk of volatilization and off-target damage.
AROMATIC AMINO ACID INHIBITORS	
Glyphosate *Roundup*, *Accord XRT II*, and others	**Rate:** Broadcast foliar treatment: 1.33 to 2.67 qt product (*Roundup ProMax*)/acre (1.5 to 3 lb a.e./acre). Spot treatment: 1% to 2% v/v solution **Timing:** Postemergence to plants from bolting to beginning of flowering. **Remarks:** Glyphosate is the most effective herbicide for late season control. Good coverage, clean water, and rapidly growing yellow starthistle plants are all essential for adequate control. It has no soil activity and is nonselective. To achieve selectivity, it can be applied using a wiper or spot treatment to control current year's plants.
BRANCHED-CHAIN AMINO ACID INHIBITORS	
Chlorsulfuron *Telar*	**Rate:** 1.33 to 2.6 oz product/acre (1 to 1.95 oz a.i./acre) **Timing:** Preemergence activity only. Chlorsulfuron does not have postemergence activity on yellow starthistle and must be used in combination with 2,4-D, dicamba, or triclopyr to provide effective control. **Remarks:** Chlorsulfuron has mixed selectivity on both broadleaf and grass species but is generally safe on grasses. It has fairly long soil residual activity. Herbicide solution requires constant agitation during application.
Imazapyr *Arsenal*, *Habitat*, *Stalker*, *Chopper*, *Polaris*	Not often used for yellow starthistle control but has been shown to be somewhat effective at 3 to 4 pt product/acre. It has preemergence and some postemergence activity, and a long soil residual.
Sulfometuron *Oust* and others	Not often used for yellow starthistle control but has been shown to be somewhat effective at 1 to 2 oz product/acre. It has preemergence activity only, and a long soil residual.
PHOTOSYNTHETIC INHIBITORS	
Hexazinone *Velpar L*	Not often used for yellow starthistle control but has been shown to be somewhat effective at 1 to 2.5 gal product/acre. It has preemergence activity only, and a long soil residual. High rates of hexazinone can create bare ground, so only use high rates in spot treatments.

RECOMMENDED CITATION: DiTomaso, J.M., G.B. Kyser et al. 2013. *Weed Control in Natural Areas in the Western United States*. Weed Research and Information Center, University of California. 544 pp.

United States Department of Agriculture

Natural Resources Conservation Service

Plant Guide

Prickly Russian Thistle

Salsola tragus L.

Plant Symbol = SATR12

Common Names: Tumbleweed, Russian thistle, Tumbling thistle

Scientific Names: Salsola iberica (Sennen & Pau) Botsch. ex Czerep., *Salsola kali* L. ssp. *ruthenica* Soó, *Salsola kali* L. ssp. *tenuifolia* Moq., *Salsola kali* L. ssp. *tragus* (L.) Čelak., *Salsola pestifer* A. Nelson, *Salsola ruthenica* Iljin *nom. illeg.* (illegitimate name), *Kali tragus* (L.) Scop., *Kali soda* Moench *nom. illeg.*

Figure 1: Prickly Russian thistle measuring approximately three feet (0.9m) tall and five feet (1.5m) wide. May produce over 200,000 seeds. Photo C. Bernau, Great Basin Plant Materials Center (GBPMC).

Salsola australis R. Br. was considered a Prickly Russian thistle synonym until tests in the 2000's identified it as genetically distinct. (Ryan and Ayres 2000, Borger et al. 2008, Hrusa and Gaskin 2008, Ayres et al. 2009, Chinnock 2010).

Scientific names for Prickly Russian thistle are mired in confusion, with over 55 synonyms (Rilke 1999) and an uncertain number of misapplied names throughout the scientific literature (Mosyakin 1996). In 2007, Akhani et al. recommended splitting the polyphyletic genus *Salsola* into ten or more genera, confirming the opinions earlier expressed by Tzvelev (1993) and other authors. Akhani suggested *Kali tragus* for *S. tragus*, which saw some adoption until the Nomenclatural Committee voted to conserve *Salsola* at the 2017 XIX International Botanical Congress (Akhani et al. 2007, 2014; Mosyakin et al. 2014, 2017; Wilson 2017). The vote reverted *K. tragus* back to *S. tragus*.

Description
General: Amaranth Family (Amaranthaceae; APG IV 2016). Alternatively, the Goosefoot family (Chenopodiaceae). The Goosefoot family is accepted as a distinct family by nearly all experts in this group (Hernández-Ledesma et al. 2015, Mosyakin and Iamonico 2017).

Prickly Russian thistle is an introduced, C4 photosynthetic, warm season, annual forb that reproduces by seed. It is native to arid and semi-arid ecosystems in southeastern Europe to Central Asia (probably also partly in northern Africa). It is an erect

Figure 2: Prickly Russian thistle is highly variable. These photos are flowering structures from three different plants found in the same location, all S. tragus. Stem venation color, bract and bracteoles length and coloration, and perianth size are some of the variation that exists. Photo C. Bernau, GBPMC.

(sometimes ascending or prostrate), rounded plant that can grow up to three feet (0.9 m) tall and six feet (1.8 m) in diameter (Fig 1). Its morphological characteristics are highly variable and may be expressed at any given location (Fig 2). Stems are opposite and many branched, often with red to purple longitudinal striations. Leaves are alternate, semi-succulent to succulent, 0.6-2 in (1.5-5 cm) long and 0.01-0.04 in (0.3-1 mm) thick, and end in a sharp spine. The plants are soft when immature, but as the plant matures it becomes stiff with sharp prickly spines. The root system consists of a taproot that can grow over 6 ft. (1.8 m) deep with extensive lateral roots over 5 ft. (1.5 m) long (Pan et al. 2001), though this may be stunted with competition (Boerboom 1995). Inflorescence is an open or somewhat condensed spike of a solitary flower or cluster of 2-3 flowers; clusters normally producing only a single developed fruit. Flowers are small, bisexual, with 3-5 stamens, 0.04-0.05 in (1.1-1.3 mm) long anthers, and a short style with 2 stigma branches. Flowers are subtended by a single 0.15-0.24 in (4-6 mm) long bract and two 0.09-0.20 in (2.5-5 mm) long slightly recurved bracteoles; all three rigid and sharply tipped (Fig 2, Fig 3). The undifferentiated perianth is five lobed, about 0.09-0.12 in (2.5-3 mm) long, and winged at midlength; typically with three well developed colorless translucent broad wings and two narrow wings. The upper half of the perianth becomes incurved over the fruit, sometimes forming a short and weak columnar beak. The fruit is a tightly coiled immature embryo (2n=36) covered by a thin membrane. It lacks stored energy reserves or any complex covering, though it is enclosed in the persistent perianth (Welsh et al. 2003, Holmgren et al. 2012). Seed production is prolific but highly variable and dependent on plant size, with smaller plants producing seeds in the thousands and the largest plants capable of producing over 250,000 seeds (Dewey 1894, Young 1991).

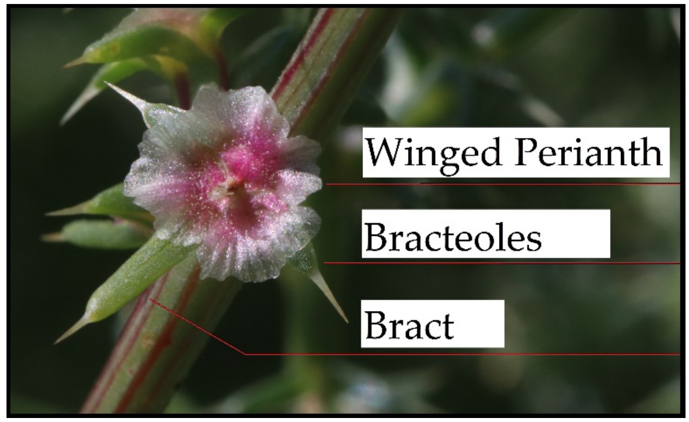

Figure 3: Prickly Russian thistle flower with Bract, Bracteoles, and Perianth labeled. Notice the upper half of the perianth in the center is incurved over the fruit, forming a short columnar beak. Photo C. Bernau, GBPMC.

Prickly Russian thistle typically matures in late summer to fall where, upon senescence, specialized abscission cells allow for a clean break at its base. The plant is then free to tumble in the wind to disperse seeds. The incurved tips of the persistent perianth prevent seeds from all dispersing immediately while the winged portion of the perianth assists in farther dispersal once the seed breaks away from the plant. Fresh seed germination is restricted by temperature, requiring a minimum day/night temperature of 68/41 °F (20/5 °C). Germination restrictions are relaxed over winter, allowing the seeds to germinate in virtually any soil temperature the following spring (Young 1991). Germination consists of the fully formed embryo simply uncoiling, and can be completed within minutes of contact with the proper temperature and as little precipitation as 0.1 in (0.25 mm; Young et al. 1995). Should the embryo desiccate prior to uncoiling, it can return to dormancy until suitable moisture is available (Wallace et al. 1968). Seeds are not persistent, with over 90% germinating in the first year and the remaining seeds typically surviving for less than two years (Boerboom 1995; Young et al. 1995).

Prickly Russian thistle readily hybridizes where sympatric with closely related species. Hybrids tend to show all variations of introgression. A hybrid between *S. tragus* and *S. australis,* identified as *S. ryanii*, is currently found only in California and is a fertile allohexaploid (n=54) with sterile offspring when backcrossed with the founding species (Hrusa and Gaskin 2008, Ayres et al. 2009, Mosyakin 2017). One complex hybrid, nicknamed *Salsola paulsenii* lax because of a lax tip on the perianth, has genetic markers of *S. tragus, S. paulsenii, S. australis,* and unique genetic markers that may represent a lack of genetic sampling or a fourth unknown species. It is of interest because it is hexaploid (2n=54) and might be a new species (Arnold 1972, McGray et al 2008, Ayres et al. 2009). The name *S. gobicola* Iljin was applied to hybrids of S. tragus and S. paulsenii (Rilke 1999).

Distribution: Prickly Russian thistle is an introduced species that can be found in every state in the USA except for Alaska and Florida. Its native range is from northern Africa east through Saudi Arabia, Pakistan, and Nepal and north into South-Eastern Ukraine, northeast China, and southeast Siberia (Rilke 1999). It has been introduced worldwide, most prominently in arid, semi-arid, and disturbed ecosystems in southern Africa, South America, and North America. Prickly Russian thistle is considered present in Australia by some sources, however, it has been determined that all herbarium records from western Australia are actually *S. australis,* and it is suspected that this is the case for all of Australia (Dr. Catherine Borger, personal communications, October 16, 2017).

Prickly Russian thistle was first introduced in the USA in the 1870s in Bonhomme County, South Dakota, in contaminated flax seed imported from Southwestern parts of the former Russian Empire (Ukraine or southwestern Russia; Dewey 1893, 1894). The wind tumbling seed dispersal mechanism meant that the seed could be spread for miles in a single season, with the newly completed transcontinental railroad moving it hundreds of miles. Within a few decades after introduction, it had spread nationwide in one of the fasted plant invasions in United State's history (Rilke 1999).

For current distribution, please consult the Plant Profile page for this species on the PLANTS Web site: https://plants.usda.gov/core/profile?symbol=SATR12

Habitat: Prickly Russian thistle is widely distributed and occurs in most habitats across the United States. It is highly adapted to arid and semi-arid ecosystems. It can be found in disturbed sites in ecosystems as varied as Salt Desert Scrub to Alpine (Howard 1992).

Adaptation

Prickly Russian thistle is a shade intolerant initial colonizer adapted to well drained soils in a wide variety of ecosystems. It is most prolific in arid to semi-arid ecosystems and can be found at elevations from below sea level to 8,500 ft. (0-2590 m). Prickly Russian thistle tends to be less adapted to arid ecosystems below 4,000 ft. (1219 m) than barbwire Russian thistle (*Salsola paulsenii;* Bernau 2018) and can be replaced by it in those areas (Evans and Young 1980). Prickly Russian thistle is adapted to alkaline and saline soils, which allows it to grow in areas that naturally have reduced vegetation. Prickly Russian thistle is a poor competitor and is often replaced by other vegetation after a few years of dominance. In most ecosystems, it relies on frequent disturbance to maintain its population.

Uses

Agriculture: Prickly Russian thistle is extremely water efficient and is known to produce relatively high yields with minimal water resources. As such, there is some potential for hay production in semi-arid and arid ecosystems (Fowler and Hageman 1978, Hageman et al. 1988). Prickly Russian thistle seeds are high in protein and fiber, and seed meal has been shown to increase weight gain in mice trials (Coxworth et al. 1969). Agricultural potential has not yet been realized as Prickly Russian thistle is considered a pest rather than a commodity crop. However, Prickly Russian thistle hay production during the American dustbowl is credited with saving the cattle industry throughout North America (Young 1991, Holmgren et al. 2012). Kansas alone produced 400,000 tons of thistle hay in 1934 (Cave et al. 1936). Currently there is a small niche commercial market selling Prickly Russian thistle as decorations, ornaments, craft material, movie props, and gag gifts.

Pollinators: Despite possessing small flowers, Prickly Russian thistle is a source of pollen for a wide variety of insects; such as bees, flies, moths, and butterflies (Fig 4). It is a larval host plant of the introduced Western Pygmy Blue, *Brephidium exilis*, which is the smallest butterfly in North America.

Figure 4: Prickly Russian thistle pollinators. Left-Right, Top-Bottom: Prickly Russian thistle flower with pollen laden anthers and spilled pollen on bract; Wasp (Crabronidae); European Honey Bee (Apis mellifera); Hover Fly (Syrphidae); Female Sweat Bee (Lasioglossum sp.); Pygmy Blue (Brephidium exilis), underwing view; Pygmy Blue, top-wing view; Male Sweat Bee (Lasioglossum sp.). Identified by Dr. Joseph Wilson (Utah State University) and Dr. Kevin Burls (University of Nevada Reno; Nevadabugs.org). Photo C. Bernau, GBPMC.

Wildlife: Prickly Russian thistle has value for wildlife habitat and food. The plant can provide shelter for small mammals, reptiles, and birds, while it is nutritious and palatable to a wide variety of herbivores. The stems and leaves are eaten by bison, deer, elk, pronghorn, and prairie dogs. Young plants are the most palatable, but standing dead are consumed when softened by moisture. Seeds are readily consumed by a variety of birds and small mammals.

Human Use: Prickly Russian thistle is edible to humans. Young shoots and tips may be eaten raw or cooked like greens (Tull 2013). In his book, The Worst Hard Time, Timothy Egan documents the Lowerys, a family of five that sustained themselves largely on "canned tumbleweeds" during the American dustbowl. The county they lived in, Cimarron County, Oklahoma, "declared a Russian thistle Week, with county officials urging people who were on relief to get out to fields and help folks harvest tumbleweeds." (Egan 2006: pg. 162). Prickly Russian thistle contains small amounts of oxalates, which may cause oxalate poisoning if eaten in abundance. Be absolutely certain of a plant's identity and latest binomial nomenclature prior to consumption.

Landscape Restoration: While Prickly Russian thistle is often an invasive species that can negatively impact rangeland and agricultural landscapes, in some cases it may have value in landscape restoration. That restoration value tends to depend on the presence of mycorrhizal fungi in the soil. Prickly Russian thistle does not form associations with mycorrhizal fungi. Rather, it is infected by the fungi and often killed by it. This fungal food source increases the population of mycorrhizal fungi, allowing it to infect more thistle roots and continue increasing in population. Eventually mycorrhizal associating vegetation will colonize the area in the next stage of plant succession. The increased mycorrhizal fungi population can facilitate the transition and accelerate the rate of re-vegetation (Allen and Allen 1988, Howard 1992, Johnson 1998). In addition, Prickly Russian thistle may act as a nurse plant for native vegetation and protect small plants from grazing. This may be particularly useful in highly degraded landscapes.

Livestock: Prickly Russian thistle can provide forage for cattle, horses, and sheep. The nutritional value of this forage is considered fair when young and is higher once the plant has dried. It is a high source of vitamin A and phosphorous (Howard 1992). It is most palatable in spring when young or in winter when the dead spines are softened by moisture. In some locations, it is viewed as security for livestock when more palatable options are not available. During the dust bowl, Prickly Russian thistle was hayed for cattle feed, as it was one of the few plants that was abundantly available (Cave et al. 1936). Welsh et al.'s 2003, A Utah Flora, describes the plant as poisonous due to oxalates, however, several other sources say it is nutritious and palatable (Dewey 1894, Blaisdell and Holmgren 1984, Howard 1992, Mosyakin 2003, Holmgren et al. 2012). Oxalate and nitrate concentrations are highly variable and highest in younger plants, but are typically below toxic levels. Oxalate poisoning is rare and may be more of a problem for sheep than cattle (Hageman et al. 1988, Boerboom 1995). Nitrate poisoning is also rare. Hageman et al. (1988) evaluated 70 collections of *Salsola* spp. around New Mexico and found 6 collections to have potentially toxic levels (>2%) of nitrate.

Ethnobotany

Salsola species have been used since antiquity in the production of glass and soap. *Salsola* accumulates salts when grown in sodium-rich soils. The plants are burned and the ash mixed with water to create a solution high in sodium carbonate and potassium carbonate. The water is extracted and boiled off, leaving behind sodium carbonate of varying purity. The sodium carbonate is then used to reduce the melting point of sand to make glass, or mixed with oil or fat to make soap. Glass objects dating back to 2500 BC have been found in Syria, and a Babylonian clay tablet dated to 2200 BC listed water, cassia oil, and alkali (sodium carbonate and/or potassium carbonate) as ingredients for soap. This process remained relatively unchanged since antiquity. Kingzett (1877) reported that the quality of ancient Egyptian glass was similar to 19^{th} century crown glass from England.

Prior to 1793, sodium carbonate was produced primarily from the ashes of salt adapted plants. At this time, Spain was a major producer of sodium carbonate, cultivating *Salsola soda* (syn. *Soda inermis*), *Salsola kali*, and *Salsola sativus* (syn. *Halogeton sativus*) for this purpose. The industry was viewed as critical to Spain's economy, and they created laws forbidding the export of seeds; punishable by death (Kingzett 1877).

In 1793, French chemist Nicolas Leblanc invented a new process for creating sodium carbonate through the use of salt, limestone, sulfuric acid, and coal. Shortly thereafter the global production of sodium carbonate shifted away from plant based products.

Status

Weedy or Invasive:

This plant may become weedy or invasive in some regions or habitats and may displace desirable vegetation if not properly managed. Please consult the PLANTS Web site (http://plants.usda.gov/), your local NRCS Field Office, Department of

Natural Resources, Cooperative Extension Service office, state natural resource, or state agriculture department regarding its status and use (e.g., threatened or endangered species, state noxious status, and wetland indicator values).

Planting Guidelines

Prickly Russian thistle can be planted in late fall or early spring. Optimal temperature for germination is 44-50 °F (6.7-10°C), but it can germinate in virtually any temperature once it overwinters. Prickly Russian thistle should be planted in a weed free bed as it is a poor competitor and shade intolerant. Planting depths is optimal less than 1 in (2.5 cm) and should be no deeper than 3 in (7.5 cm; Young et al. 1995). Broadcast seeding can be effective, however, crusting of the soil or soil compaction issues may prevent seedling establishment.

When seed cleaning, it may be difficult to remove the seed from the chaff. One effective method is to sink the seed in Hexane, which results in the chaff floating for easy removal (Coxworth et al 1969)

Management

Prickly Russian thistle management has typically focused on control and eradication for both rangeland and agricultural settings. For both, minimizing disturbance and providing for competing vegetation tends to be an effective strategy. Prickly Russian thistle is shade intolerant and a poor competitor that takes advantage of disturbed sites for growth. If competing vegetation is established and maintained, and disturbance is minimized, then thistle populations may start to decline. In rangelands this may include adjusting grazing rotations, strategic water and mineral placement, or herding strategies. Planting high traffic areas with resilient vegetation may also be useful. Prickly Russian thistle is also palatable when young, so adjusting grazing strategies to take advantage of thistle as forage may be useful.

Minimizing disturbance may be difficult in an agricultural setting where disturbance may be necessary. Planting competing vegetation in field margins and unused acres may reduce Prickly Russian thistle pressure and reduce on-site recruitment from off-site seed sources. Infested fields can be treated by herbicide or carefully timed tilling, though no-till strategies may be worth considering.

Environmental Concerns

Prickly Russian thistle is considered an invasive species and may be listed as Noxious in your area. Please consult the PLANTS Web site (http://plants.usda.gov/) and your state's Department of Natural Resources for this plant's current status prior to planting.

Prickly Russian thistle is able to rapidly colonize harsh environments and disturbed landscapes throughout the United States. It is specifically a problem in arid and semi-arid ecosystems. The tumbling seed dispersal mechanism can spread seed for miles, which makes controlling seed sources difficult. Herbicide may be effective in controlling Prickly Russian thistle, but chemical resistance has been documented.

Prickly Russian thistle is an agricultural pest. It infests fields, reducing crop yields, and it can harbor harmful crop pests. One such pest is the curly top virus, which is transmitted to adjacent vegetation via infected leafhoppers. This virus negatively impacts crops such as sugar beets and tomatoes. In addition, the dead tumbleweeds damage infrastructure such as blocking fences and clogging irrigation ditches and canals (Fig 5).

Figure 5: Infrastructure damage from Prickly Russian thistle. Left: Prickly Russian thistle choking an irrigation canal with debris stacked 3ft (0.9m) high and far into the distance. Right: Prickly Russian thistle piled against a fence for the entire length of the fence. Photo C. Bernau, GBPMC

Prickly Russian thistle is a problem for human safety. As the dead plants tumble they become flying road debris, annually causing several car accidents nationwide. A 2014 outbreak in Colorado became such a nuisance, with clogged roads and buried houses, that two counties declared a state of emergency. The dead tumbleweeds are highly flammable and threaten structures that they rest against (Fig 6). Burning tumbleweeds are particularly problematic as they can bounce over fire lines and escape containment. Prickly Russian thistle pollen negatively impacts human health, with breathing issues and hay fever in some individuals (Wodehouse 1945)

Control

Please contact your local agricultural extension specialist or county weed specialist to learn what works best in your area and how to use it safely. Always read labels and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA NRCS does not guarantee or warranty the products and control methods named, and other products which may be equally effective.

Biocontrol: As of yet, no biocontrol agent has been effective in controlling Prickly Russian thistle. In the 1970s, two moths, *Coleophora klimeschiella* and *C. parthenica*, were released as biocontrols. They have since become naturalized in America, thriving on the Prickly Russian thistle host, but they have been ineffective in controlling it. There are currently an Eriophyd mite (*Aceria salsolae*; Smith 2005, Smith et al. 2009) and two fungal pathogens (*Colletotrichum gloeosporoides*; Bruckart et al. 2004, *Uromyces salsolae*; Hasan et al. 2001) in development as potential biocontrols.

Figure 6: Home in Victorville, California (April 2018), buried by Prickly Russian thistle. This is a nuisance to home owners as well as a significant fire hazard. Photo by James Quigg with the Victor Valley Daily Press.

Herbicide: There are a wide variety of herbicides that have been effective at controlling Prickly Russian thistle (DiTomaso et al. 2013). Preemergence herbicides are best applied in late winter to early spring. Post emergence systemic and broad spectrum herbicides tend to be most effective for young seedlings to mature plants prior to flower. Non-selective herbicides may negatively impact non-target species, which may increase the potential for Prickly Russian thistle establishment and invasion. Prickly Russian thistle is a prolific initial colonizer. It will recolonize treated sites if those sites remain unoccupied by competing vegetation.

Herbicide resistance can develop if a chemical is overused. Herbicide resistant Prickly Russian thistle populations have been reported for a wide variety of chemicals. However, due to *Salsola*'s taxonomic confusion in the literature, it is difficult to know if the reported resistant species is actually *Salsola tragus*. Glyphosate and sulfonylurea resistance has been reported for Prickly Russian thistle in the Pacific Northwest (DiTomaso 2013, Spring 2017), Canadian prairie provinces reported resistance to sulfonylurea and imidazolinone (Morrison and Devine 1994), and resistance to triazines is suspected (DiTomaso 2013). There are several strategies for preventing and managing weed resistance (See Beckie 2006 and Beckie and Harker 2017). Please consult your local agricultural extension specialist or county weed specialist to learn what works best in your area, and always read all herbicide labels.

Mechanical: Hand pulling is effective with small infestations. Mowing is not very effective as it tends to result in low growing plants that still produce seed. Mowing after seed set will spread the infestation.

Prescribed Fire: Prescribed fire is not an effective tool in controlling Prickly Russian thistle. Fire may aid in spreading and increasing Prickly Russian thistle since germination and survival is increased in disturbed sites and it readily colonizes those disturbed sites from off-site sources. Prickly Russian thistle is also a fire hazard, with highly flammable standing dead plants and plants piled on fences, against buildings, and in gullies. There is also the risk of ignited plants tumbling over fire lines, preventing wildfire containment.

Soil health: Prickly Russian thistle does not form mycorrhizal associations with any fungi. Instead, mycorrhizal fungi invade the Prickly Russian thistle's roots causing reduced growth and eventual death. In disturbed landscapes with depleted top soil there is a dearth of mycorrhizal fungi. Strategies that reduce disturbance and improve soil health may work to increase mycorrhizal fungi and thus reduce Prickly Russian thistle populations. In an agricultural system these strategies may include reduced-till or no-till, cover crops, and/or soil amendments that include mycorrhizal fungi.

Targeted Grazing: Targeted grazing may be a useful strategy in controlling Prickly Russian thistle. The plant is considered fair forage with adequate nutrition (Dewey 1894, Blaisdell and Holmgren 1984, Howard 1992, Mosyakin 2003, Holmgren et al. 2012). It is most palatable in early spring before sharp spines form upon flowering. Palatability returns after senescence when the sharp spines are softened by moisture. Heavy grazing prior to flowering may reduce seed production and decrease future thistle recruitment. Some caution is needed as Prickly Russian thistle has oxalates that may become toxic, especially for sheep, if eaten in abundance (Boerboom 1995, Welsh et al. 2003). Nitrate poisoning, while rare, may also be an issue (Hageman et al. 1988).

Tillage: Tillage can be an effective control since the seeds have almost no soil dormancy and typically do not survive or emerge from depths greater than 3 in. This would need to be repeated annually until the seed bank is depleted (<2 years). For best results, delay spring tilling until after the initial flush of Prickly Russian thistle seedlings. Tillage also disturbs the soil, which makes the area more susceptible to reinvasion. It may be necessary to follow up tillage with additional plantings to prevent reinvasion.

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Convolvulus arvensis L.

Field bindweed

Family: Convolvulaceae
Range: Found in all contiguous states and Hawaii.
Habitat: Cultivated crops, gardens, pastures, abandoned fields, and roadsides. Grows best on moist, deep fertile soils. Tolerates poor, dry gravelly soils, but seldom grows in wet soils. Inhabits regions with temperate, Mediterranean, and tropical climates. Found at elevations up to 9000 ft.
Origin: Native to Europe.
Impact: Field bindweed is considered one of the most noxious weeds in agricultural climates in the temperate zone. Plants typically form large patches that are difficult to control due to their extensive root system and long-lived seeds. It is not as important a problem in wildlands and natural areas as it is in croplands.
Western states listed as Noxious Weed: Arizona, California, Colorado, Idaho, Montana, New Mexico, North Dakota, Oregon, South Dakota, Utah, Washington, Wyoming

Field bindweed is a long-lived herbaceous perennial with vine-like stems and an extensive system of deep roots. The glabrous stems twine around other plants for support and are up to 4 ft in length. The leaves are typically dull green and arranged alternately on the stem. Leaves vary in size and shape depending on environmental factors. They are typically 1 to 2 inches in length and vary from arrowhead-shaped to almost round. The root system is an extensive network of vigorous primary and secondary taproots, horizontal creeping roots, and lateral feeder roots. The taproots can grow to a depth of 10 ft or more depending on the available soil moisture and soil depth, while most of the horizontal creeping roots develop in the top 2 ft of soil.

Plants flower from spring to the first frost. The white or pinkish flowers open for one day; they are insect pollinated and self-incompatible. The flowers are axillary, solitary or in cymes of 2 to 4, on stalks about 1 to 3 inches long. The flowers are typically 1 to 2 inches long, funnel-shaped with five fused petals with pleating that is spiraled in the bud.

Field bindweed reproduces sexually through seed and vegetatively through deep horizontal creeping roots and rhizomes. Seeds form in capsules and are dispersed only short distances. One plant can produce up to 500 seeds that can survive buried for 15 to 20 years or more. Most young plants do not produce seed in their first season.

NON-CHEMICAL CONTROL

Mechanical (pulling, mowing, tilling, solarization)	Pulling can be effective on seedlings or young adults but is not effective when the plant has developed a deep, extensive root system. Mowing is not effective due to the low profile of the plant. Intensive cultivation will control new seedlings but spreads the roots and seeds, which may spread the plant. Tilling conducted 8 to 12 days after each emergence throughout the growing season can control field bindweed, but this requires repeated treatments for 1 to 5 years. Deep tillage using shanks down to 3 ft with a cross bar will reduce emergence for a season. Shallow cultivation that kills all above-ground shoots can be effective if repeated several times over a couple of years.

	Solarization is an effective control method, but the black plastic or mulch must be left on the site for 3 to 5 years to eradicate field bindweed.
Cultural	Sheep and cattle have been used to graze field bindweed but this does not affect the roots of the plant and regrowth occurs quickly. Burning is not considered an effective control method as it only removes the aboveground biomass while the root system and seeds are left intact. A combination of burning with other control measures in an integrated approach is more effective.
Biological	Three biological control species have been released in the United States. *Tyta luctuosa* (European field bindweed moth) defoliates field bindweed as a caterpillar. *Chelymorpha cassidea* (tortoise beetle) is native to the United States and feeds on the leaves. *Aceria malherhae* (bindweed gall mite) is a gall mite that has established in several states and feeds on the leaves, stem, and root bud. None of these species has controlled field bindweed in most areas, although the gall mite has shown some success in Colorado.

CHEMICAL CONTROL

The following specific use information is based on published papers and reports by researchers and land managers. Other trade names may be available, and other compounds also are labeled for this weed. Directions for use may vary between brands; see label before use. Herbicides are listed by mode of action and then alphabetically. The order of herbicide listing is not reflective of the order of efficacy or preference.

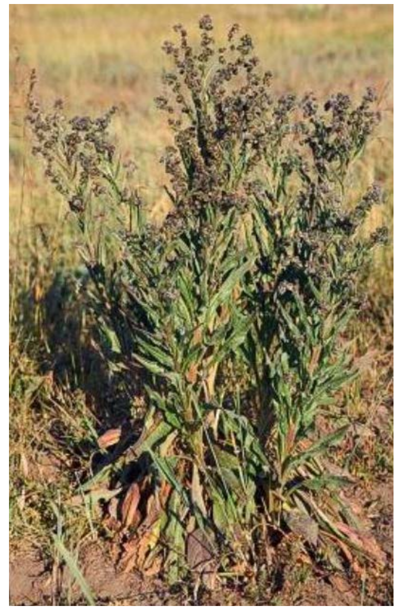
GROWTH REGULATORS	
2,4-D amine Several names	**Rate:** 4 to 6 pt product/acre (1.9 to 2.85 lb a.e./acre) **Timing:** Postemergence at bud stage or in fallow in mid-summer, before bindweed is under moisture stress. **Remarks:** Use 2,4-D to help reduce bindweed stand 60 to 80% and prevent seedling establishment. 2,4-D applications must be made for several years consecutively to prevent regrowth. Avoid drift to sensitive crops.
Aminocyclopyrachlor + chlorsulfuron *Perspective*	**Rate:** 4.75 to 8 oz product (*Perspective*)/acre **Timing:** Postemergence when vegetation is fully developed. **Remarks:** *Perspective* provides broad-spectrum control of many broadleaf species. Although generally safe to grasses, it may suppress or injure certain annual and perennial grass species. Do not treat in the root zone of desirable trees and shrubs. Do not apply more than 11 oz product/acre per year. At this high rate, cool-season grasses will be damaged, including bluebunch wheatgrass. Not yet labeled for grazing lands. Add an adjuvant to the spray solution. This product is not approved for use in California and some counties of Colorado (San Luis Valley).
Dicamba *Banvel*, *Clarity*	**Rate:** 1 to 4 lb product/acre (0.5 to 2 lb a.e./acre) **Timing:** Postemergence when weeds are growing rapidly. Do not apply after bud break. **Remarks:** Recommended rates only suppress field bindweed. Follow-up treatments are generally necessary. Dicamba can be tank mixed with 2,4-D (0.5 to 2 lb a.e./acre) or glyphosate (3 lb a.i./acre). Dicamba is available mixed with diflufenzopyr in a formulation called *Overdrive*. This has been reported to be effective on field bindweed. Diflufenzopyr is an auxin transport inhibitor which causes dicamba to accumulate in shoot and root meristems, increasing its activity. *Overdrive* is applied postemergence at 4 to 8 oz product/acre. Higher rates should be used when treating perennial weeds. Add a non-ionic surfactant to the treatment solution at 0.25% v/v or a methylated seed oil at 1% v/v solution.
Fluroxypyr *Vista XRT*	**Rate:** 22 oz product/acre (7.7 oz a.e./acre) **Timing:** Postemergence when the target plants are growing rapidly. **Remarks:** Provides suppression and not control. Control is reduced if the plants are under stressed growth conditions.
Picloram *Tordon 22K*	**Rate:** 1 to 2 qt product/acre (0.5 to1 lb a.e./acre) **Timing:** Postemergence in the growing season when bindweed is visible. Timing is not critical, but results are most consistent if bindweed is in early bud to full bloom.

	Remarks: Apply as a coarse, low-pressure spray in sufficient volume to cover adequately. Picloram has long soil residual activity. Picloram is a restricted use herbicide. It is not registered for use in California.
Triclopyr *Garlon 3A*	**Rate:** 3 to 4 pt *Garlon 3A*/acre (1.13 to 1.5 lb a.e./acre) **Timing:** Postemergence at bud stage or at summer fallow in mid-summer. **Remarks:** Retreatment is usually necessary for effective control. Triclopyr has no soil residual activity and controls many broadleaf species.
AROMATIC AMINO ACID INHIBITORS	
Glyphosate *Roundup*, *Accord XRT II*, and others	**Rate:** 3 to 4 qt product (*Roundup ProMax*)/acre (3.4 to 4.5 lb a.e./acre) **Timing:** Postemergence when plants are growing rapidly, up to the beginning of seed production. Plants should not be under drought stress at time of application. Application in late summer is also effective. **Remarks:** Cover foliage thoroughly but avoid spray runoff. Repeat treatments may be needed for complete control. Control improves if treated area is tilled 2 to 3 weeks after treatment. Add non-ionic surfactant or 10 to 15 lb of ammonium sulfate. Glyphosate is a nonselective herbicide. It can be tank mixed with 2,4-D or dicamba.
BRANCHED-CHAIN AMINO ACID INHIBITORS	
Imazapic *Plateau*	**Rate:** 8 to 12 oz product/acre (2 to 3 oz a.e./acre) **Timing:** Postemergence, from 25% bloom through fall to rapidly growing bindweed. **Remarks:** For more effective control add 1 qt/acre methylated seed oil. Imazapic is not registered for use in California.
Imazapyr *Arsenal*, *Habitat*, *Stalker*, *Chopper*, *Polaris*	**Rate:** 1 pt product (*Arsenal*)/acre (4 oz a.e./acre) **Timing:** Preemergence or postemergence when plants are growing rapidly. **Remarks:** Imazapyr is fairly nonselective and may injure some desirable species, including grasses and broadleaves. It has fairly long soil residual activity, depending on the site.
Metsulfuron *Escort*	**Rate:** 1 to 2 oz product/acre (0.6 to 1.2 oz a.i./acre) **Timing:** Postemergence to rapidly growing bindweed in bloom stage. **Remarks:** Metsulfuron only suppresses field bindweed. Use a non-ionic or silicone surfactant to improve control. Metsulfuron is not registered for use in California.
Propoxycarbazone-sodium *Canter R+P*	**Rate:** 0.9 to 1.2 oz product/acre (0.63 to 0.84 oz a.i./acre) **Timing:** Postemergence to young, rapidly growing plants. **Remarks:** Propoxycarbazone is a broad-spectrum herbicide that will control many species. It will provide only partial control of field bindweed. Perennial grass species vary in tolerance. A non-ionic surfactant should be added at 0.25 to 0.5% v/v solution.

RECOMMENDED CITATION: DiTomaso, J.M., G.B. Kyser et al. 2013. *Weed Control in Natural Areas in the Western United States*. Weed Research and Information Center, University of California. 544 pp.

Cynoglossum officinale L.

Houndstongue



Family: Boraginaceae
Range: Throughout contiguous United States, except Texas, Oklahoma, Louisiana, Mississippi, and Florida. Found in all western states.
Habitat: Woodlands, pastures, fields, rangeland, forest margins and disturbed sites such as roadsides, sand dunes, abandoned cropland, ditch banks, and urban waste areas. Often on sandy or gravelly soil; colonizes bare soil and under dripline of trees and shrubs, making control difficult.
Origin: Native to Eurasia and accidentally introduced in the late 1800s as a seed contaminant in cereal grain.
Impacts: Houndstongue can be a serious problem in rangeland, pasture and forest settings. The weed is highly invasive and can form dense monotypic stands. Foliage, especially young leaves, and fruits contain pyrrolizidine alkaloids and are liver toxins in all livestock classes, especially horses, when ingested in small amounts over time or in a single large quantity. Plants have a distinctive scent that appears to deter animals from consuming live foliage, thus most poisonings occur when animals consume hay over time.
Western states listed as Noxious Weed: Colorado, Montana, Nevada, Oregon, Utah, Washington, Wyoming
California Invasive Plant Council (Cal-IPC) Inventory: Moderate Invasiveness

Houndstongue is a biennial or short-lived perennial, with erect flower stems to 4 ft tall. The leaves can vary in size, depending on growing conditions, from 4 to 12 inches long and 1 to 3 inches wide. During its first year, the plant stores carbohydrates in a large developing taproot that becomes black and woody by the season's end.

During the second year of growth, plants develop additional leaves followed by an inflorescence, up to 4 ft tall, with reddish-purple flowers, 0.25 inch wide, often horizontal to slightly drooping. The seeds are contained within four distinctive nutlets. Each nutlet is 0.5 inch long, brown or grey-brown and covered with short, hooked prickles that cling to hair, fur, or clothing. Often referred to as "beggar's lice", these nutlets are exceptional dispersal agents. A few of the nutlets drop from the plant, but most stay attached to the persistent inflorescence many months or even years until they are picked up by a passing animal. Houndstongue reproduces solely from seed and a single plant can produce up to 2,000 seeds that can remain viable for 2 to 3 years.

NON-CHEMICAL CONTROL

Mechanical (pulling, cutting, disking)	Digging, pulling, and cutting can be effective if the root crown is severed. Cut young rosettes below the crown in fall or early spring. Clipping or mowing second-year plants close to the ground during flowering can greatly reduce seed production, even in plants which survive and regrow. Mechanical control must be done frequently to have any effect, and is only feasible for small infestations. Houndstongue will not withstand regular cultivation of the young rosettes.
Cultural	Grazing is not practical due to risk of poisoning. Reseeding problem areas with fast growing grasses, and not overgrazing can prevent invasion. Long-term reduction of houndstongue must involve planting competitive plant species. Many improved grass species can be seeded in late fall or winter.
Biological	A biological control program for houndstongue was initiated in 1988. The first North American releases for biological control were the root-mining flea beetle *Longitarsus quadriguttatus* and the houndstongue root-mining weevil, *Mogulones cruciger*, in British Columbia in 1997-1998. *M. cruciger* has become well-

	established in Alberta and has greatly reduced houndstongue there. However, this species has not been approved yet for release in the U.S. Several other insects are being evaluated, although initial results are not as promising as those of the root weevil. The native fungal pathogen that causes powdery mildew (*Golovinomyces cynoglossi*) has been reported to cause some foliar damage to houndstongue in many western states.

CHEMICAL CONTROL

The following specific use information is based on published papers and reports by researchers and land managers. Other trade names may be available, and other compounds also are labeled for this weed. Directions for use may vary between brands; see label before use. Herbicides are listed by mode of action and then alphabetically. The order of herbicide listing is not reflective of the order of efficacy or preference.

GROWTH REGULATORS	
2,4-D Several names	**Rate:** 4 pt product/acre (1.9 lb a.e./acre) **Timing:** Postemergence when plants are growing rapidly. Applications in spring provide the best control. **Remarks:** Selective herbicide for broadleaf species. In areas where desirable grasses are growing around houndstongue, 2,4-D can be used without non-target damage. Good coverage is necessary.
Aminocyclopyrachlor + chlorsulfuron *Perspective*	**Rate:** 4.75 to 8 oz product/acre plus 0.25 to 0.5% v/v surfactant **Timing:** Preemergence or postemergence. **Remarks:** *Perspective* provides broad-spectrum control of many broadleaf species. Although generally safe to grasses, it may suppress or injure certain annual and perennial grass species. Do not treat in the root zone of desirable trees and shrubs. Do not apply more than 11 oz product/acre per year. At this high rate, cool-season grasses will be damaged, including bluebunch wheatgrass. Not yet labeled for grazing lands. Add an adjuvant to the spray solution. This product is not approved for use in California and some counties of Colorado (San Luis Valley).
Aminopyralid + metsulfuron *Opensight*	**Rate:** 2.5 to 3.3 oz product/acre plus 0.25 % v/v surfactant **Timing:** Apply rosette to mid-bolt when plants are actively growing. **Remarks:** Use the higher rate on bolting plants.
AROMATIC AMINO ACID INHIBITORS	
Glyphosate *Roundup*, *Accord XRT II*, and others	**Rate:** Broadcast treatment: 1 to 2 pt product (*Roundup ProMax*)/acre (0.56 to 1.1 lb a.e./acre). Spot treatment: 1.5 to 2% v/v solution *Roundup* (or other trade name) and water to thoroughly wet all leaves. **Timing:** Postemergence when plants are growing rapidly. **Remarks:** Glyphosate is a nonselective systemic herbicide with no soil activity.
BRANCHED-CHAIN AMINO ACID INHIBITORS	
Chlorsulfuron *Telar*	**Rate:** 1 to 1.5 oz product/acre (0.75 to 1.125 oz a.i./acre) plus 0.25 to 0.5% v/v surfactant **Timing:** Preemergence or postemergence. Spring applications are most effective. **Remarks:** Selective herbicide effective for controlling broadleaf weeds and some grasses.
Imazapic *Plateau*	**Rate:** 8 to 12 oz product/acre (2 to 3 oz a.e./acre) plus 0.25 to 0.5% v/v surfactant **Timing:** Preemergence or early postemergence. **Remarks:** Imazapic is a selective herbicide effective for controlling broadleaf weeds and some grasses. Imazapic is not registered for use in California.
Imazapyr *Arsenal*, *Habitat*, *Stalker*, *Chopper*, *Polaris*	**Rate:** 1 pt product/acre (4 oz a.e./acre) plus 0.25 to 0.5% v/v surfactant **Timing:** Preemergence or postemergence. **Remarks:** Imazapyr is a preemergent and postemergence herbicide effective for controlling broadleaf and grass weeds.
Metsulfuron *Escort*	**Rate:** 1 oz product/acre (0.6 oz a.i./acre) plus 0.25 to 0.5% v/v surfactant **Timing:** Early postemergence. Spring applications are most effective. **Remarks:** Selective herbicide for broadleaf species. Can be used safely around desirable grasses. It

	can be used as a premix with aminopyralid (*Opensight*) at 2.5 to 3.3 oz product/acre. Metsulfuron is not registered for use in California.
Sulfometuron + chlorsulfuron *Landmark* XP	**Rate:** 0.75 to 2.25 oz product/acre plus 0.25 to 0.5% v/v surfactant **Timing:** Preemergence or postemergence. **Remarks:** Effective for controlling broadleaf weeds and some grasses. Long soil residual activity.

RECOMMENDED CITATION: DiTomaso, J.M., G.B. Kyser et al. 2013. *Weed Control in Natural Areas in the Western United States*. Weed Research and Information Center, University of California. 544 pp.

Plant Guide

PURPLE LOOSESTRIFE
Lythrum salicaria L.
Plant Symbol = LYSA2

Contributed by: USDA NRCS National Plant Data Center & Louisiana State University-Plant Biology; partial funding from the US Geological Survey and the US National Biological Information Infrastructure

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@ PLANTS

Alternate Names
purple loosestrife, spiked lythrum, salicaire, bouquet violet

Uses
Noxious and highly invasive.

Ethnobotanic: Immigrants might have deliberately introduced *L. salicaria* for its value as a medicinal herb in treating diarrhea, dysentery, bleeding wounds, ulcers, and sores, for ornamental purposes, or as a source of nectar and pollen for beekeepers (Hayes 1979; Jones 1976; Malecki et al. 1993; Stuckey 1980). In states where it is permitted, purple loosestrife continues to be promoted by horticulturists for its beauty as a landscape plant and for bee-forage. Purple loosestrife has been of interest to beekeepers because of its nectar and pollen production. However, honey produced from it is apparently of marginal quality (Feller-Demalsy & Parent 1989).

Horticultural: Horticultural cultivars of purple loosestrife (*Lythrum* spp.) were developed in the mid-1900s for use as ornamentals. Initially, these were thought to be sterile, and therefore safe for horticultural use. Recently, under greenhouse conditions, experimental crosses between several cultivars and wild purple loosestrife and the native *L. alatum* produced hybrids that were highly fertile (Ottenbreit 1991; Ottenbreit & Staniforth 1994). Comparable, subsequent experiments performed under field conditions produced similar results, suggesting that cultivars of purple loosestrife can contribute viable seeds and pollen that can contribute to the spread of purple loosestrife (Lindgren & Clay 1993). Ottenbreit & Staniforth (1994) indicate that such results suggest the need to prohibit cultivars of this species.

Noxiousness: Purple loosestrife grows most abundantly in parts of Canada, the northeastern United States, the Midwest, and in scattered locations in the West. Although this species tolerates a wide variety of soil conditions, its typical habitat includes cattail marshes, sedge meadows, and bogs. It also occurs along ditch, stream, and riverbanks, lake shores, and other wet areas. In such habitats, purple loosestrife forms dense, monospecific stands that can grow to thousands of acres in size, displacing native, sometimes rare, plant species and eliminating open water habitat. The loss of native species and habitat diversity is a significant threat to wildlife, including birds, amphibians, and butterflies, that depend on wetlands for food and shelter. Purple loosestrife monocultures also cause agricultural loss of wetland pastures and hay meadows by replacing more palatable native grasses and sedges (Mal et al. 1992; Thompson et al. 1987).

Having a noxious weed designation in some states prohibit its importation and distribution, but it is readily available commercially in many parts of the country. *Lythrum salicaria* has been labeled the "purple plague." because of its epidemic devastation to natural communities. The species is included on the Nature Conservancy's list of "America's Least Wanted -The Dirty Dozen" (Flack & Furlow 1996).

Impact/Vectors: Naturalized purple loosestrife was relatively obscure from the time of its introduction into North America in the early 1800s (Pursh 1814) until 1930, when a significant increase in populations invading wetlands and pastures was documented (Strefeler et al. 1996b). Reasons for the apparent sudden colonization and spread of this species include the disturbance of natural systems by human activities including agricultural settlement, construction of transport routes such as canals, highways, and perhaps, nutrient increases to inland waters (Mal et al. 1992; Malecki et al. 1993). Absence of natural enemies and ornamental use are other possible causes for purple loosestrife's rapid expansion in North America (Thompson et al, 1987). Recently created irrigation systems in many western states have supported further establishment and spread of *L. salicaria* (Malecki et al. 1993).

The acquisition of adaptive characteristics from native species of *Lythrum* may have enhanced purple loosestrife's invasive success. It will hybridize with *Lythrum alatum*, a widespread, native North American species, in natural settings. Under certain circumstances fertile hybrids are produced that can cross with weedy purple loosestrife. Such interspecific hybrids could serve as a "hybrid bridge" for the transfer of adaptive traits from native *L. alatum* into weedy populations of purple loosestrife (Anderson & Ascher 1993; Strefeler et al. 1996b).

North American naturalized populations of purple loosestrife often form monospecific stands, whereas, in its native Eurasian habitat the species comprises 1-4% of the vegetative cover (Batra et al. 1986; Strefeler et al. 1996b). Purple loosestrife causes annual wetland losses of about 190,000 hectares in the United States (Thompson et al. 1987; Mal et al. 1997). The species is most abundant in the Midwest and Northeast where it infests about 8,100 hectares in Minnesota, 12,000 ha in Wisconsin, over 12,000 ha in Ohio, and a larger area in New York State. Recent distributional surveys document the occurrence of monocultures in every county in Connecticut, where it has been found in 163 wetland locations (Ellis and Weaver 1996; Ellis 1996). At the Effigy Mounds National Monument (EFMO), combined populations of purple loosestrife cover an area of 5 to 10 hectares growing in regularly disturbed sites. This species has a major visual impact on the vegetation of EFMO, and it has the potential to invade and replace native communities endangering the areas' primary resources. (Butterfield et al. 1996). In response to the alarming spread of this exotic species, at least 13 states (e.g., Minnesota, Illinois, Indiana, Ohio, Washington, and Wisconsin) have passed legislation restricting or prohibiting its importation and distribution (Malecki et al. 1993; Strefeler et al. 1996b).

Numerous studies demonstrate the aggressive and competitive nature of purple loosestrife. Fernald (1940) reported a loss of native plant diversity in the St. Lawrence River floodplain following the invasion of purple loosestrife and another exotic, *Butomus umbellatus* L. Gaudet and Keddy (1988) report declining growth for 44 native wetland species after the establishment of *Lythrum*. Among the species tested, Keddy (1990) found that purple loosestrife was the most competitive. His hierarchical rank, arranged from most to least competitive, illustrates the dominance of this invasive weed over many common natives: *Lythrum*>*Cyperus*>*Juncus*> *Eleocharis*> *Mimulus*>*Verbena*. In the Hamilton Marshes adjacent to the Delaware River, annual above-ground production of *L. salicaria* far exceeded all other plant species' production combined.

Purple loosestrife provides little food, poor cover, and few nesting materials for wildlife (Mann 1991). Waterfowl nesting becomes more difficult as clumps of *L. salicaria* restrict access to open water and offer concealing passageways for predators such as foxes and raccoons (Mal et al. 1992). Non-game species, including black terns and marsh wrens, also lose nesting sites when purple loosestrife infests their normal habitats. Balogh and Bookhout (1989a) report that dense stands of purple loosestrife provide poor waterfowl and muskrat habitat. Red-wing blackbirds appear to be the only species to cope with changes in wetlands caused by purple loosestrife (Balogh and Bookhout 1989a). In many areas where *L. salicaria* populations have increased, wildlife species have declined. While some studies may fail to demonstrate cause and affect relationship, they firmly establish circumstantial evidence implicating that *Lythrum*'s invasion is responsible for major changes in wetland communities (Mal et al. 1992).

Purple loosestrife prefers moist, highly organic soils but can tolerate a wide range of conditions. It grows on calcareous to acidic soils, can withstand shallow flooding, and tolerates up to 50% shade. Purple loosestrife has low nutrient requirements and can withstand nutrient poor sites. Under experimental, nutrient-deficient conditions, the root/shoot ratio increased and provided purple loosestrife with a competitive advantage over the native species *Epilobium hirsutum.* Survival and growth of *L. salicaria* was greatly improved by fertilizer treatment and greater spacing between plants. Such results suggest that excessive use of fertilizers and the release of phosphates, nitrates, and ammonia into the environment has enhanced the success of *Lythrum* (Mal et al., 1992; Shamsi and Whitehead, 1977a and b).

Purple loosestrife flowers from July until September or October. Flowering occurs 8-10 weeks after initial spring growth. The lowermost flowers of the inflorescence open first and flowering progresses upward. The capsules mature in the same sequence and the lowermost will ripen and disperse its seeds while flowering is still occurring further up the inflorescence (Butterfield et al. 1996). Thompson et al. (1987) estimated that on average, a mature plant produces about 2,700,000 seeds annually. Purple loosestrife seeds are mostly dispersed by water, but wind and mud adhering to wildlife, livestock, vehicle tires, boats, and people serve also as agent. Seeds are relatively long-lived, retaining 80% viability after 2-3 years of submergence (Malecki 1990). Welling & Becker (1990) investigated seed bank dynamics in three wetland sites in Minnesota and noted a mean density of 410,000 seeds per square meter in the top 5 cm of soil, which was more than all other species combined.

Spring-germinated seedlings have a higher survival rate than summer-germinated seedlings. Seedlings that germinate in the spring will flower the first year, whereas, summer-germinated seedlings develop only five or six pairs of leaves before the end of the growing season. Since its seeds are small, weighing about 0.06 mg each and carry little food reserves, germination must occur under conditions where photosynthesis can occur immediately. A strong taproot develops quickly in seedlings and persists throughout the life of the plant. The aerial shoots die in the fall and new shoots arise the following spring from buds on the rootstocks. Shoots destroyed by fire, herbicides, or mechanical removal can also regenerate from the rootstock. As plants mature, they produce more and more aerial shoots forming very dense clumps of growth. Purple loosestrife can spread vegetatively by resprouting from stem cuttings and from regeneration of pieces of root stock (Mal et al. 1992). Rhizomatous growth is insignificant in purple loosestrife (Shamsi & Whitehead 1974a; Thompson et al. 1987).

Status

Please consult the PLANTS Web site and your State Department of Natural Resources for this plant's current status, such as, state noxious status, and wetland indicator values.

Description

General: Loosestrife Family (Lythraceae). Purple loosestrife is an erect perennial herb that grows up to 2.5 m tall, develops a strong taproot, and may have up to 50 stems arising from its base. Its 50 stems are four-angled and glabrous to pubescent. Its leaves are sessile, opposite or whorled, lanceolate (2-10 cm long and 5-15 mm wide), with rounded to cordate bases. Leaf margins are entire. Leaf surfaces are pubescent.

Each inflorescence is spike-like (1-4 dm long), and each plant may have numerous inflorescences. The calyx and corolla are fused to form a floral tube (also called a hypanthium) that is cylindrical (4-6 mm long), greenish, and 8-12 nerved. Typically the calyx lobes are narrow and thread-like, six in number, and less than half the length of the petals. The showy corolla (up to 2 cm across) is rose-purple and consists of five to seven petals. Twelve stamens are typical for each flower. Individual plants may have flowers of three different types classified according to stylar length as short, medium, and long. The short-styled type has long and medium length stamens, the medium type has long and short stamens, and the long-styled has medium to short stamens. The fruit is a capsule about 2 mm in diameter and 3-4 mm long with many small, ovoid dust-like seeds (< 1 mm long).

Mal et al., 1992, provide a detailed morphological description for L. salicaria. The authors also give details of the tristylous features of this species, as well as an account of its pollen structure and chromosome numbers. The plant's habit, vegetative, and reproductive structures are illustrated with line drawings.

Other species of *Lythrum* that grow in the United States have 1-2 flowers in each leaf-like inflorescence bract and eight or fewer stamens compared to *L. salicaria*, which has more than two flowers per bract and typically twelve stamens per flower. *Lythrum virgatum*, another species introduced from Europe closely resembles *L. salicaria*, but differs in being glabrous (lacking plant hairs), and having narrow leaf bases. The latter two species interbreed freely producing fertile offspring, and some taxonomists (Rendall 1989) consider them to be a single species.

Distribution: Purple loosestrife is a hardy perennial herb with stunning spikes of purple flowers. A native of Eurasia, it was introduced to North America in the early 1800's where it first appeared in ballast heaps of eastern harbors (Stuckey 1980). Most likely seeds were transported as contaminants in the ballast or possibly attached to raw wool or sheep imported from Europe (Cole, 1926; Thompson et al., 1987).

The native range of *L. salicaria* is thought to extend from Great Britain to central Russia from near the 65th parallel to North Africa. It also occurs in Japan, Korea, and the northern Himalayan region. The species has been introduced to Australia, Tasmania, and New Zealand. Since its introduction to North America, this alien plant has spread rapidly into Canada, and throughout most of the United States where it has been reported from all states except Alaska, Florida, Louisiana, and South Carolina. Several factors have contributed to the spread of purple loosestrife such as its potential for rapid growth, its enormous reproductive capacity, lack of natural diseases or predators, its use as an ornamental, and for bee forage (Mal et al. 1992). For current U.S. distribution, please consult the Plant Profile page for this species on the PLANTS Web site.

Control

Please contact your local agricultural extension specialist or county weed specialist to learn what works best in your area and how to use it safely. Always read label and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA, NRCS does not guarantee or warranty the products and control methods named, and other products may be equally effective.

An important consideration in controlling purple loosestrife is its prolific seed production, the ease with which seeds are dispersed, and their ability to remain viable for several years. Also, this plant can spread vegetatively by resprouting from stem and rootstock cuttings. Other considerations in selecting control methods are their detrimental effects on native species and the possibility for reinvasion by purple loosestrife or other exotic species. In addition, native plants of similar appearance should not be subjected to control. Purple loosestrife may superficially resemble plants of the mint family or species of the genera *Epilobium* and *Liatris*. Proper identification is an important consideration in controlling exotic loosestrife.

In natural areas, it may be more feasible to contain populations of purple loosestrife than control them. Large populations extending over one hectare or more will be difficult to eradicate. Containing them may be more feasible. Removing plants or applying herbicides to ones extending beyond the main population can accomplish this. If loosestrife cannot be eradicated, efforts should then concentrate on keeping it from invading the highest quality areas (Butterfield et al., 1996.

Manual, Mechanical, and Replacement: Mowing, burning, and flooding are largely ineffective. Cutting followed by flooding so that cut plant stalks are completely immersed has shown some success. However, flooding may encourage the spread of purple loosestrife seed present in the soil and may result in the regeneration of new plants from stem fragments. Mature plants can withstand short-term immersion. Burning is largely ineffective and it may also stress native plants and subsequently enhance loosestrifes' competitive advantage (Butterfield et al., 1996).

Hand removal is effective for small populations and isolated plants. Younger plants (one to two years old) can be pulled by hand. Plants should be removed, prior to seed set, with minimal disturbance to the soil. Removal after seed-set will scatter the seeds. The entire rootstock must be pulled out because of the potential for regeneration from root fragments. A hand cultivator or similar implement will be helpful for older plants, especially those in deep organic soils. Uprooted plants and broken stems need to be removed from the site since such fragments can re-sprout. Bagging plants for removal will prevent their spread along the exit route. Follow-up treatments are recommended for three years after plants are removed. Clothing and equipment used during plant removal should be cleaned to remove contaminating seeds.

Replacement control has been attempted in several wildlife refuges. Research has shown that Japanese millet (*Echinochloa frumentacea* Link) seedlings outcompete purple loosestrife seedlings. The millet must be planted immediately after marsh drawdown and replanted each year because it does not regenerate well. Replacement seeding trials using native pale smartweed (*Polygonum lapathifolium* L.) showed that it also out-competed purple loosestrife. Replacement methods have obvious limited application in natural areas, but they may provide control of loosestrife populations on bordering property (Butterfield et al. 1996).

Herbicide Control: Various chemical treatments have been used on purple loosestrife with varying success. Many herbicides are not specific to purple loosestrife and may not be specifically licensed for such use. Label directions for application and use according to local, state, and federal regulations must always be observed.

In areas with populations exceeding 100 plants (up to 1.6 ha in size) where hand-pulling is not feasible, application of a glyphosate herbicide to individual purple loosestrife plants provides effective control Glyphosate is available under the trade names Roundup® and Rodeo®. Rodeo is registered for use over open water and is the most commonly used herbicide to control purple loosestrife. Glyphosate is nonselective and can kill desirable plants associated with loosestrife if applied carelessly. Application to the tops of plants alone can be effective and limits exposure of non-target species (Butterfield et al. 1996).

Herbicide treatment should be conducted as early as possible during the manufacturer's recommended time of application in order to kill the plants and prevent seed production. Application is most effective when plants have just begun flowering. Timing is important because seed set can occur if plants are in mid- to late flower. Where possible, the flower heads should be cut, bagged, and removed from the site prior to application to prevent seed set. Rodeo applied as a 1.5% solution (2 oz. Rodeo/gallon clean water) with the addition of a wetting agent, as specified on the label has been shown to provide control. Another option, which may be more effective, is to apply glyphosate twice during the growing season. The plants should be sprayed as described above when flowering has just started and a second time two to three weeks later (Butterfield et al. 1996).

Application of ghyphosate from a vehicle-mounted sprayer is generally necessary in areas with extensive stands of purple loosestrife. The most effective control can be achieved by beginning treatment at the periphery of large patches and working toward the center in successive years. This technique allows native vegetation to re-invade the treated area as the loosestrife in eliminated (Butterfield et al. 1996).

A combination of 2,4-D and Banvel® (dicamba) has been used on a limited basis. This formulation is broadleaf specific and apparently would not hurt the dominants if sprayed in a cattail marsh or communities dominated by rushes, sedges, and grasses. Spraying produces good control once loosestrife has reached 10-15% of its mature growth. Treatment is more effective if repeated once during the growing season (Butterfield et al. 1996).

Biological Control: Several biological control agents have the potential to aid in the control of purple loosestrife. Of 120 species of phytophagous insects associated with purple loosestrife in its natural range in Europe, 14 species were considered host-specific to the target plant. From this group, six species have been selected as the most promising for biological control. These species were a root-mining weevil, *Hylobius transversovittatus* Goeze, which attacks the main storage tissue of purple loosestrife; two leaf-eating beetles, *Galerucella calmariensis* L., and *G. pusilla* Duftschmid, which are capable of completely defoliating the plant; two flower-feeding beetles, *Nanophyes marmoratus* Goeze and *N. brevis* Boheman, which severely reduce seed production; and a gall midge, *Bayeriola salicariae* Kieffer, which similarly reduces seed production by attacking the flower buds. Five of the six species are found throughout its range in Europe and the sixth, *N. brevis*, is restricted to southern Europe (Malecki et al. 1993; Weedin et al. 1996).

The most promising insects appear to be the root-mining weevil, *H. transversovittatus*, and the two leaf-eating beetles, *G. calmariensis* and *G. pusilla*, because of their broad geographic ranges and the amount of damage done to the host plant. In June of 1992, all three species were approved by USDA, APHIS for introduction into the United States. The insects were released in New York, Pennsylvania, Maryland, Virginia, Minnesota, Oregon, and Washington. Releases were also approved in Canada (Malecki et al. 1993).

The two *Galerucella* species successfully over-wintered and began oviposition at all release sites. The other species, *H. transversovittatus*, was proving more difficult to establish, because of its long life cycle and low fecundity. The investigators predict that all three species will become established throughout the North American range of purple loosestrife. Furthermore, *H. transversovittatus* is expected to have the greatest negative impact to *L. salicaria*. However, a combination of various phytophagous insects will provide greater control than any one species. Control of purple loosestrife will be achieved more rapidly in mixed plant communities where competition for space and nutrients is greater. A reduction in the abundance of purple loosestrife to approximately 10% of its current level over about 90% of its range is expected (Malecki et al. 1993).

In order to evaluate the potential of fungus pathogens to control purple loosestrife, a survey was conducted on fungi associated with that plant. During the three year study, 5265 fungal isolates were obtained. Thirty-one taxa were found that had not previously been reported from purple loosestrife. Tests for the pathogenicity to purple loosestrife are being tested (Nyvall 1995).

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For more information about this and other plants, please contact your local NRCS field office or Conservation District, and visit the PLANTS Web site<http://plants.usda.gov> or the Plant Materials Program Web site <http://Plant-Materials.nrcs.usda.gov>

Plant Guide

COMMON REED

Phragmites australis (Cav.) Trin. ex Steud.

Plant Symbol = PHAU7

Contributed by: Idaho Plant Materials Program

R.A. Howard @ USDA-NRCS PLANTS Database

Alternate Names

Alternate Common Names:
Giant reed, Giant reedgrass, yellow cane, Phragmite, Carrizo, Danube grass, Roseau cane

Alternate Scientific Names:
Arundo australis Cavanilles*; A. phragmites* L. *P. berlandieri* Fourn*; P. communis* Trinius

Uses

Livestock
Young plants of common reed are considered very palatable and readily grazed by sheep and cattle (Frankenberg, 1997). Mature plants are tough and unpalatable to livestock and wildlife (Letihead et al., 1971).

Wildlife
Common reed provides excellent cover for wildlife (Skinner, 2010) including hiding cover for deer, rabbits, pheasants and other animals. Common reed also provides nesting cover for wide variety of waterfowl and shoreline birds. Waterfowl eat the seed, and muskrats and nutrias eat the rhizomes and stems (Stubbendieck et al., 2003).

Erosion control
Due to its dense root matrix and coarse stems, common reed has been recommended for shoreline and earthen dam stabilization (USDA NRCS, 1999). It is used by mining operations for stabilizing ditch banks (Walker and Grimes, 1997). Common reed has also been used to trap silt and improve water quality (Frankenberg, 1997). Native grass species are recommended to prevent accidental spread of invasive type common reed (Saltonstall, 2010). Only native common reed should be used, and it should be used only where it can be properly managed.

Status

Common reed is considered an invasive or problematic weed in numerous states. It is a Class C noxious weed in Alabama. It is a banned invasive weed in Connecticut and prohibited in Massachusetts. Common reed is labeled a plant pest and an invasive aquatic plant in South Carolina. In Vermont it is designated as a Class B noxious weed. The state of Washington separated out the non-native genotype from native forms. The invasive form is a class C noxious weed (USDA-NRCS, 2012). Please consult the PLANTS Web site and your State Department of Natural Resources for this plant's current status (e.g., threatened or endangered species, state noxious status, and wetland indicator values).

Weediness

A non-native form of this plant may become weedy or invasive in some regions or habitats and may displace desirable vegetation if not properly managed. Please consult with your local NRCS Field Office, Cooperative Extension Service office, state natural resource, or state agriculture department regarding its status and use. Weed information is also available from the PLANTS Web site at http://plants.usda.gov/. Please consult the Related Web

Sites on the Plant Profile for this species for further information.

Description

General: Grass family (Poaceae). Common reed is a large rhizomatous/stoloniferous cool season grass obtaining heights of up to 4 m (13 ft) with stems averaging 0.5 to 1.5 cm (0.2 to 0.6 in) in diameter. The leaf sheath is open. The ligule is a ring of hairs averaging 1 to 2 mm (0.4 to 0.8 in) in length. Auricles are absent. The leaves are cauline, rolled or flat, and1 to 5 cm (2 in) wide and 10 to 60 cm (4 to 24 in) long (Hitchcock and Cronquist, 1973). The inflorescence is a loose to tight purple tinged 15 to 40 cm (6 to 16 in) long panicle. Spikelets are 12 to 15 mm (0.5 to 0.6 in) long and several-flowered. The rachilla (stem between the florets) is covered with long silky hairs. Glumes are unequal, 3- to 5- nerved and shorter than the florets (Barkworth et al., 2007; Skinner, 2010). Common reed produces male, female and perfect flowers.

Common reed has an extensive system of scaly rhizomes and stolons which allow the plants to spread into dense monotypic stands. Stolons have been measured up to 18 m (60 ft) long (Welsh et al., 2003). The rhizomes produce a dense mat that ranges from 10 cm (4 in) to 2.5 m (8 ft) below the soil surface (Gucker, 2008). Rhizome depth is dependent on site conditions.

There are three taxa or lineages of common reed found in the United States. The broadly distributed native (*P. australis* ssp. *americanus*) covers most of the United States and portions of southern Canada. The Gulf Coast type (ssp *berlandieri*) occurs in southern U.S. and Mexico. The nativity of this subspecies is uncertain. It may be a relatively recent migrant from Mesoamerica (Barkworth et al., 2007). The third type is native to Eurasia and has become invasive in most of the U.S. This taxon has not been officially named to subspecies as its relationships are unclear (Barkworth et al., 2007). It will hereafter be referred to as “invasive type.” Swearingen and Saltonstall (2010) provide images for distinguishing native and exotic forms. The following key from Saltonstall and Hauber (2007) will aid in separation of the three taxa.

1. Ligules 1.0 to 1.7 mm long; lower glumes 3.0 to 6.5 mm long; upper glumes 5.5 to 11.0 mm long; lemmas 8.0 to 13.5 mm long; leaf sheaths falling off with age; culms exposed in the winter, smooth and shiny; rarely occurs in a monoculture
……………..P. *australis* ssp. *americanus* (native lineage)

1. Ligules 0.4 to 0.9 mm long; lower glumes 2.5 to 5.0 mm long; upper glumes 4.5 to 7.5 mm long; lemmas 7.5 to 12.0 mm long; leaf sheaths not falling off with age; culms not exposed in the winter, smooth and shiny or ridged and not shiny; usually occurring as a monoculture……………………..……………………….2

2. Culms smooth and shiny; southern CA, AZ, NM, TX to FL
……..….*P. australis* ssp. *berlandieri* (Gulf Coast lineage)

2. Culms ridged and not shiny; southern Canada from British Columbia to Quebec south throughout the United States……………………*P. australis* (introduced lineage)

Distribution:
Common reed has been described as the most broadly distributed flowering plant in the world (Good, 1974). It is found on every continent except Antarctica (Gucker, 2008). Subspecies *americanus* is native to North America occurring in much of the US with the exception of the southeastern states. A second subspecies *berlandieri* is found along the southern edge of the United States from California to Florida. The non-native form was introduced into North America in the early 19th century (Saltonstall, 2002). It established first along the Atlantic coast and moved westward with the westward human expansion, likely spreading with aid of roadway and railroad development in late 19th and early 20th centuries (Saltonstall, 2002). Common reed (in the broadest sense) is currently found in all states but Alaska (USDA NRCS, 2012). Refer to Saltonstall et al. (2004) for distribution maps of the three lineages. For current distribution, please consult the Plant Profile page for this species on the PLANTS Web site.

Habitat: Common reed occupies a variety of habitats throughout its range including tidal and non-tidal wetlands, marshes, springs, seeps, riparian and lacustrine areas from sea level to 7,000 ft (Hickman, 1993; Welsh et.al., 2003).

Common reed often occupies disturbed sites forming monotypic stands, although the native subspecies are less likely to form dense stands than the invasive type. It is a dominant species in several vegetation types in the US and Canada. Because of its vast distribution, common reed grows in association with a wide variety of species associated with wetland and riparian plant communities.

Ethnobotany

Common reed was used extensively by Native Americans. The plants were used medicinally to treat diarrhea, and made into a poultice to treat boils (University of Michigan, 2012). Several tribes used common reed for building and weaving material from which they made mats, baskets, arrow shafts, flutes and rafts (University of Michigan, 2012). The seed was eaten as food, and the sugary sap from common reed was heated into a ball and dried to be eaten like candy (University of Michigan, 2012).

Adaptation

Common reed is adapted to a wide range of soil conditions from fine to coarse soil types. It is adapted to anaerobic conditions and soils with a pH range of 3.7 to 8.7 (Chabreck, 1972; USDA NRCS, 2012). Common reed is found in highly saline areas including salty tidal marshes and inland saline playas. The invasive type is more tolerant to salinity than the native lineages. Common reed is also adapted to frequent, prolonged flooding; however plant mortality has been reported after 3 or more years with more than 1 m (3 ft) of water (Shay and Shay, 1986).

Establishment
Common reed is established using stem cuttings or rhizomes (Frankenberg, 1997). Rhizomes should be planted into weed free soil that has been tilled to a depth of 10 to 15 cm (4 to 6 in). Recommendations for rhizome spacing vary. Rhizome sections 30 to 46 cm (12 to 18 in) long should be planted 10 to 15 cm (4 to 6 in) deep at a rate of 1 rhizome per foot of row. For shoreline erosion control plantings, a minimum of three rows are recommended at 40 inch row spacing parallel to the shoreline (Texas Agricultural Experiment Station, 1979). Erosion control plantings in northeast Texas reported >50% survival using 30 x 60 cm (12 x 24 in) spacing (Walker and Grimes 1997).

The source area for rhizome collection should share similar characteristics to the planting site. Common reed clones from fresh water sites should not be used in saline situations (Frankenberg, 1997).

Management
For erosion control plantings it is recommended that livestock be excluded from planting sites during establishment (Texas Agricultural Experiment Station, 1979).

Pests and Potential Problems
There are no known pests associated with common reed. For information on potential problems, refer to "Environmental Concerns."

Environmental Concerns
Native forms of common reed can form dense stands in suitable habitat, but they do not have the weedy tendencies of the non-native invasive type. The invasive type poses a threat to native wildlife and vegetation. It will crowd out native plants, alter wetland hydrology and increase fire potential (Keller, 2000; Saltonstall, 2010). It will form dense monocultures spreading by seed into open areas and then spreading rapidly vegetatively. It will spread by rhizome and stem fragments carried on water or via machinery (DiTomaso and Healy, 2003)

Control
Glyphosate treatments are the best option to reduce or control populations of common reed. Young populations with a less-developed rhizome network are more easily controlled than mature stands. Apply herbicides in late summer/early fall after flowering as foliar spray or on cut stumps. Repeat treatments for several years may be necessary to completely kill rhizomes (Saltonstall, 2010).

Other control methods for common reed include deep "root burns". Under typical conditions, common reed will be top killed by fire but the rhizomes will persist. Root burns require substantial litter accumulation and a completely dry rooting area for successful control. Repeated mechanical treatments can decrease growth but not cause plant mortality. Burning followed by flooding can cause mortality by eliminating oxygen transport from above ground leaves and stems to below surface tissues. It is recommended to use fire in conjunction with physical, mechanical and chemical control.

Please contact your local agricultural extension specialist or county weed specialist to learn what works best in your area and how to use it safely. Always read label and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA NRCS does not guarantee or warranty the products and control methods named, and other products may be equally effective.

Seed and Plant Production
Seed production in common reed is variable. DiTomaso and Healy (2003) indicate that viable seeds have not been observed in North American populations. However, seedling emergence has been recorded in seed bank studies in Utah, Washington and elsewhere (Comes et al., 1978; Smith and Kadlec, 1983).

Seed is dispersed by wind and water; however germination from seed is rare and dependent on site conditions. Germination requirements include full light, warm temperatures and moist but not flooded conditions (Gucker, 2003).

Rhizome fragments are the primary means of spread of common reed. Rhizomes can be broken apart by environmental conditions including wave and wind action, or by mechanical disturbance. Non-native rhizome sprouts survived significantly better under higher levels of salinity than native genotypes (Vasquez et al., 2005).

Commercial availability of common reed appears to be limited to rhizomes. Production fields should be planted in rows on sandy soils. Rhizomes can be harvested after a single growing season, but two years of growth are recommended. The USDA-NRCS James E. "Bud" Plant Materials Center in Knox City, Texas reported production yields of 75,000 to 100,000 rhizomes per acre per year (Texas Agricultural Experiment Station, 1979).

Cultivars, Improved, and Selected Materials (and area of origin)
'Shoreline' common reed was selected at the James E. "Bud" Smith Plant Materials Center in Knox City TX

with the intended use of shoreline stabilization and erosion control. It was released in 1978 for its superior wave action control and salinity tolerance. Shoreline was originally collected from a railroad right-of-way at Lawrence, Texas in 1970. Rhizomatous breeder stock is available from the PMC (Alderson and Sharp, 1994).

'Southwind' common reed was released in 1988 by the NRCS Manhattan, Kansas Plant Materials Center and the Kansas State University Agricultural Experiment Station. It is recommended for streambank and shoreline stabilization, rehabilitation of polluted waters, filter strips and constructed wetlands for sewage and sludge treatment. It is recommended for use in eastern Nebraska, Kansas and Oklahoma.

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For more information about this and other plants, please contact your local NRCS field office or Conservation District at http://www.nrcs.usda.gov/ and visit the PLANTS Web site at http://plants.usda.gov/ or the Plant Materials Program Web site http://plant-materials.nrcs.usda.gov.

Saltcedar (*Tamarix ramosissima*)
Ecological Risk Screening Summary

U.S. Fish & Wildlife Service, March 2011
Revised, May 2019
Web Version, 9/20/2021

Organism Type: Plant
Overall Risk Assessment Category: High

Photo: Larry Heronema. Licensed under Creative Commons BY-NC. Available: https://www.inaturalist.org/photos/32999308. (May 10, 2019).

1 Native Range and Status in the United States

Native Range

From Villar García and Beech (2017):

"Southeastern Europe is the westernmost range of the natural distribution of this widespread species. Within the European region, this species occurs in the Balkans, Ukraine, Romania, Moldova and southern European Russia (Baum 1978, Sokolov et al. 1986). The species is recorded in Bulgarian floras, however herbarium material should be checked and it is possible that records may refer instead to *T. smyrnensis* (A. Petrova pers. comm. 2016). In Greece the plant is recorded from seven regions (southern and eastern parts of the mainland, the East Aegean Islands, the Cyclades and the West Aegean Islands; Dimopoulos et al. 2013), though it might have also been confused with *T. smyrnensis* or even *T. nilotica*. There are also two records of this species from FYR Macedonia (V. Matevski pers. comm. 2016) and Serbia (see Villar 2017). Records from European Turkey also refer to *T. smyrnensis*.

Outside Europe it is found from China westwards in most Central Asian countries."

In addition to the locations listed above, GISD (2017) lists *Tamarix ramosissima* as native in Afghanistan, Armenia, Azerbaijan, Iran, Iraq, Kazakhstan, North Korea, South Korea, Kyrgyzstan, Mongolia, Pakistan, Tajikistan, Turkmenistan, and Uzbekistan. CABI (2019) lists *Tamarix ramosissima* as native in Georgia.

Status in the United States

From Kennedy et al. (2005):

"It is currently the dominant tree of riparian forests along streams and rivers throughout the western United States, covering over 600 000 ha of this habitat (DiTomaso 1998), and it is also common along springs and springbrooks throughout this region (Sada et al. 2001)."

GISD (2017) lists *Tamarix ramosissima* as introduced and established in Arizona, Arkansas, California, Colorado, Georgia, Hawaii, Kansas, Louisiana, Mississippi, Montana, Nebraska, Nevada, New Mexico, North Carolina, North Dakota, Oklahoma, Oregon, South Carolina, South Dakota, Texas, Utah, Virginia, Washington, and Wyoming. Additionally, CABI (2019) lists *Tamarix ramosissima* as introduced in Idaho, and Missouri.

According to USDA, NRCS (2019), *Tamarix ramosissima* is a B list noxious weed in Colorado; a Category 2 noxious weed in Montana; a noxious weed in Nebraska, Nevada, North Dakota, South Dakota, Texas, and Wyoming; a Class C noxious weed in New Mexico; a "B" designated weed in Oregon; a Class B noxious weed in Washington; and a quarantine species in Oregon and Washington.

T. ramosissima is listed as a Noxious Weed in California (California Department of Food and Agriculture 2015).

T. ramosissima is in trade in the United States (e.g., Klyn Nurseries 2021; Plant Delight Nursery 2021).

Means of Introductions in the United States

From Kennedy et al. (2005):

"Saltcedar (*Tamarix ramosissima* (Ledeb)), […] was intentionally introduced to arid regions of the western United States in the mid-1800s as an ornamental tree and to prevent soil erosion (Everitt 1980)."

Remarks
From GSID (2017):

"There are few plants that are true genetic species of *Tamarix ramosissima* in infested areas, at least in North America. Most of what is called *T. ramosissima* represents a variety of hybrids, including haplotypes of *T. ramosissima*, *T. chinensis*, *T. gallica* and others (Gaskin and Schaal 2002); it even hybridizes with athel (*T. aphylla*), an evergreen species, in some southwest U.S. locations (Gaskin and Shafroth, in press). The most common genotype in the U.S. is a morphologically cryptic hybrid of *T. ramosissima* and *T. chinensis* not detected in Eurasia (Gaskin & Schaal, 2002)."

From CABI (2019):

"*Tamarix* spp. are difficult to differentiate in the field, and also often in the laboratory. Within their native distribution in the Old World, many species of *Tamarix* can be distinguished by gross morphological characters of the flowers, stems and leaf bracts, or by foliage coloration, time of blooming or shape and size of the plant. However, a group of several species, including *T. ramosissima*, are quite similar and can be distinguished only by taxonomic specialists, and especially by the structure of the androecium, visible only with a hand lens or dissecting microscope."

2 Biology and Ecology

Taxonomic Hierarchy and Taxonomic Standing
According to WFO (2021), *Tamarix ramosissima* Ledeb. is the accepted name for this species.

From ITIS (2019):

Kingdom Plantae
 Subkingdom Viridiplantae
  Infrakingdom Streptophyta
   Super Division Embryophyta
    Division Tracheophyta
     Subdivision Spermatophytina
      Class Magnoliopsida
       Superorder Caryophyllanae
        Order Caryophyllales
         Family Tamaricaceae
          Genus *Tamarix*
           Species *Tamarix ramosissima* Ledeb.

Size, Weight, and Age Range

From CABI (2019):

"*T. ramosissima* is a shrub or shrubby tree, 1-5(-6) m high, […]"

Environment

From GSID (2017):

"*Tamarix ramosissima* is a facultative phreatophyte, meaning that its roots are able to reach deep water tables but it is capable of tolerating periods without access to water (Carpenter 2003)."

From CABI (2019):

"Plants can survive up to 70 days of complete submergence and up to 98 days if part of the canopy is exposed (Warren and Turner, 1975); however, seedlings can be killed by 30 days submergence (Horton et al., 1960; Gladwin and Roelle, 1998)."

"Saltcedars probably grow best in silty alluvial soils but they can grow on a wide range of soil textures from clay to sand, and at relatively high pH levels, and at elevations from sea level up to 2500 m."

Climate

From CABI (2019):

"In the Old World, *T. ramosissima* is adapted to a very wide range of [air] temperatures, from 45°C or more in summer to -20°C or less in winter."

Distribution Outside the United States

Native

From Villar García and Beech (2017):

"Southeastern Europe is the westernmost range of the natural distribution of this widespread species. Within the European region, this species occurs in the Balkans, Ukraine, Romania, Moldova and southern European Russia (Baum 1978, Sokolov et al. 1986). The species is recorded in Bulgarian floras, however herbarium material should be checked and it is possible that records may refer instead to *T. smyrnensis* (A. Petrova pers. comm. 2016). In Greece the plant is recorded from seven regions (southern and eastern parts of the mainland, the East Aegean Islands, the Cyclades and the West Aegean Islands; Dimopoulos et al. 2013), though it might have also been confused with *T. smyrnensis* or even *T. nilotica*. There are also two records of this species from FYR Macedonia (V. Matevski pers. comm. 2016) and Serbia (see Villar 2017). Records from European Turkey also refer to *T. smyrnensis*.

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In addition to the locations listed above, GISD (2017) lists *Tamarix ramosissima* as native in Afghanistan, Armenia, Azerbaijan, Iran, Iraq, Kazakhstan, North Korea, South Korea, Kyrgyzstan, Mongolia, Pakistan, Tajikistan, Turkmenistan, and Uzbekistan. CABI (2019) lists *T. ramosissima* as native in Georgia.

Introduced

From GISD (2017):

"*Tamarix ramosissima* has shown weedy tendencies in both New South Wales and Western Australia, […]"

From CABI (2019):

"*T. ramosissima* has recently invaded South Africa, where it has become weedy and is damaging grazing lands and natural areas (John Hoffmann, University of Cape Town, South Africa, personal communication, 2004; USDA-NRCS, 2007)."

Gullón and Verloove (2015) list *Tamarix ramosissima* as present and naturalized in Spain.

Marlin et al. (2017) list *T. ramosissima* as introduced in Mozambique, Namibia, and Zimbabwe, and present in Botswana.

In addition to the locations listed above, GISD (2017) lists *Tamarix ramosissima* as introduced and established in Argentina, Canada (Manitoba), Mexico; as introduced but present only in containment facilities in Canada (Alberta). CABI (2019) lists *Tamarix ramosissima* as introduced in Mexico, and Italy; and present in Qatar but does not specify native or non-native status. DAISIE (2019) lists *Tamarix ramosissima* as introduced but not established in Austria and France.

Means of Introduction Outside the United States

From GSID (2017):

"Introduced as ornamentals and for windbreaks (Sobhian et. al 1998)."

Short Description

From GSID (2017):

"*Tamarix ramosissima* is a semi-deciduous, loosely branched shrub or small to medium-sized tree. The branchlets are slender with minute, appressed scaly leaves. The leaves are rhombic to ovate, sharply pointed to gradually tapering, and 0.5 - 3.0mm long. The margins of the leaves are thin, dry and membranaceous. Flowers are whitish or pinkish and borne on slender racemes 2-5cm long on the current year's branches and are grouped together in terminal panicles. The pedicels are short. The flowers are most abundant between April and August, but may be found any time of the year. Petals are usually retained on the fruit. The seeds are borne in a lance-ovoid capsule 3-4mm long; the seeds are about 0.45mm long and 0.17mm wide and have unicellular hairs about 2mm long at the apical end. The seeds have no endosperm and weigh about 0.00001 gram. (Carpenter, 2003; Dudley, pers. comm.)."

Biology

From GSID (2017):

"*Tamarix ramosissima* will produce roots from buried or submerged stems or stem fragments. This allows the species to produce new plants vegetatively following floods from stems torn from the parent plants and buried by sediment. Ideal conditions for first-year survival are saturated soil during the first few weeks of life, a high water table, and open sunny ground with little competition from other plants. The seedlings of this species grow more slowly than many native riparian plant species and it is highly susceptible to shading (Carpenter, 2003)."

"*Tamarix ramosissima* is highly fecund. It produces massive quantities of minute seeds that are readily dispersed by wind (Carpenter 2003) but are usually only viable for a few days (Dudley pers. comm.). *T. ramosissima* seeds have no dormancy or after-ripening requirements. Germination can occur almost immediately upon reaching a moist site, and germination conditions are broad, good germination being found from 10 to 35°C [air temperature], but mid-summer seed collections indicated poorer germination rates than those collected in late spring (Young et al. 2004). *T. ramosissima* flowered in two flushes, one in April-May and another in late July in northern Arizona, presumably reflecting availability of spring snowmelt and summer monsoon moisture. This species flowered continuously under favourable environmental conditions but the flowers require insect pollination to set seed (Carpenter 2003)."

From Villar García and Beech (2017):

"*Tamarix* plants have salt glands and exert salt causing salt rain under their shrubs. Therefore, these plants need leaching by freshwater during their life cycle (Akhani 2014)."

From CABI (2019):

"Saltcedars are fire adapted and resprout readily from the basal stem buds after the above-ground plant has burned (Busch and Smith, 1992). Regrowth can reach 3 m high the first year after burning."

Human Uses

From GSID (2017):

"Often planted as an ornamental and to prevent erosion in arid areas. […] and is widely used in the old world for furniture making and for firewood, for tannin extraction, and for cover for livestock (Dudley, pers. comm.). *T. ramossisima* may also be useful for bioremediation, for instance it takes up perchlorate from groundwater, perchlorate being a pollutant derived from jet fuel (Urbansky et al. 2000)."

"*Tamarix ramosissima* is reported being sold in garden centers and nurseries throughout Alberta [Canada]."

From CABI (2019):

"From central Texas to southern California they are used to a minor extent for honey production and somewhat more for pollen and colony maintenance by honeybees. The honey is off-colour and off-flavour and is not of table grade but is used in the baking industry."

Diseases

According to Poelen et al. (2014), *Tamarix ramosissima* is parasitized by *Phoradendron californicum*.

Threat to Humans

No records of threats to humans from *Tamarix ramosissima* were found.

3 Impacts of Introductions

From Cleverly et al. (1997):

"Because of its ability to maintain sap flows at high canopy level transpiration rates (Sala et al. 1996), *Tamarix* can desiccate floodplains and lower water tables (Blackburn et al. 1982). This creates an environment to which *Tamarix* is better adapted than are the native phreatophytes, which are more intolerant of water stress (Busch and Smith 1995) and do not utilize unsaturated soil moisture sources when water tables become depressed (Busch et al. 1992)."

From Lovell et al. (2009):

"In fact, invasive species can directly alter environmental conditions to promote their own establishment and persistence through time. *Tamarix ramosissima* (Tamaricaceae) is such a species; it has caused massive changes to riparian ecosystems and stream bank structures over the last century throughout the southwestern United States (Robinson, 1965; Stromberg, 1998; Pearce and Smith, 2002). Growing as either small trees or dense stands of shoots, *T. ramosissima* can displace or actively outcompete native species of willow (*Salix exigua*) and cottonwood (*Populus deltoides*) in the western United States (Robinson, 1965)."

From Kennedy et al. (2005):

"Saltcedar removal was a highly effective restoration tool because it led to significant increases in pupfish abundance and significant decreases in crayfish abundance. Further, the response of speckled dace (increase) and mosquitofish [also non-native in this system] (decrease), though not statistically significant, was also consistent with the restoration goal of increasing native fish abundance and decreasing exotic consumer abundance. Algal productivity increased significantly following saltcedar removal (Kennedy and Hobbie 2004), and stable isotope analysis provides conclusive evidence that this drove significant increases in pupfish and screw snail density, both of which are strongly dependent on algae-derived carbon. Saltcedar removal had a significant negative impact on crayfish density during the winter sampling period because crayfish consume saltcedar leaf litter and are not strongly dependent on algae-derived carbon."

From Marlin et al. (2017):

"A preliminary study of arthropods, identified mainly to morphospecies, associated with *T. usneoides* and *T. ramosissima* growing together at the Vaal River Mining Operations, Gauteng province, South Africa, showed relatively low species richness and abundance on *T. ramosissima* (Buckham 2011). This suggests that the majority of indigenous insects which utilise the indigenous *T. usneoides* as a host, are not able to use the alien *T. ramosissima* as a host, […]"

From GSID (2017):

"Kennedy and Hobbie (2004) observe that the spread of salt cedar has shifted reaches of Jackrabbit Spring in the Ash Meadows National Wildlife Refuge, from a system based on autochthonous production to dependence on allochthonous inputs, with salt cedar sites having lower temperature-adjusted chlorophyll and macrophyte production rates and greater allochthonous inputs than virtually all native and cleared sites. The effects of the spread of salt cedar on macrophyte and algal inputs probably resulted from dense shading by the trees, because stream nitrogen and phosphorus concentrations were not affected by the large salt cedar stands or by its removal (Kennedy, 2002)."

"Control of the flood regime by large dams and river channelisation has removed the dominant fluvial processes of the lower Colorado River s [*sic*] riparians areas, leading to the desiccation and salinisation of riparian habitats and an almost complete lack of native gallery forest regeneration. These conditions facilitated invasion by the exotic tree *T. ramosissima* and its displacement of native Fremont cottonwood and Goodding s [*sic*] willow trees (Ellingson and Andersen 2002)."

"*Tamarix ramosissima* and *T. chinensis* have been declared as Category 1 weeds in Northern, Eastern and Western Cape, category 3 weeds in other parts of South Africa. (Category 1 Plants. […] These plants may not occur on any land or inland water surface other than in a biological control reserve. Except for the purposes of establishing a biological control reserve, one may not plant, maintain, multiply or propagate such plants, import or sell or acquire propagating material of such plants except with the written exception of the executive officer. Category 3 Plants. The regulations regarding these plants are the same as for category 1, except that plants already in existence at the time of the commencement of these regulations are exempt, unless they occur within 30 metres of a 1:50 year flood line of river, stream etc) (SANBI, 2001)."

From CABI (2019):

"The list of plants, both indigenous and introduced, that are displaced by saltcedar invasions would include virtually every plant known in riparian areas of the western USA and northern Mexico. The invasion and domination of native riparian plant communities most often follows the recession of flood waters or wildfires, which kill the native plants, and then allows the saltcedar seedlings to establish without competition."

“In a 3-year comparison of insect populations on saltcedar compared with native willows (*Salix* spp.), poplar/cottonwood (*Populus* spp.) and seepwillow baccharis (*Baccharis salicifolia*) in northwestern and southwestern Texas and southern New Mexico, USA, both species diversity and populations of native herbivorous insects (immature specimens and adults) were significantly greater on the native plants than on saltcedar. […] Although many nectar and pollen feeding insects were abundant on saltcedar flowers, all of these developed as immatures on nearby native plants.”

“The greatest economic losses caused by saltcedars relate to the large losses of streamflow and ground water, especially in arid areas of the western USA and in northern Mexico. This entire area is experiencing severe water shortages for agricultural irrigation and for municipal use. […] The US Bureau of Reclamation in Albuquerque, New Mexico estimates that one-third of the total amount of water allowed to be taken from the Rio Grande is used by saltcedar (S Hansen, US Bureau of Reclamation, Albuquerque, New Mexico, USA, personal communication, 2002). Zavaleta (2000) estimated water losses from saltcedar at US $133 to 285 million annually, and this does not include losses in Mexico. Saltcedar also reduces water quality by increasing the salinity of stream flow and ground water.”

“The increased frequency of wildfires caused by saltcedar damages fences and sometimes farm buildings, other buildings and kills livestock. These damages are probably relatively small and economic analyses are not known.”

“Saltcedars cause economic losses by reducing the utilization of parks and natural areas by hunters, fishers, campers, bird watchers, wildlife photographers and others (USDI Fish and Wildlife Service, 1988). In an attempt at determining the proportion of losses caused by saltcedar, DeLoach (1989) estimated losses to these non-consumptive, recreational-type uses in Arizona, USA, at US$29.5 million and in New Mexico at probably US$15.8 million annually, and twice that if the value of the time of the participants were included.”

“Dense thickets of saltcedar along streams cause increased sedimentation, bank aggradation, narrowing and deepening of channels, filling in of backwaters, modification or elimination of riffle structure, overgrowth of sand and gravel bars, and changes in turbidity and temperature of the water. Channels are sometimes completely blocked with debris and overbank flooding is more severe (Busby and Schuster, 1971; Burkham, 1972, 1976; Graf, 1978). Saltcedars are probably the greatest users of scarce groundwater in the infested desert ecosystems (reviewed by DeLoach et al., 2000). Estimates of groundwater use from a number of experiments averaged 1676 mm per year along the lower Colorado River near Blyth, California, USA (the hottest area, lowest elevation and longest growing season in the southwestern USA) to 940 mm per year along the middle Rio Grande, New Mexico at a higher elevation and shorter growing season.”

“Saltcedars increase the natural salinity level by using saline ground water and excreting the excess salts through leaf glands. The salt then drips to the soil surface or falls with the foliage in the autumn, forming a layer of saline litter and soil under the trees in which only saltcedar can survive.”

“The dry foliage and twigs that accumulate under the deciduous saltcedars are highly flammable. Saltcedar thickets burn more intensely and more frequently than native riparian plant communities in North America (which only rarely burn) (Agee, 1988). This situation, like that of soil salinity, is further worsened by the additional interaction with altered hydrologic cycles below dams, preventing the natural spring floods from washing out the accumulated litter (DeLoach et al., 2000).”

“In North America, the greatest direct negative environmental impact of the saltcedar invasion is the displacement of native riparian plant communities by dense thickets of saltcedar, that now cover an estimated 800,000 ha of prime bottomlands along major rivers, small streams and lakeshores. Along many major rivers, saltcedar thickets occupy 50-60% of all the vegetative area (summarized by DeLoach, 1991) and 93% on the Pecos River of Texas and New Mexico (Hildebran[d]t and Ohmart, 1982).”

“The most seriously affected plants are the obligate phreatophytic trees and shrubs, especially poplars/cottonwoods (*Populus* spp.), willows (*Salix* spp.), screwbean mesquite (*Prosopis pubescens*), seepwillow baccharis (*Baccharis salicifolia*) and a few others. The large (to 20 m tall) stands of poplar/cottonwood trees which formally comprised the dominant upper canopy in most areas, are now reduced to small, scattered trees except for one remaining stand of ca. 115 ha at the confluence of the Bill Williams river of Arizona and the Colorado River. Willows, screwbean mesquite and seepwillow baccharis also have been displaced by saltcedars but to a somewhat lesser extent because they are less sensitive to some of the environmental changes than are poplars/cottonwoods. Some other important plants have been harmed to a lesser extent than the obligate phreatophytes, such as honey mesquite and velvet mesquite (*Prosopis glandulosa* and *P. velutina*) and quailbush (*Atriplex lentiformis*) which can also occupy higher terraces (Wiesenborn, 1995).”

“One effect of the saltcedar invasion has been to cause some rare plant species to become more rare and some to become endangered. For example, the threatened Pecos sunflower (*Helianthus paradoxus*) was believed to be extirpated from areas of the Pecos River until saltcedar was cleared, and then it reappeared as a common plant.”

“The major effect of the saltcedar invasion on native plant communities has been the drastic degradation of wildlife habitat (Kerpez and Smith, 1987, and reviewed by DeLoach et al., 2000). The population of all birds found in saltcedar on the lower Colorado, USA, was only 39% of the levels in native vegetation during the winter and 68% during the rest of the year; and the number of bird species found in saltcedar was less than half that in native vegetation during the winter (Anderson and Ohmart, 1977, 1984). Saltcedar was the most important negatively correlated variable identified with bird populations (Anderson and Ohmart, 1984). Frugivores, granivores and cavity dwellers (woodpeckers, bluebirds and others) are absent, and insectivores are reduced in saltcedar stands (Cohan et al., 1979). At Camp Cady in southern California, the bird population was only 49% as great in saltcedar as in cottonwood/willow/mesquite (Schroeder, 1993). Bird preference for saltcedar was much lower than for native vegetation along the middle Rio Grande, Texas (Engle-Wilson and Ohmart, 1978) and somewhat lower on the middle Pecos River (Hildebrandt and Ohmart, 1982). Recent surveys at release sites in northwestern Texas showed that both the number of birds and the number of bird species per point count were twice as great in 2003 (a dry year) in native vegetation compared to near pure saltcedar stands. In 2004 (a wet year), populations were 37% greater in the native vegetation (T Robbins and K Johnson, USDA-ARS, Temple, Texas, USA, unpublished data, 2002-2004)."

"Populations of game animals, furbearers and small rodents are lower in saltcedar than in other vegetation types on the Rio Grande of western Texas (Engle-Wilson and Ohmart, 1978) and on the Pecos of New Mexico (Hildebrant and Ohmart, 1982). In Big Bend National Park, Ord's kangaroo rat and beavers have been nearly eliminated because of the saltcedar invasion (Boeer and Schmidly, 1977)."

"Along the Gila River near Florence, Arizona, Jakle and Gatz (1985) trapped three- to five-times as many lizards, snakes and frogs in native vegetation types as in saltcedar."

"DeLoach and Tracy (1997) and Anon. (1995) reviewed 51 listed or proposed threatened and endangered species that occupy western riparian areas infested by saltcedar. These included two mammals, six birds, two reptiles, two amphibians, one arthropod and four plants. Some 34 species of threatened and endangered fish are found in saltcedar infested areas. Their habitat is seriously degraded by reduced water levels, modified channel morphology, silted backwaters, altered water temperature, and probably by reduced and modified food resources. Several of these threatened and endangered species may utilize saltcedar to some extent, but not to a degree that would make it appear important to them or as valuable as the native vegetation it has replaced (Anon., 1995)."

"A very unusual wildlife situation involves the interaction between the proposed biological control programme and the southwestern willow flycatcher (*Empidonax trailii extimus*) that was listed as endangered in 1995 and that had begun nesting in saltcedar in Arizona (though little or none in neighbouring states) (DeLoach et al., 2000). Extensive population surveys during several years throughout its breeding range revealed that most of the known mortality factors of the flycatcher could be made worse by its association with saltcedar. Yet, in spite of these losses, the birds almost entirely selected saltcedar trees for nesting even in sites where abundant healthy native willows were present. Apparently, the birds had developed a very high preference for the almost ideal branching structure of saltcedar for nest placement."

"*T. ramosissima* is […] a declared noxious weed in South Africa, category 1 in Northern, Eastern and Western Cape, category 3 in other parts of South Africa."

4 History of Invasiveness

Tamarix ramosissima is native to much of Eurasia. It is introduced and established in many U.S. States, and there are numerous regulations on the plant. *T. ramosissima* has been introduced for ornamental and soil erosion purposes. It is also established outside of its native range in Australia, many places in Africa, Canada and Mexico. Impacts are well established and include altering hydrology and stream banks, competition with native plants, impacts to water quality, and increased fire risk. The history of invasiveness for this species is classified as High.

5 Global Distribution

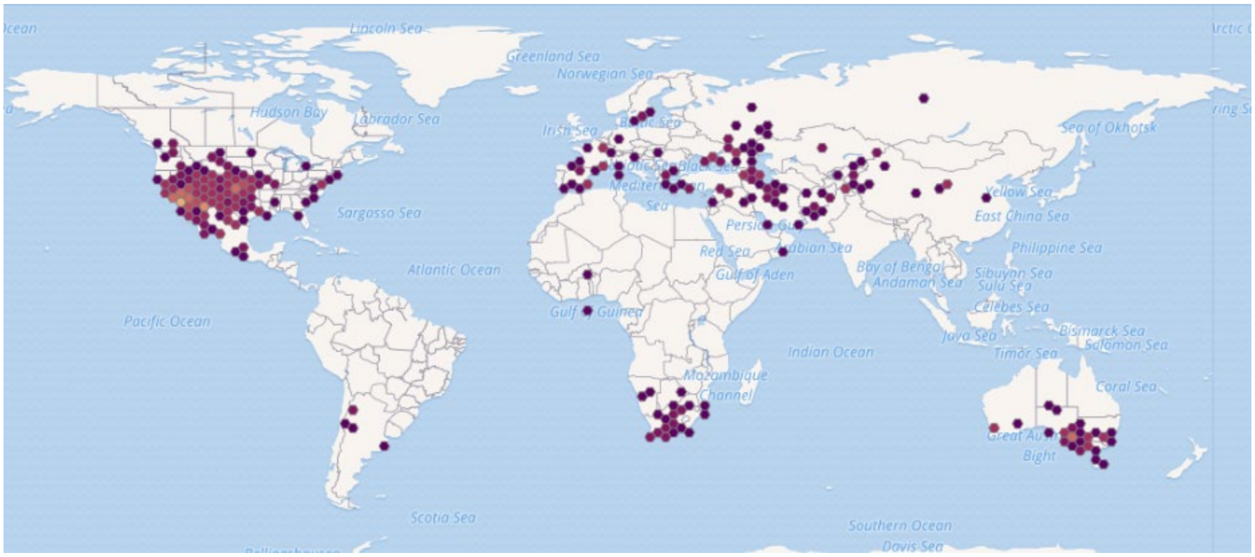

Figure 1. Known global distribution of *Tamarix ramosissima*. Map from GBIF Secretariat (2019). The locations in the ocean to the west of Africa and in Burkina Faso (western Africa) were not used to select source points for the climate match. The specimens those records are based on were collected in New Mexico and Utah. The location in southern Ontario, Canada was not used to select source points for the climate match, the observation information for that location indicates that the observer was unsure about the identification and there are no other records in Ontario. Locations in Bulgaria, Greece, and European (western) Turkey were not used to select source points for the climate match. According to Villar García and Beech (2017), those observations are most likely of other *Tamarix* spp. and not *T. ramosissima*. Locations in France and Austria were not used to select source points; *T. ramosissima* is not established in those countries (CABI 2019).

Due to some apparent confusion and difficulty in *Tamarix* species identification (see Villar García and Beech 2017; CABI 2019 and references therein), locations in figure 1 outside the native range, where presence could not be confirmed with another source and not in close proximity to a verified location were not used to select source locations for the climate match. These locations not used are in Sweden, Germany, and Oman.

6 Distribution Within the United States

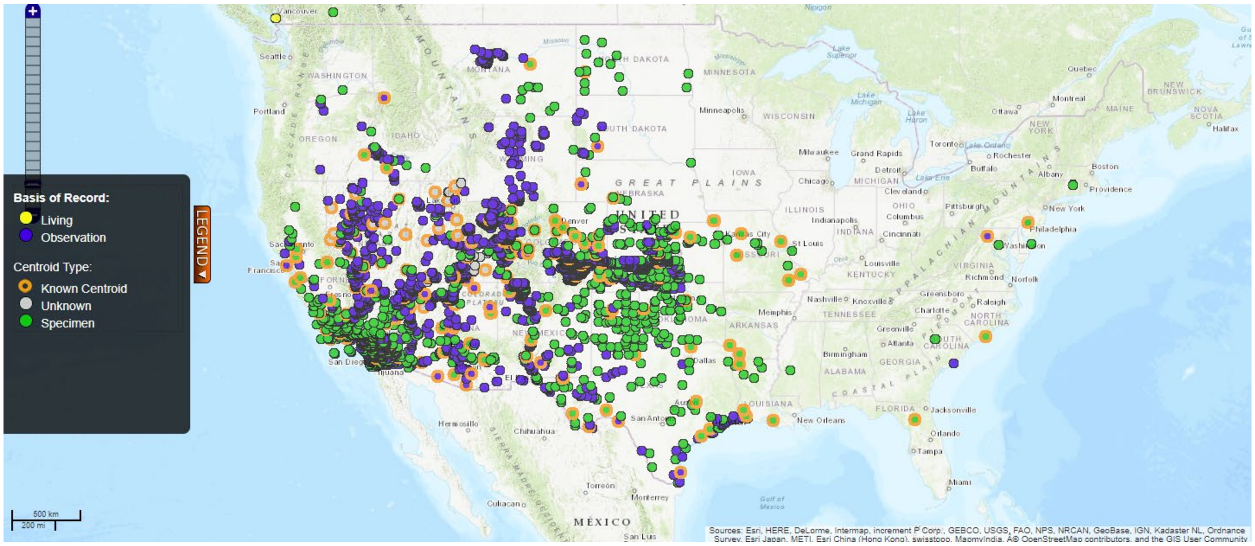

Figure 2. Known distribution of *Tamarix ramosissima* in the United States. Map from BISON (2019).

The following locations were not used to select source points for the climate match. Record information indicates that the record in Connecticut (figures 1, 2) it may be a captive specimen (GBIF Secretariat 2019). The specimens in New Jersey and Washington D.C. (figures 1, 2) belong to *Tamarix* spp. other than *Tamarix ramosissima* (GBIF Secretariat 2019). Locations near coastal North Carolina (figures 2, 3) are specimens in captivity (EDDMapS 2019). Locations in Florida (figures 1, 2) are held in captivity (GBIF Secretariat 2019).

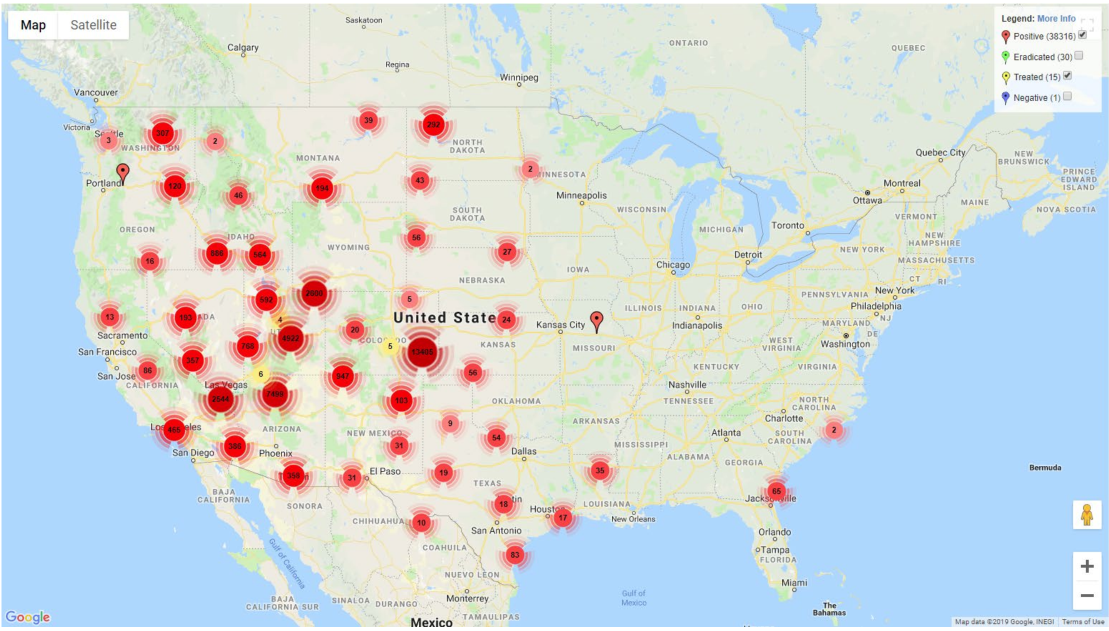

Figure 3. Additional data on the known distribution of *Tamarix ramosissima* in the United States. Map from EDDMapS (2019).

7 Climate Matching

Summary of Climate Matching Analysis

The climate match for *Tamarix ramosissima* and the contiguous United States was high. There were areas of medium match in the Northeast, eastern Great Lakes and in patches down through the Appalachian Mountains. Southern Florida and the coastal areas of the Pacific Northwest also had medium matches. There were small areas of low match in the Olympic Peninsula, the northern Northeast, and a small area of the southern Appalachian Mountains. Everywhere else had a high match. The overall Climate 6 score (Sanders et al. 2018; 16 climate variables; Euclidean distance) for contiguous United States was 0.963, high. (Scores of 0.103 and greater are classified as high.) All States had a high individual climate match except for Maine, New Hampshire, and Rhode Island which had medium individual climate matches.

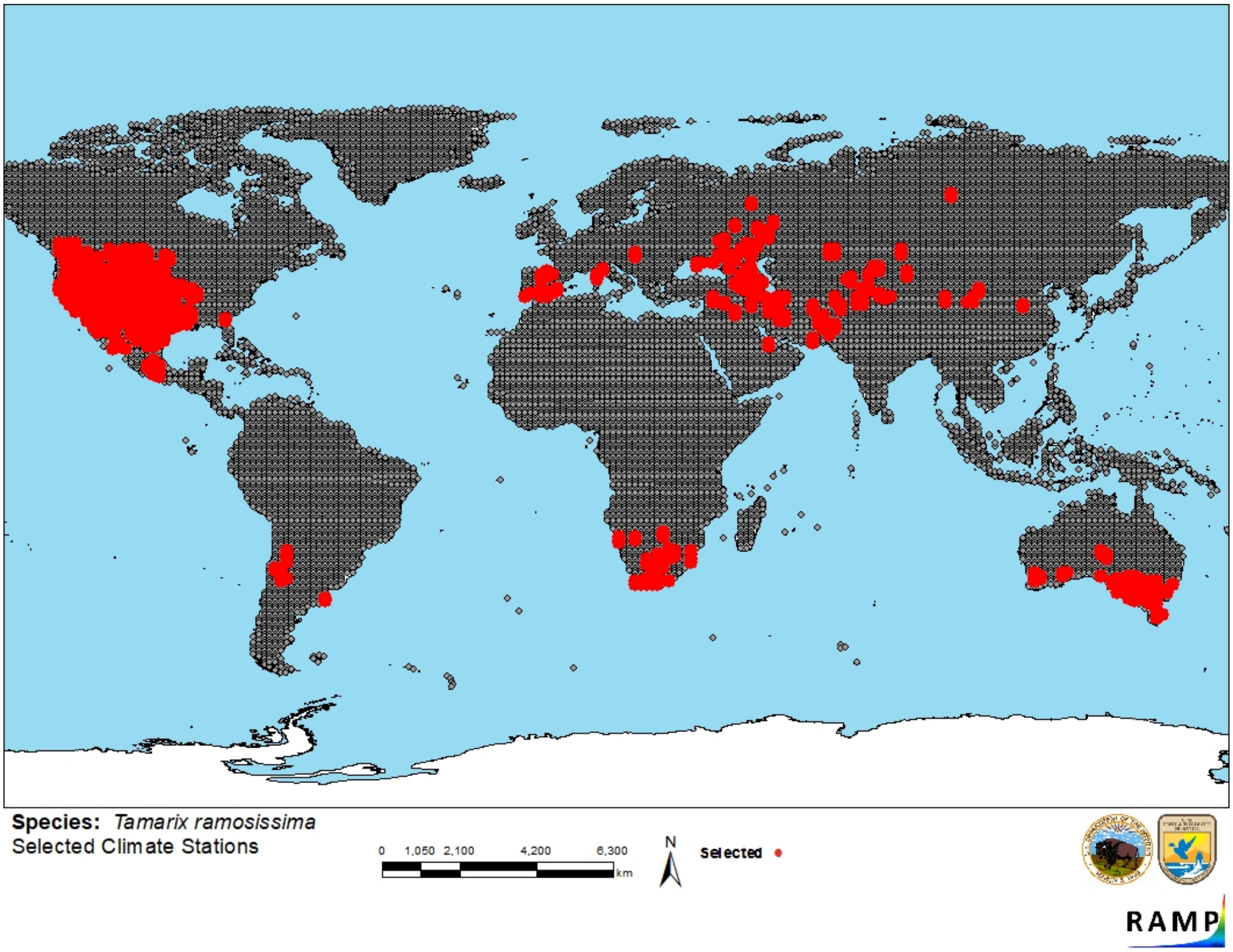

Figure 4. RAMP (Sanders et al. 2018) source map showing weather stations on all continents selected as source locations (red) and non-source locations (gray) for *Tamarix ramosissima* climate matching. Source locations from BISON (2019), EDDMapS (2019), and GBIF Secretariat (2019). Selected source locations are within 100 km of one or more species occurrences, and do not necessarily represent the locations of occurrences themselves.

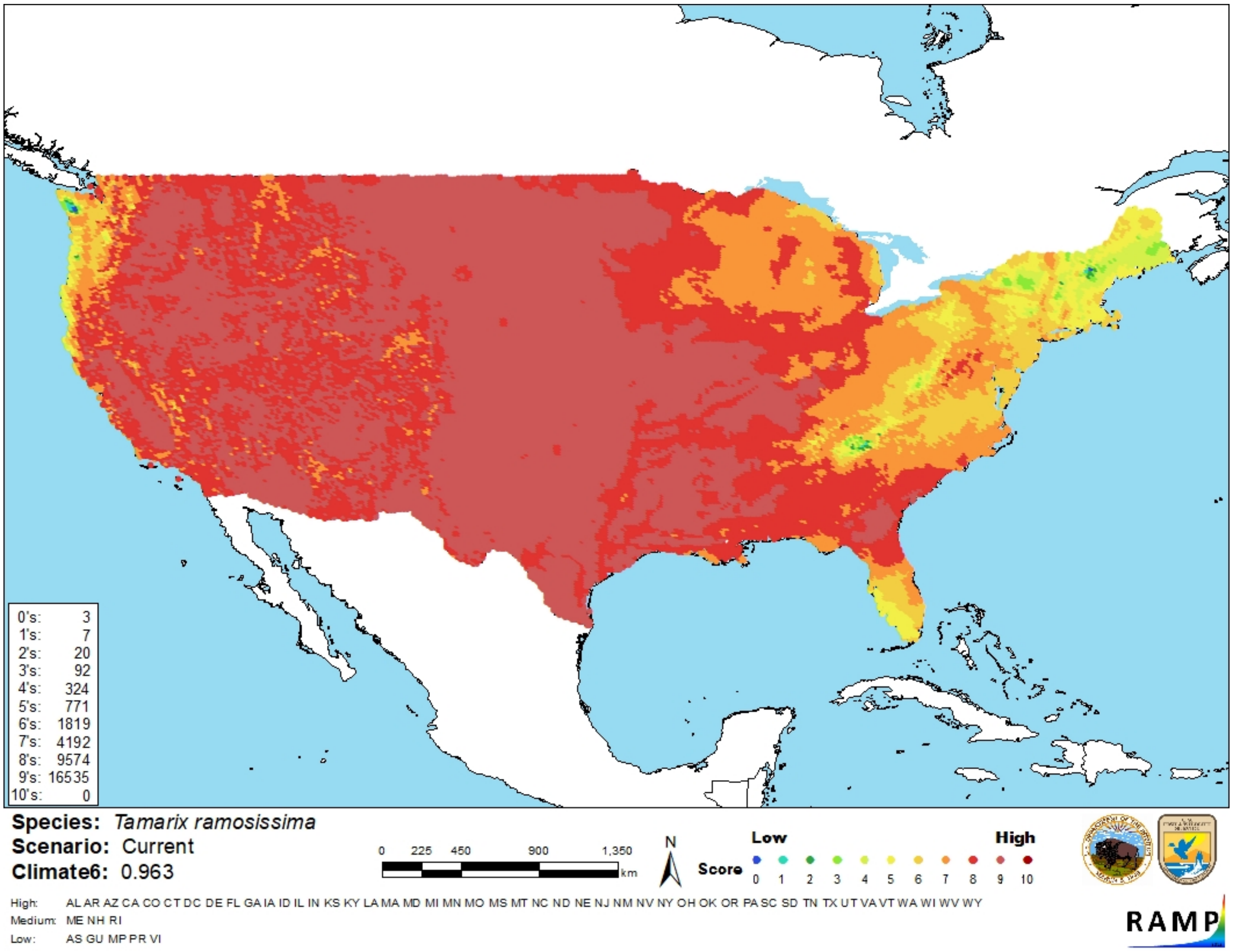

Figure 5. Map of RAMP (Sanders et al. 2018) climate matches for *Tamarix ramosissima* in the contiguous United States based on source locations reported by BISON (2019), EDDMapS (2019), and GBIF Secretariat (2019). Counts of climate match scores are tabulated on the left. 0/Blue = Lowest match, 10/Red = Highest match.

The High, Medium, and Low Climate match Categories are based on the following table:

Climate 6: (Count of target points with climate scores 6-10)/ (Count of all target points)	Overall Climate Match Category
0.000≤X≤0.005	Low
0.005<X<0.103	Medium
≥0.103	High

8 Certainty of Assessment

Information on the biology, invasion history and impacts of this species is substantial, including considerable peer-reviewed literature. There is enough information available to identify the risks posed by this species. Certainty of this assessment is high.

9 Risk Assessment

Summary of Risk to the Contiguous United States

Saltcedar (*Tamarix ramosissima*) is a semi-deciduous shrub native to parts of Eastern Europe and Asia. It has been used as an ornamental and the wood has been used for various purposes including as fuel and for firewood. *Tamarix ramosissima* are tolerant of flooding and saline substrates. They have salt glands on the leaves that will excrete the excess salt which will then 'rain' onto the substrate below the plant. The history of invasiveness is classified as High. It has been introduced around the world as an ornamental, to create windbreaks, or to prevent erosion. This species has become established in many countries, including across the western half of the United States. This species, when introduced, has initiated a number of hydrological and ecological changes including reductions in plant and animal biodiversity, replacement of native riparian trees, and altering bank structure and geomorphological processes. The climate match for *T. ramosissima* is very high. There are few areas that had a medium match, mainly in northern areas, and even fewer locations of low match. The certainty of assessment is high. There is a large body of peer-reviewed literature about the species and its invasion history in the United States. The overall risk assessment category is high.

Assessment Elements

- **History of Invasiveness (Sec. 3): High**
- **Overall Climate Match (Sec. 6): High**
- **Certainty of Assessment (Sec. 7): High**
- **Remarks/Important additional information:** No additional comments.
- **Overall Risk Assessment Category: High**

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United States Department of Agriculture

Natural Resources Conservation Service

Plant Guide

RUSSIAN OLIVE

Elaeagnus angustifolia L.

Plant Symbol = ELAN

Common Names: Oleaster, trebizond-date

Scientific Names: Synonyms are *Elaeagnus angustifolia* var. *orientalis*

New stems and leaves of Russian olive are covered with silvery-gray hairs; older stems are smooth and reddish brown; immature fruits are silver. Photo by NRCS.

Description

General: Russian olive is a large, thorny, perennial deciduous tree or small shrub that usually grows 10 to 25 feet tall. It is a non-native, invasive species. The alternately arranged leaves are 1 to 4 inches long and 0.5 to 1.5 inches wide with smooth edges. The upper leaf surface is green-gray while the lower surface is silver. The numerous thorns are 1 to 2 inches long and arranged alternately on stems. The flowers have four yellow sepals that resemble petals. They appear bell-shaped and are arranged in clusters (USDA, NRCS, 2019). New stem growth is covered with hairs that give it a silvery-gray appearance. Stems become smooth and reddish brown with age. Mature trunks can have a circumference up to 20 inches with dark gray, ridged bark. Roots can grow to depths of 40 feet. Russian olive has clusters of 0.5 inch, hard, olive-shaped fruit that each contain one seed. Immature fruits are silver and ripen to tan or brown.

Distribution: Russian olive is native to Europe and western Asia. It was introduced to the United States in the early 1900s and became widely distributed due to its extensive use as an ornamental species in drier regions of the Great Plains and Rocky Mountains. Russian olive has been used in shelterbelts, windbreaks, wildlife habitat plantings, and as an ornamental. Russian olive has escaped cultivation and become invasive. Plants thrive and spread along riparian corridors, irrigation systems, pastures, saline affected areas, and some wetland sites. For current distribution, please consult the Plant Profile page for Russian olive (*Elaeagnus angustifolia*) on the PLANTS Website.

Habitat: Russian olive thrives under a wide range of moisture and soil conditions. It grows effectively on poor mineral soils because of symbiotic nitrogen-fixing bacteria in the roots (USFS, 2014). It prefers areas where the water table is near the soil surface in riparian areas, flood plains, valley bottoms, irrigation ditches, springs, and sub-irrigated pastures and grasslands. It also grows well in uplands that receive as little as 8 inches of annual precipitation such as along roads, railways, and fence lines. It grows in sandy, silty or loamy soils with low fertility and low to moderate soluble salt concentrations and is described as tolerant to very tolerant of salt injury. It occurs from sea level to about 8,000 feet of elevation and is shade tolerant (USDA, NRCS, 2019).

Adaptation

Until the 1970s, Russian olive was one of a few commercially available medium-height trees used for dryland windbreaks and shelterbelts because of its ease of establishment and value for wildlife. More recently, the availability of tree species for dryland conservation practices has improved. Unfortunately, Russian olive escaped cultivation by the 1950s, and has become a widespread threat to plant communities in riparian areas, grasslands, irrigated pastures, and haylands. Russian olive can become the dominant species as it forms dense, monotypic stands that can prevent the establishment and regeneration of desired vegetation such as cottonwood and willows. It grows relatively quickly and develops a dense canopy which crowds out vegetation or prevents shade-intolerant vegetation establishment, thereby reducing species diversity and plant productivity. Its growth on streambanks can also alter the natural flood regime of a waterway and reduce availability of nutrients and moisture.

Uses

Livestock sometimes browse young Russian olive trees, but once thorns develop, they are deterred. Native birds and mammals eat the fruits produced by this species. Game birds are particularly fond of Russian olive seed. Several birds feed on sprouts from new seeds as they emerge from the soil. Smaller mammals such as squirrels and pocket gophers can heavily feed on the roots and bark of younger trees causing them to die. Several mice species feed on its seed and prevent Russian olive seed germination. Bees and other pollinators will occasionally visit Russian olive flowers in low densities (Zouhar, 2005).

Status
Threatened or Endangered: Russian olive is not a threatened or endangered species.

Wetland Indicator: Russian olive is a facultative (FAC) wetland indicator species in the western mountains, valleys and coast, and the arid west regions of North America indicating that is likely to occur in wetlands and non-wetlands. Its wetland status is facultative upland (FACU) in all other regions indicating that it usually occurs in non-wetlands but occasionally grows in wetlands (USDA, NRCS, 2020).

Weedy or Invasive: Russian olive is considered an aggressive invader, especially along waterways. It is listed on 46 state noxious weed lists and, for many states, the intentional spread or sale of this species is prohibited. This plant may become weedy or invasive in some regions or habitats and may displace desirable vegetation if not properly managed. Please consult with your local NRCS Field Office, Extension office, state natural resource, or state agriculture department regarding its status and use.

Please consult the PLANTS Website (http://plants.usda.gov/) and your state's Department of Natural Resources for this species current status (e.g., threatened or endangered species, state noxious status, and wetland indicator values).

Planting Guidelines
Russian olive is an undesired, invasive species, and should not be cultivated, planted, or propagated and it is unlawful to do so in many states. In the mid- to late-1900s, Russian olive was a recommended conservation species, however research has proven it is too difficult to manage and control (USDA, NRCS, 2019). A study of emergence and seed viability at the Bridger Plant Materials Center found that Russian olive seeds planted at a depth of 3 inches or deeper do not emerge and are not viable afterwards. This finding suggests that natural environmental conditions, like a flood event, could bury seeds to depths at which they will not emerge nor be viable if uncovered later (Hybner and Espeland, 2014). Please contact your local agricultural extension specialist or county weed specialist to learn how to best manage it in your area.

Management
Russian olive management typically focuses on control. Please see the control section.

Environmental Concerns
Russian olive is an aggressive invader, capable of out competing desired species. It spreads easily through a variety of ways, but its hardiness is the reason it is difficult to control. Russian olive is tolerant to high winds, floods and drought, extreme hot and cold temperatures, and can grow on both saline and alkaline soils (USFS, 2014). There is evidence that Russian olive is one of the most salt-tolerant tree species on saline soils (Scianna, 2016). These tolerances, combined with its aggressive growth and competitiveness with native species, make it difficult to control, especially after establishment.

Seeds spread easily through several different modes and account for most new plants that emerge. Most commonly, birds and other animals such as coyotes, deer, racoons, and smaller mammals consume the fruit and excrete seed in new areas. Fruit floats and is easily dispersed along waterways. While seed is not produced until the tree is at least 4 years old, viable seed can persist in the soil for many years thereafter (USFS, 2014). In a seed longevity study, there was significant evidence that a Russian olive seed can remain viable for up to 28 years and possibly longer (Scianna et al., 2012). Although less frequent, Russian olive can also spread by vegetative sprouts, stem cuttings, and root pieces (USDA, NRCS, 2019).

A study by Lesica and Miles (2004) found that areas with greater beaver populations may support the spread of Russian olive. Beavers prefer native woody species, such as cottonwood and willow, rarely using invasive woody species in their diets. As with other invasive species, Russian olive thrives when there is less competition and no natural predators.

The southwestern willow flycatcher (*Empidonax traillii extimus)* is an endangered, native bird that uses Russian olive and saltcedar (*Tamarix ramosissima*) for nesting habitat (USFWS, 2014). The southwestern willow flycatcher will nest in native riparian areas wherever possible but are forced to use invasive species as an alternative in areas where native plants have been displaced (USFS, 2014). Cautionary measures should be used when removing these invasive plant species to ensure the endangered bird is not harmed.

Control
Cultural: Preventing establishment is the most effective and least expensive control tactic. In several western states, it is unlawful to plant Russian olive as a landscape or ornamental tree. As with other non-native invasive species, detecting new infestations early and acting quickly to eradicate or contain an infestation is advised. Targeting control on low-density sites is

less costly on a per-acre basis and helps limit future seed production while allowing the understory to return to desired species. On high-density sites it can be challenging and expensive to remove Russian olive without adverse impacts for the environment. Machinery, humans, and livestock should be checked and cleaned after travelling through infested areas in order to prevent the spread of seed. Increased awareness and education about Russian olive is beneficial to all communities in the U.S., especially those near wetlands and waterways. Regardless of control methods used, sites should be monitored for at least two years following treatment to manage new seedlings and herbaceous weeds, and to make sure desired competitive vegetation is establishing.

Russian olive infestation in a riparian area. Photo by USDA-USFS.

Mechanical: Control options include pulling, mowing, cutting, and girdling. Seedlings can be controlled by hand-pulling or frequent mowing until stems get larger than one inch in diameter. Russian olive can be cut with chainsaws, axes, shears, etc. Cutting closely to the ground will eliminate top growth for a short period but sprouts will develop from the base of the stumps. Girdling interrupts the transport of photosynthates to the root system which effectively starves the entire plant. Girdling is the complete removal of a horizontal 2 to 5-inch wide strip of bark from the entire circumference of the trunk.

Chemical: Herbicides can provide effective control as foliar and basal-bark applications and should be combined with mechanical treatments in order to manage Russian olive. Foliar applications are useful on developed trees only after there is sufficient foliage to uptake the applied herbicide. Thoroughly wet green leaves and shoots, especially near the top of the plant, while minimizing dripping. It is advised to conduct foliar spraying in the late fall to reduce the chances of injury to desirable vegetation; however, more than one foliar application may be needed each year. Basal bark applications are applied directly to the entire circumference of the lower two feet of an uncut trunk at any time of the year and are most effective on stems <5 inches in diameter. In addition, cut stumps and girdling combined with herbicide treatments will improve control, limit sprouting, and can be applied at any time of the year except freezing conditions. Thoroughly wet the cut surface or girdle wound with herbicide immediately after cutting. Use individual plant herbicide treatments (i.e., spot spraying foliage, basal bark applications, cut stump, girdling) for light infestations, areas with difficult access, or areas with desirable vegetation. Use broadcast foliar applications for dense infestations and when desired vegetation is absent. Effective herbicides for Russian olive control contain the active ingredients triclopyr (Garlon 3A, Garlon 4), 2,4-D + triclopyr (Crossbow), imazapyr (Arsenal, Habitat), or glyphosate (Roundup). Consult the label on the need to add a nonionic surfactant (USDA, NRCS, 2019; USFS, 2014).

Please contact your local Extension specialist or county weed specialist to learn what control methods work best in your area and how to use it safely. Always read label and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA NRCS does not guarantee or warranty the products and control methods named, and other products may be equally effective.

Grazing: Trained goats will selectively graze Russian olive seedlings and young trees. Grazing will be most effective when combined with other controls. There are currently no classical biological control options (USDA, NRCS, 2019).

Prescribed burning: Prescribed fire will not eliminate Russian olive but can be considered for suppression of saplings. Integration with herbicides can increase effectiveness of control. Russian olive can grow from buds that are in contact with soil, so it is important to make sure all plant remnants are destroyed by fire, shredding, or mulching. In some cases, Russian olive can come back more effectively and quickly after prescribed burning or wildland fire. It is important to create and maintain a monitoring plan because early detection is the key to managing Russian olive. This may include monitoring areas adjacent to the burn area (Zouhar, 2005).

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Helping People Help the Land

Plant Guide

SPOTTED KNAPWEED

Centaurea steobe L.

Plant Symbol = CEST8

Contributed by: USDA NRCS Montana Plant Materials Program

Figure 1. Spotted knapweed flower heads. Photo by Christina Herron, Montana State University, Bozeman, Montana. Used with permission.

Alternate Names

Common Alternate Names: None

Scientific Alternate Names: Centaurea maculosa Lam., *Centaurea biebersteinii* DC.

Taxonomy: Spotted knapweed is in the Asteraceae (sunflower) family. In its native range of Western, Central, and Eastern Europe, two sub-species have been identified; *Centaurea stoebe* spp. *stoebe* is diploid and biennial, and subspecies *C. stoebe* spp. *micranthos* is tetraploid and perennial. The perennial subspecies is considered more invasive in Europe than the biennial subspecies. The invasiveness of the North American taxon, *C. stoebe* spp. *micranthos*, has been ascribed to it being perennial because it can tolerate dense vegetation once it has become established, whereas the biennial is more dependent on disturbance.

In the Ukraine, spotted knapweed hybridized with diffuse knapweed (*Centaurea diffusa*) on sites where the two species coexisted. In North America, hybrids of the two species are only found on sites invaded by diffuse knapweed leading to the hypothesis that hybrid individuals were introduced into North America with diffuse knapweed (Blair and Hufbauer, 2009).

Uses

Bee keepers value the flowers of spotted knapweed because of the flavorful honey produced from its nectar.

Status

Spotted knapweed is listed as noxious, prohibited, banned or otherwise regulated in 16 states. Please consult the PLANTS Web site and your State Department of Natural Resources for this plant's current status (e.g., threatened or endangered species, state noxious status, and wetland indicator values).

Weediness

This plant is weedy or invasive in some regions or habitats and may displace desirable vegetation if not properly managed. Please consult with your local NRCS field office, cooperative extension service office, state natural resource, or state agriculture department regarding its status and use. Weed information is also available from the PLANTS Web site at http://plants.usda.gov/. Please consult the Related Web Sites on the Plant Profile for this species for further information.

Description

General: Rosette leaves grow from buds on the root crown of a deep taproot. They have short stalks and grow up to eight inches long and two inches wide and are deeply divided once or twice into oblong lobes on both sides of the center vein. Flower stems are eight inches to four feet tall and branch on the upper half. Stem leaves are smaller toward the stem apex, alternately arranged, sessile, and have few lobes or are linear and entire. Flower heads are solitary or in clusters of two or three on the branch ends, ovate to oblong, ¼-inch wide and ½-inch long. The involucres bracts of the flower head are imbricate, widest and yellow-green at the base, with black margins, obvious dark longitudinal veins, and a fringe of spines, the central spine shorter than the lateral ones. There are 20 to 30 purple to pink (rarely white) flowers per flower head. Seeds are ⅛-inch long, oval, brown to black, with pale longitudinal lines and a pappus of short, simple, and persistent bristles.

Life History: There are four relatively distinct and measurable life history stages of spotted knapweed; seeds, seedlings, rosettes, and flowering plants. Spotted knapweed reproduces only by seeds. Seed production of spotted knapweed in western Montana ranged from 10,760 to 83,950 per square foot on an Idaho fescue habitat-type (Jacobs and Sheley, 1998), was measured at 46,285 per square foot in Colorado (Seastedt et al., 2007), and was reported as high as 430,550 per square foot in Washington (Shirman, 1981). In Montana, viable seeds were recovered from soil that had no seed inputs for eight years (Davis et al., 1993). Seedlings are first-year emergents and are difficult to distinguish from seedlings of other forb species. Spotted knapweed can persist for an entire growing season in the seedling stage. Rosettes can develop from seedlings within one growing season, and can be identified, in most cases, by the distinctive pinnatifid, oblong-lobed leaves. In addition, plants that flower in one year may persist in the following year as rosettes. Flowering plants are distinguished by the production of an upright (except when heavily grazed or repeatedly mowed) paniculate inflorescence with few to many branches and reaching heights of eight inches to four feet depending on the environmental conditions and plant competition. Development of flowering plants from seedlings in one growing season is common. Flower heads develop on branch ends and are distinguished by the comb-like fringed, black-tipped involucre bracts and the pink to light-purple flowers. Each flower head can have as many as 30 flowers each producing one seed. Sensitivity analysis of life history stages and calculated transitions has identified early summer rosette survival, the transition from the rosette stage to the flowering plant stage, flowering plant survival, and seeds produced per flowering plant as life history stages and transitions critical to spotted knapweed population fitness (Jacobs and Sheley, 1998).

Distribution: Spotted knapweed is native to Central Europe east to Central Russia, Caucasia, and Western Siberia. It was first reported in North America in 1883 from Victoria, British Columbia and has since spread to all but eight continental provinces and states. For current distribution, please consult the Plant Profile page for this species on the PLANTS Web site.

Habitat: In its native range spotted knapweed commonly grows in the forest-grassland interface on deep, well-developed to dry soils. It forms dense stands in more moist areas on well-drained soils including gravel, and on drier sites where summer precipitation is supplemented by runoff.

Ethnobotany

Centaurea is appropriately derived from the Greek word for Centaurs, *kentaruion*, which were the mythical creatures with human heads, arms, and chests, and the rest of the body like that of a horse. The unruly Centaurs that lived in herds around Mount Pelion in Thessaly, Greece, were a plague to people around them.

Adaptation

In North America, Spotted knapweed has been reported from elevations ranging from 1,900 to over 10,000 feet, in precipitation zones ranging from 8 to 79 inches annually, and growing on a wide range of soils types.

Establishment

Spotted knapweed establishes from seed only. Seed crops have a high percent viability and will germinate in the fall of the year produced, the following spring, or will remain dormant and viable at a significant percentage for eight or more years (Davis et al., 1993).

Pests and Potential Problems

Spotted knapweed is a difficult to manage weed pest in the semi-arid west and in the mid-western United States and Canada.

Environmental Concerns

Areas with large-scale and dense infestations of spotted knapweed have increased surface water runoff and stream sedimentation and reduced soil water infiltration (Lacey et al., 1989), reduced forage production for some classes of livestock (Watson and Renney, 1974), and reduced wildlife habitat (Spoon et al., 1983).

Control

Herbicides: Short-term control of spotted knapweed populations is effective using herbicides. The length of control (i.e., the time the population regenerates from seeds in the soil) will depend on the size of the soil seed bank (effected by how long the population has been there), soil residual activity of the herbicide (effected by soil texture and precipitation), and the competitiveness of the plant community. Picloram applied at one pint product per acre (0.25 pounds active ingredient per acre) can provide 90 percent or more population reduction for three or more years on loamy soils with a well-maintained grassland community. However, picloram is a restricted-use herbicide and cannot be applied near surface water or where there is a high water table. It is water soluble, mobile, and will leach quickly from the rooting zone in sandy soils. Picloram also breaks down in sunlight which reduces its residual activity. Because of the residual activity, timing of picloram application is not as critical as with other herbicides with less residual activity. Spring, early summer, and fall applications result in the greatest control. Application during the hot and dry part of the summer should be avoided because uptake into the plant is limited when plants are dormant and the active ingredient breaks down rapidly in the sun.

An alternative to picloram is 2,4-D, a broadleaf selective herbicide, which can be applied to sensitive areas or where the use of picloram is prohibited. The timing of 2,4-D application is important for maximizing control because this herbicide has brief residual soil activity. To have the greatest reduction of spotted knapweed populations, 2,4-D should be applied after most of the seeds have germinated and before plants flower, generally at the late bud stage but before flowers appear. This timing will target early summer rosettes, prevent the transition from rosette to flowering plant, and eliminate seed production. These are life history stages and transitions that are most important to spotted knapweed population fitness. Repeated annual applications of 2,4-D may be necessary to maintain control of plants that regenerate from the soil seed bank. However, re-application will depend on the degree of seedling suppression by competitive plants. Other herbicides available for control of spotted knapweed include products that contain aminopyralid, dicamba, clopyralid, or triclopyr.

Biological control: There are eight flower head insects and five root-boring insects that have been approved and released for biological control of spotted knapweed in the United States (Story et al., 2004). Most of these insects are available commercially or through state, federal, or private programs. Once insects are established they can be collected on site and re-distributed. Bio-control insects may reduce spotted knapweed populations where competitive plants are available, but without other management, are unlikely to eradicate populations.

Urophora seed head flies were released over 20 years ago and are well established throughout most of the spotted knapweed-infested areas in the western United States. These species have been observed to reduce seed production by 50 percent or more. Other flower head feeding insects are not as widely distributed, but may be as effective as the *Urophora* fly. The *Larinus* flower-head weevils and *Metzneria* seed head moth are believed to be effective in reducing seed production. *Larinus* species prefer hot dry sites and *Metzneria* does best on sites with winter snow cover. A Montana study calculated the reduction in seed production by the combination *Urophora* and *Larinus* feeding to be 84.2 to 90.5% (Story et al., 2008). However, seed feeding insect species are not compatible with each other. On a site in Colorado, *Larinus* consumed about 40% of *Urophora* in co-infested spotted knapweed flower heads (Seastedt et al., 2007) and in Montana *Urophora* reproduction was 71 percent lower when *Larinus minutus* was present (Smith and Mayer, 2005). Poor establishment of the *Chaetorellia* and *Terellia* flies is believed to be the result of competition with other flower head insect species. The larva of *Metzneria* and *Bangasternus* will attack other insects in the seed head. The *Bangasternus* seed head weevil feeds primarily on diffuse and squarrose knapweed, but reduces spotted knapweed seed production by up to 95%.

Of the root feeding insects, the *Agapeta* root moth, *Cyphocleonus* root weevil, and *Sphenoptera* root borer are well established in parts of the western states. These species prefer hot, dry, open sites. Reductions in spotted knapweed biomass and density have been noted 10 years after the release of root-boring weevils (Jacobs et al., 2006). Observations suggest that the *Cyphocleonus* root weevil reduces the longevity of spotted knapweed plants making their duration more biennial than perennial, and thus less competitive with perennial grasses. Combining the root herbivore *Cyphocleonus* with the seed feeding weevil *Larinus* reduced spotted knapweed biomass and seed production additively compared to either insect alone in a common garden study, and the presence of plant competition further decreased knapweed growth (Knochel et al., 2010).

The *Pelochrista* root moth first released in Montana in 1984 has been slow to establish for unknown reasons. The *Pterolonche* root moth was released and established in Oregon in 1986 but has not been recovered since 2000, presumably because of the dramatic control of diffuse knapweed (another host for this species) by the seed head weevils (Story et al., 2004).

Reductions in spotted knapweed densities have been observed in southwestern Montana after the release of flower head-and root-feeding biological control insects. In most cases, two or more insect species establish on the spotted knapweed population. Ten years after the release and establishment of *Cyphocleonus achates* and *Larinus spp*. in large scale and dense populations of spotted knapweed, three different plant communities were observed depending on management treatments. Where the biological control insects were released and no other management was used, the plant community remained dominated by spotted knapweed. Where picloram was applied and the biological control insects established on spotted knapweed regenerating from the seed bank, cheatgrass (*Bromus tectorum*) was the dominant plant in the plant community. Where picloram was used and perennial grass seeded and established, and the insects established, the perennial grasses dominated the plant community. These observations illustrate how plant community composition was important in influencing the outcome of management actions.

Grazing Control: Spotted knapweed has adequate nutritional quality during the growing season to sustain livestock and wildlife based on crude protein and neutral detergent fiber concentration of harvested and dried rosettes, bolting, and flowering/seed set plants (Ganguli et al., 2010). Crude protein concentration was greater in rosettes (20%), than bolting (12%) and flowering/seed set plants (11%). Neutral detergent fiber was lowest in rosettes (30%), followed by bolting plants (29%) and highest in flowering/seed set plants (40%). In a cafeteria-type preference trial, sheep readily consumed all spotted knapweed phenological stages, but generally selected rosettes and bolting plants over flowering and seed set plants (Ganguli et al., 2010).

In a confined grazing study, five years of repeated sheep grazing reduced spotted knapweed density and biomass compared to a control and to a first year only application of 2,4-D (Sheley et al., 2004). Also in this study, repeated sheep grazing after a one time application of 2,4-D reduced spotted knapweed density and biomass compared to the one time application of 2,4-D after five years indicating confined sheep grazing can be used in an integrated pest management program to maintain spotted knapweed below a threshold achieved after pesticide application.

A prescribed grazing study herded a band of 800 ewes and 1,120 lambs on lightly infested (13% spotted knapweed vegetative composition) or moderately infested (36% spotted knapweed vegetative composition) rough fescue/bluebunch wheatgrass (*Festuca campestris/Pseudoroegneria spicata*) foothills rangeland in western Montana in mid-June or mid-July (Thrift et al., 2008). Sheep diets averaged 64% and 26% spotted knapweed in the moderate and light infestations, respectively. Fewer graminoids were eaten in June than July in the light infestation whereas fewer graminoids were eaten July than June in the moderate infestation. The authors concluded this prescription for sheep grazing on these sites would make herbicide application uneconomical suggesting prescribed sheep grazing can be used as an alternative to pesticide application in an integrated pest management program for spotted knapweed.

Prescribed grazing to maintain the vigor and competitiveness of grassland plant communities will prevent spotted knapweed invasion. On the other hand, intense and frequent grazing pressure opens grassland plant communities to spotted knapweed invasion and re-invasion after management treatments (Jacobs et al., 2000; Jacobs and Sheley, 1999; Jacobs and Sheley, 1997).

Mowing: A hand-clipping study in west-central Montana removed spotted knapweed buds and flowers at seven different timings and frequencies (Benzel et al., 2009). Clipping when plants were bolting to flower, or later in the season, reduced spotted knapweed seed production by 90% and 100%, respectively, compared to a no clipping control. The results suggest defoliation by mowing or prescribe grazing will suppress spotted knapweed viable seed production. The results of a mowing study led to the recommendation of a single annual mowing applied at the flowering or seed producing stage for partial control of spotted knapweed (Rinella et al., 2001). However, mowing at the flowering stage or later may cause mortality of biological control insect larvae where they are established on spotted knapweed (Story et al., 2010).

Prescribed Burning: A study in western Michigan applied prescribed burning to spotted knapweed-infested gravel mine spoils in late April or May for three years reducing spotted knapweed density and biomass and increasing the dominance of warm-season grasses (MacDonald et al., 2007). The results support using carefully timed burns to optimize the reduction of low-density spotted knapweed populations while benefiting fire-adapted plant communities with abundant warm-season grasses. Prescribed burns on sites in western Montana in cool season grass communities resulted in increases in invasive species biomass and seed production (Jacobs and Sheley, 2003).

Hand Pulling: Hand pulling that extracts the root crown can temporally reduce spotted knapweed on small-scale infestations or as a follow-up treatment to initial herbicide treatment on larger-scale infestations. Pulling or grubbing the root crown is most easily accomplished when the soil is moist and a shovel is used to pry-up the tap root. When the soil is dry the plant tends to break-off above the root crown enabling it to regenerate. If flowering plants have been pulled, they should be sealed in plastic bags and disposed of in the trash to prevent seed spread. Wearing gloves while pulling spotted knapweed will protect against potential skin irritation from chemicals produced by knapweed.

Please contact your local agricultural extension specialist or county weed specialist to learn what works best in your area and how to use it safely. Always read label and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA NRCS does not guarantee or warranty the products and control methods named, and other products may be equally effective.

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USDA NRCS
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Natural Resources Conservation Service

Plant Guide

RUSH SKELETONWEED

Chondrilla juncea L.

Plant Symbol = CHJU

Contributed by: USDA NRCS Montana State Office

Rush skeletonweed rosette and flowering stem. Photo by Tim Prather, University of Idaho

Caution: This plant may be weedy or invasive.

Alternate Names
skeletonweed, naked weed, gum succory**.**

Uses
Rush skeletonweed is palatable and nutritious in the rosette and early bolting stages and makes good sheep and goat fodder.

Honey bees use it for pollen and honey.

Status
Rush skeletonweed is a non-native, invasive terrestrial forb listed as noxious, prohibited, or banned in nine western states.

Weediness
This plant may be weedy or invasive in some regions or habitats and may displace desirable vegetation if not properly managed. Consult with your local NRCS Field Office, Cooperative Extension Service office, state natural resource, or state agriculture department regarding its status and use. Additional weed information is available from the PLANTS Web site at plants.usda.gov. Consult other related web sites on the Plant Profile for this species for further information.

Description
Rush skeletonweed forms a rosette of prostrate, glabrous leaves 1.6 to 24.7 inches (4-12 centimeters) long, 0.6 to 1.8 inches (1.5 to 4.5 centimeters) wide, and oblanceolate in shape. The leaf margins are deeply and irregularly toothed with lobes pointing backward toward the leaf base (runcinate) similar to the rosette leaves of dandelion (*Taraxacum* spp). The leaf base narrows to a short, winged petiole. Normally, one flowering stem grows per rosette. Flowering stems reach heights of 1.6 to 3.3 feet (50-100 centimeters) and have numerous spreading or ascending branches. They are glabrous except for short, rigid, downward-pointing hairs near the base, similar to prickly lettuce (*Lactuca sirriola*). Generally, the stems are leafless, they may have long-linear, bract-like leaves, or they may have leaves similar to the rosette leaves but smaller and only on the lower part of the stem. The rosette leaves die at flowering leaving a skeleton-like stem.

The flowerheads (capitula) are solitary or in groups of two to five in the stem branch axils, along the branches, and at the branch ends. The cylindrical involucre has two rows of bracts; the outer row is very short and crown-like, the inner row has seven to nine linear-lanceolate bracts with either no hairs, sparsely tomentose, or sometimes a row of rigid hairs on the median line. Each capitulum bears nine to 12 bright yellow, ligulate florets. The florets produce achenes (small fruits) three to four millimeters long and with numerous ribs. At the tip of the achene is a beak five to six millimeters long that bears a pappus of numerous soft bristles. The pappus facilitates wind dispersal.

The taproot of rush skeletonweed is small in diameter but penetrates deeply into the soil. Lateral roots are produced along its entire length. Rosettes can grow from adventitious buds at the top of the tap root and along major lateral roots. The roots are brittle and easily break during cultivation or other soil disturbance. Thick white latex exudes from the leaves, stems and roots when they are broken or cut.

Distribution
For current distribution, consult the Plant Profile page for this species on the PLANTS Web site.

Habitat
Rush skeletonweed originated from the Transcaspian region of Eurasia, its native range extending from Western Europe and northern Africa to central Asia. It has become widespread in wheat growing regions and rangelands of Idaho, Oregon, and Washington. It is considered an early seral species invading disturbed areas in crop, pasture, range and forest lands. It may also invade intact plant communities in low rainfall areas such as southwestern Idaho. Optimum climatic conditions for rush skeletonweed are cool winters and warm summers without severe drought and with winter and spring precipitation typical of semi-arid and Mediterranean climates. It has been found in areas with annual precipitation ranging from 9 to 59 inches (23 to 150 centimeters) and elevation ranging from sea level to 6,000 feet. Summer temperatures of at least 59 degrees Fahrenheit (15 degrees Celsius) are needed for flower and seed production. Rush skeletonweed has no absolute requirement for vernalization although it accelerates flowering. It is found on a wide range of soil types but is most abundant on sandy, sandy-loam, and silt loam soils. It is a weed of cultivated sites, open areas and disturbances. Areas affected by wildfire and pastures weakened by drought, overgrazing, or with cheatgrass or medusahead invasion are susceptible to rush skeletonweed invasion.

Life History
In its native Eurasian range, rush skeletonweed is described as a biennial. In its invaded ranges in Australia and North and South America it is described as a perennial living up to 20 years. There are also variations in its form. The root system is long lived and rich in carbohydrate reserves. Adventitious buds on the roots enable it to grow year after year. New plants can arise from intact roots or root fragments and local population expansion is mainly by vegetative regeneration. One to several rosettes grows from adventitious root buds of the parent plant usually in autumn (September into November). Plants overwinter as rosettes and begin growth again in the spring (March and April). Rosettes can begin growth in summer if moisture follows drought and rosettes that initiate growth in summer usually flower immediately. Flowering stems elongate from the central growing points of rosettes in April and May. As the flowering stems grow the rosette leaves die leaving nearly leafless plants during the summer. Flower buds form in June and July, and plants bloom in July. The capitula open early in the morning and close before sunset. In hot dry conditions, the capitula will only remain open for a couple of hours. Seeds form without pollination (apomixis). Seeds are fully developed about two weeks after flowering and a small number of seeds have been observed to germinate three days after flowering. Seed production peaks in July and August but can continue into November. Seed production per plant under field conditions can be as high as 10,000, and seed production from dense populations were estimated to be 70,000 per square meter. Flowering stems usually die in October at about the time new rosettes begin to appear; however the timing is variable depending on moisture conditions.

Rush skeletonweed flower. Photo by Tim Prather, University of Idaho.

Rush skeletonweed seed is not long-lived in the seedbank because they have little to no dormancy and they only remain viable in the soil for 6 to 18 months. However, an Idaho study found that 60% of seed stored for one year maintained good viability. The seed has high viability (up to 80%). Burial of seed deeper than 25 millimeters in the soil prevents germination. Seedlings may emerge at any time moisture is available and temperatures are above 45 degrees Fahrenheit (7 degrees Celsius), but most germination occurs in the fall. As little as 5 millimeters of rain will stimulate germination. Seedlings require a continuous supply of moisture for three to six weeks to survive desiccation. Above-ground growth of seedlings is slow in the fall, but seedling roots grow rapidly. Plants overwinter as rosettes. Rosettes developed from autumn-emerged seedlings usually produce a flowering stem the spring following emergence.

Establishment
Rush skeletonweed establishes from seed and adventitious buds on roots.

Management
See Control.

Pests and Potential Problems
See Environmental Concerns.

Environmental Concerns
In Australia, rush skeletonweed is considered the most serious weed in wheat growing regions where it reduces

yields and the wiry flowering stems, or their latex, clog harvesting equipment which increases breakdown and maintenance costs. Infestations reduce grazing forage potential, the stems interfere with livestock grazing, and there have been reports of the stems causing choking when eaten by cattle. Dense infestations reduce native plant diversity.

Seeds and Plant Production
Not applicable.

Control
Contact your local agricultural extension specialist or county weed specialist to learn what works best for control in your area and how to use it safely. Always read label and safety instructions for each control method. Trade names and control measures appear in this document only to provide specific information. USDA NRCS does not guarantee or warranty the products and control methods named, and other products may be equally effective.

Rush skeletonweed's ability to regenerate from roots deep in the soil profile along with poor translocation of herbicide to the extensive root system makes this weed difficult to control with herbicides. Successful control of this plant using herbicides usually requires multiple reapplications. The poor soil conditions favored by the plant (e.g., dry, coarse, and low in organic matter) also reduce herbicide persistence in the soil. Additionally, the morphology of rush skeletonweed, specifically the lack of leaf area, reduces herbicide translocation as a result of inadequate retention and adsorption. Translocation can be improved with silicone surfactants and water conditioning agents. Picloram (one quart product per acre) or picloram combined with 2,4-D (one quart plus one quart per acre) applied to autumn rosettes are the herbicide treatments that give the best root killing results. A single application is not likely to kill all root buds and applications in subsequent years will be necessary. Clopyralid, aminopyralid, and dicamba also translocate into the roots. Hand pulling and digging can provide control of small populations if plants are pulled several times each year for many years. Hand pulling will stimulate adventitious growth from root buds for the first few years until root reserves are depleted. Six to 10 years of mechanical control will be needed to eliminate populations.

Mowing is not an effective control for rush skeletonweed. Rosettes are flat to the ground and missed by the mower blade. Mowing when plants bolt to flower may temporarily reduce seed production but plants will survive to flower again.

Root fragments of rush skeletonweed are spread by tillage which may increase infestation size. Tillage every six to eight weeks may effectively eliminate the weed. In many locations in Idaho, Montana, Oregon, and Washington, rush skeletonweed does not occupy sites where tillage is practical.

Irrigation is not recommended as a control by itself because it stimulates seedling and rosette emergence. Where rush skeletonweed invades irrigated pasture and hayland, carefully planned irrigation management will stimulate the competitiveness of the forage crop and when combined with nutrient, forage harvest, and grazing management practices will help prevent the re-establishment of rush skeletonweed after other control practices are applied.

Rush skeletonweed produces larger and leafier rosettes, but not more rosettes, when nitrogen fertilizer is applied. One study found application of superphosphate (about 125 pounds per acre) reduced rosette densities by an average of 80%, probably due to increased competition from pasture plant species. Rush skeletonweed survival relies on a lack of competition, which is of greater importance than increased nutrient levels. That said, nutrient management of hay lands and pastures will stimulate desired plant vigor and reduce the risk of invasion by rush skeletonweed.

A study in Idaho shrub-steppe communities found a nearly six-fold increase in rush skeletonweed rosette emergence where wildfires burned compared to non-burned sites the autumn following the burn. Insulated by the soil, rush skeletonweed roots are protected from killing heat of fire. There was also greater seed germination on fire-affected soil compared to unaffected soil. The disturbance of fire produces conditions favorable to rush skeletonweed invasion and population expansion. Prescribed burning should not be conducted in or near areas where rush skeletonweed has invaded unless follow-up management is applied.

Rush skeletonweed is good forage for sheep and goats because it is palatable and nutritious in the rosette and early bolting stages. Continuous grazing in the spring and summer will keep it in the rosette stage, but it will quickly flower if grazing is discontinued. Continuous grazing of larger populations is a good strategy to prevent flowering and seed production and thus restrict spread to distant sites along wind currents. Many populations throughout the Intermountain West are small and therefore prescribed grazing as a control may not be practical. However, prescribed grazing is recommended as a preventative management by maintaining a competitive pasture or rangeland plant community.

Three biological control agents have been released to manage rush skeletonweed but they have been successful only in certain locations. The skeletonweed root moth, *Bradyrrhoa gilveolella*, was introduced in Idaho in 2002 but establishment has not been confirmed by 2009. The rush skeletonweed gall midge, *Cystiphora schmidti*, was first released in California in 1975 and is available for mass collection in California, Idaho, and Oregon. It damages rosettes and flowering stems reducing seed production. The rush skeletonweed rust fungus, *Puccinia chondrillina*, is the first exotic plant pathogen to succeed as a classical biological control agent in North America by reducing rush skeletonweed to "tolerable levels." It is readily available for redistribution in California, Idaho, Oregon, and Washington. The effectiveness of biocontrol agents vary depending on local conditions and plant genotype. The rust appears more effective in California and the mite appears to be more important in eastern Washington.

Rush skeletonweed is not tolerant of shade and is seldom found on closed forest canopy sites. Disturbance is favorable to rush skeletonweed and removal of natural vegetation provides opportunities for establishment. Revegetation of disturbances is therefore an important measure to provide competition and hinder rush skeletonweed invasion. The use of legumes in crop-pasture rotations has been effective in reducing populations of the weed. The deeply-rooted alfalfa is advantageous because it is competitive for deep soil moisture. Alfalfa also increases soil fertility and plant competition to reduce rush skeletonweed populations.

Cultivars, Improved, and Selected Materials (and area of origin)
Not applicable.

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