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TWENTY NON-NATIVE INVASIVE PLANTS ARMY INSTALLATION LAND MANAGERS SHOULD KNOW ABOUT

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TWENTY NON-NATIVE INVASIVE PLANTS ARMY INSTALLATION LAND MANAGERS SHOULD KNOW ABOUT

1. Purpose.

a. This Public Works Technical Bulletin (PWTB) provides an overview of 20 invasive weed species that occur on Army installations in the continental United States (CONUS). This document presents information for each species as a fact sheet that covers species biology, control/management, and impacts on the Army mission.

b. All PWTBs are available electronically at the National Institute of Building Sciences' Whole Building Design Guide website at:

http://www.wbdg.org/ccb/browse_cat.php?o=31&c=215

2. Applicability. This PWTB applies to all U.S. Army facilities engineering activities within CONUS.

3. References.

a. Army Regulation (AR) 200-1, Environmental Protection and Enhancement, 21 February 1997.

b. AR 350-4, Integrated Training Area Management, 8 May 1998.

c. Executive Order (EO) 13112, Invasive Species, 3 February 1999.

4. Discussion.

a. AR 200-1 states that the Army will plan and conduct peacetime mission activities to minimize adverse impacts on the environment. Furthermore, AR 350-4 provides for the repair and rehabilitation of training lands, including protection of natural resources, compliance with statutory regulations, prevention of future pollution, and a reduction of hazardous waste and toxic releases. To prevent introductions and spread of invasive species, EO 13112 requires federal agencies to provide for restoration of native species. Given these regulatory requirements regarding environmental stewardship on military lands, balancing competing regulatory requirements can prove difficult.

b. Invasive plant species are a primary factor in the loss of species habitat and the listing of threatened and endangered species (TES). Invasive plants have documented impacts on increased soil erosion, and increased maintenance and management costs of training facilities. Invasive plants also directly contribute to Soldier safety risks and increased security risks. All of these factors directly reduce training opportunities and mission readiness.

c. Appendix A contains background and methods information.

d. Appendix B describes the component/sections of each multi-page fact sheet for the 20 species covered in this PWTB.

e. Appendix C opens with a species listing of the 20 non-native weed species. Each species is hot-linked to the first page of its fact sheet. Each fact sheet in Appendix C covers species biology, control/management, and impacts on the Army mission.

f. Appendix D lists acronyms used in this PWTB.

5. Points of Contact (POCs).

a. Headquarters, U.S. Army Corps of Engineers (HQUSACE) is the proponent for this document. The POC at HQUSACE is Mr. Malcolm E. McLeod, CECW-CEP, 202-761-5696, or e-mail: Malcolm.E.Mcleod@usace.army.mil.

b. Questions and/or comments regarding this subject should be directed to the technical POC:

U.S. Army Engineer Research and Development Center (ERDC)
Construction Engineering Research Laboratory (CERL)
ATTN: CEERD-CN-X (Patrick Guertin)
2902 Newmark Drive
Champaign, IL 61822-1076
Tel. (217) 373-5857
FAX: (217) 373-7266
e-mail: Patrick.J.Guertin@usace.army.mil

Note: Document content was developed by Patrick Guertin and Rose Keane (ERDC-CERL) and LeeAnn Glomski (ERDC Environmental Laboratory).

FOR THE COMMANDER:

JAMES C. DALTON, P.E., SES
Chief, Engineering and
Construction Division
Directorate of Civil Works

APPENDIX A

BACKGROUND

Invasive plant species are a primary factor in the loss of species habitat and the listing of threatened and endangered species (TES). Invasive plants have documented impacts on increased soil erosion, and increased maintenance and management costs of training facilities. Invasive plants also directly contribute to Soldier safety risks and increased security risks. All of these factors directly reduce training opportunities and mission readiness.

Although many sources of invasive plant information exist, available information is tailored to agricultural or non-military natural resources interests. These “civilian”-focused sources do not account for mission-specific factors.

This document contains invasive species fact sheets to assist military land managers in identifying invasive weeds and in taking the initial steps of assessment and control. Information included in each fact sheet includes species identification, and biology and control/management information found in non-military sources; additionally, information on potential impacts to military mission and species spread is provided.

Twenty non-native species that occur across the United States are presented. These species were selected based on previous work that identifies problematic vegetation. Those previous works include a report to Congress on training range vegetation encroachment (Army 2007), a DoD-funded study completed by the National Wildlife Federation (Westbrook and Ramos 2005), and research databases developed as part of U.S. Army Engineer Research and Development Center (ERDC) studies on invasive plants (Denight and Busby 2007; Guertin and Tess 2006).

Sources:

Denight, M.L., and R. Busby. 2007. U.S. Army Installation Floristic Inventory Database. Public Works Technical Bulletin 200-1-52 (November). Washington, DC: U.S. Army Corps of Engineers.

Guertin, P.J., and S. Tess. 2006. Invasive Species and TES Interactions on Army Lands. ERDC/CERL Technical Report TR-06-12. Champaign, IL: U.S. Army Engineer Research and Development Center.

Office of the Assistant Secretary of the Army. 2007. “Report on Army Training Range Vegetation Encroachment.” U.S. Department of Army Report to Congress, U.S. Senate Armed Services Committee, January 2007.

Westbrook, C., and K. Ramos, with M. La. 2005. Under Siege: Invasive Species on Military Bases. http://www.nwf.org/nwfwebadmin/binaryVault/UnderSiegeInvasiveSpeicesReport1.pdf.

APPENDIX B

FACT SHEET FORMAT

The multi-page fact sheets for the 20 species (listed at the beginning of Appendix C) comprise the following sections:

1. Species Name and Description: Species common and scientific name along with a brief description of the species and photographs of general plant appearance and flowering structures.

2. Reproductive biology: Synopsis of plant’s reproductive strategies; both sexual and asexual (if applicable).

3. Origin and Distribution: Brief history of the species’ introduction and information on current distribution in the United States.

4. Probability of Future Expansion: Available information on species’ local and geographic spread.

5. Control Technologies: Synopsis of control technologies available, including chemical, mechanical, and biological.

6. Direct and Indirect Military Impacts: Description of potential impacts the weed species may have on Army lands. Direct impacts are those that impinge directly upon the mission (e.g., rapid plant growth rates that interfere with laser targeting devices). Indirect impacts are those that adversely impact non-military factors causing impact to the mission (e.g., weed species that threat critical wildlife habitat).

7. Installations: A listing of Army installations where the species of interest may occur. These data were collected by comparing installation location with plant occurrence in the U.S. Department of Agriculture (USDA) Weeds Database. Installations considered were Tier 1 and Tier 2. Entries with “*” had the focus weed occurring in an adjacent county.

8. Sources: Citations for all documents referenced in the fact sheet.

9. Photo credits: The source for each photograph in the fact sheet is provided.

NOTE: Units of measure used in the fact sheets are mixed between the international system (SI) and inch-pound system of units, depending on the source being cited. The following conversion chart may be used if needed.

Multiply	**By**	**To Obtain**
acres	4,046.873	square meters
degrees Fahrenheit	(F-32)/1.8	degrees Celsius
feet	0.3048	meters
hectares	1.0 E+04	square meters
inches	0.0254	meters
miles	1,609.347	meters
square feet	0.09290304	square meters
square inches	6.4516 E-04	square meters
square yards	0.8361274	square meters
yards	0.9144	meters

APPENDIX C

INVASIVE WEED FACT SHEETS

This appendix contains the fact sheets for the following invasive plant species. Page numbers at right are hot-linked to the first page of each fact sheet.

Cheatgrass (*Bromus tectorum*): Cheatgrass is a cool-season annual grass native to Eurasia. It forms tufts up to 2-ft tall, with many finely haired, drooping spikelets. The leaves and sheaths are covered in fine hair, and the flowers appear as open terminal clusters with a purple, green, or red hue, ranging from 1.5- to 8-in. long (Gasch and Bingham 2006). A cheatgrass community typically displays low species diversity but unusually high amounts of phenotypic plasticity, which enables it to respond more readily than native grasses to environmental changes (Bradford and Lauenroth 2006). Cheatgrass typically invades open areas such as rangelands, pastures, and prairies, can grow in a variety of soil types, and has been reported to be stimulated by wildfires and high levels of disturbance (Pierson 1990; Bradford and Lauenroth 2006).

Figure 1. Cheatgrass flowering head.

Figure 2. Cheatgrass monoculture.

<u>Reproductive Biology</u>: Cheatgrass is a self-fertilizing winter annual that germinates in the fall (Ramakrishnan 2006). The plant grows throughout the winter as temperatures permit, over-wintering in a semi-dormant state and continuing growth in the spring (Bradford and Lauenroth 2006; Gasch and Bingham 2006). The extensive root system of cheatgrass is then well established in the spring, giving the plant an advantage over native species. The plant resumes active growth in late April to early May, with seeds ripening in June and July and seed dispersal occurring upon maturity. Seeds need an additional after-ripening period and begin to germinate in autumn (Beckstead and Augspurger 2004). A single plant can produce hundreds of seeds that remain viable for up to 5 years (Gasch and Bingham 2006). There is a strong relationship between cheatgrass biomass and

frequency of site disturbance. Additionally, there is a correlation between cheatgrass phenology and water availability, indicating the extensive phenotypic plasticity of the weed (Bradford and Lauenroth 2006).

Origin and Distribution: The native range includes most of Europe, the northern parts of Africa, and southwest Asia. Cheatgrass was introduced to the United States as a contaminant of grain seed, but it was also occasionally introduced as forage grass (Novak and Mack 2001). It was first reported in the United States in 1790, in Lancaster County (Novak and Mack 2001). Invasion was facilitated by heavy grazing and plowing, and cheatgrass is now dominant on over 20% of the sagebrush steppe in the Great Basin (Bradford and Lauenroth 2006). Overall, it has come to occupy more than 40 million hectares in the continental United States (Beckstead and Augspurger 2004).

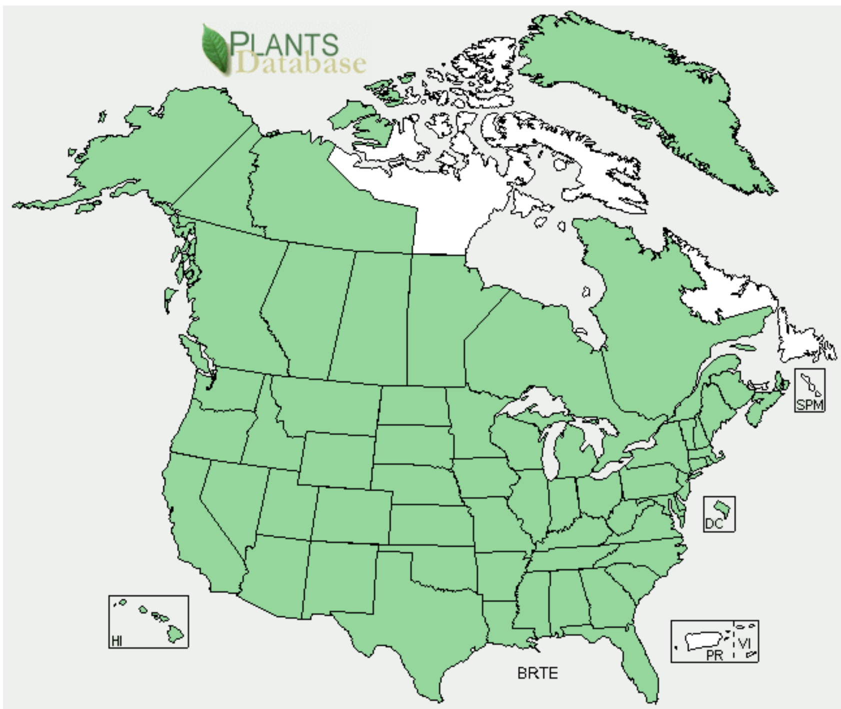

Figure 3. Distribution of Cheatgrass.

Probability of Future Expansion: Cheatgrass is a successful invasive species because of germination and dispersal patterns and its phenotypic plasticity (Gasch and Bingham 2006). In the spring, the root system of cheatgrass has already been established, giving the plant an advantage over the native grasses. Additionally, cheatgrass disperses its seeds earlier than the native grasses and therefore increases the amount of fine dry fuels (Getz and Baker 2007). Cheatgrass has been implicated in altering fire regimes, and the biomass and seed production of cheatgrass increases 10 to 30 times after native plant removal and burning (JFSP 2008). Density of cheatgrass has been reported to more than double after reduced compaction treatment (such as aeration) and removal of native competitors (Beckstead and Augspurger 2004). Climate change and carbon dioxide enrichment globally can also affect the spread of cheatgrass, as it has been reported to have the highest positive response to atmospheric carbon dioxide when compared with native plants (Billings 1990).

Control Technologies: Cheatgrass can be controlled most effectively with herbicides. In general, herbicides that should be applied in the spring include quizalofop (Assure), fluazifop-p-butyl (Fusilade), sethoxydim (Poast), paraquat (Gramaxone), glyphosate (Roundup), imazapic (Plateau), bromacil (Hyvar), tebuthiuron (Spike) and imazamox (Raptor). Quizalofop, fluazifop-p-butyl and sethoxydim should be applied before the boot stage and will not damage broadleaves (Carpenter and Murray 1999). Paraquat and glyphosate should be applied up until the plant has three to five tillers (Blackshaw 1991) and both herbicides are non-selective. Imazapic requires adequate soil moisture for optimum activity (Carpenter and Murray 1999). Bromacil should be applied before or during active growth and rain is needed for soil activation (Skinner et al. 2008). Tebuthiuron should be applied before cheatgrass begins to grow and 1 in. of rain is required to activate it (Skinner et al. 2008). Stougaard et al. (2004) reported excellent control of cheatgrass when imazamox was applied in the spring as well as consistent suppression when applied in the fall. Herbicides that should be applied in the fall include sulfometuron-methyl (Oust) and metribuzin (Sencor) (Carpenter and Murray 1999; Whitson et al. 1997). Sulfometuron-methyl can damage non-target plants if not applied correctly; therefore, care should be taken when applying this herbicide (Carpenter and Murray 1999). Metribuzin has been reported to provide 95 percent control of cheatgrass (Whitson et al. 1997). When using any of the above listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Cutting, mowing, and grazing are not effective control methods for cheatgrass; however, burning has been shown to be effective (Carpenter and Murray 1999). Although both summer and fall burns are effective, fall burns are easier to control (Wendtland 1993). Cheatgrass burns rapidly, making fires very dangerous; only trained individuals should conduct prescribed burns (Carpenter and Murray 1999). There are currently no biological control agents that have been released to control cheatgrass. Cheatgrass is susceptible to the fungal pathogens *Fusarium culmorum*, which causes crown rot (Grey et al. 1995) and to the rhizobacterium *Pseudomonas fluorescens* strain D7 (Kennedy et al. 2001). Additional research is needed to develop either one as a biological control agent.

Direct and Indirect Military Impacts: Cheatgrass has been associated with a decrease in native species and biodiversity. Additionally, the species alters soil properties and increases risks for wildfires. (Boxell and Drohan 2009; Norton et al. 2008). The change in fire regimes caused by cheatgrass also creates large and more frequent fires. The plant's ability to increase fuel loading presents a hazard to range and training lands. Additionally, its ability to alter native biodiversity can impact sensitive habitats.

Installations (25): Atterbury Reserve Training Area, Fort A.P. Hill, Fort Benning*, Fort Bliss, Fort Bragg*, Fort Campbell*, Fort Carson, Fort Chaffee*, Fort Drum*, Fort Hood, Fort Hunter Liggett, Fort Indiantown Gap, Fort Irwin, Fort Jackson*, Fort Knox, Fort Leonard Wood*, Fort Lewis, Fort McCoy*, Fort Pickett, Fort Polk, Fort Riley, Fort Rucker*, Fort Sam Houston*, Fort Sill, Fort Stewart*.

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Getz, H.L., and W.L. Baker. 2007. Initial invasion of cheatgrass (*Bromus tectorum*) into burned pinon-juniper woodlands in Western Colorado. *American Midland Naturalist* 159:489-497.

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Ramakrishnan, A.P., S.E. Meyer, D.J. Fairbanks, and C.E. Coleman. 2006. Ecological significance of microsatellite variation in western North American populations of *Bromus tectorum*. *Plant Species Biology* 21:61-73.

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Photo credits:

Figure 1: Steve Dewey, Utah State University (Bugwood.org)

Figure 2: Chris Evans, River to River Cooperative Weed Management Areas (CWMA) (Bugwood.org)

Figure 3: USDA Plants Database (plants.usda.gov)

Cogongrass (*Imperata cylindrica*): Cogongrass is a perennial colony-forming grass that can grow up to 6-ft tall. It grows in tufts that can be loose or tightly packed, with the culms growing from below ground rhizomes. Stems are present only when the grass is flowering. Leaves are slender and can be up to 6-ft long with an off center white mid-rib (MacDonald 2004). The majority of the plant's biomass is below ground and comprised of rhizomes that are tough, white, and covered in a brown protective layer. They are responsible for the grass' ability to recover quickly after cutting or herbivory. Cogongrass is a hardy species that can tolerate shade, high salinity, and drought. The plant is fire tolerant and, in areas of the country where winter climates may kill its above-ground biomass, it can add significantly to fire hazards (MacDonald et al. 2006).

Figure 4. Cogongrass mature seedhead.

Figure 5. Cogongrass.

<u>Reproductive Biology</u>: Cogongrass can spread by windborne seeds and underground rhizomes. The rhizomes can form a dense mat in the upper 6-8 in. of soil and may comprise as much as 80% of the total plant mass (Bryson and Carter 1993; Colvin et al. 1994). It is the rhizome spread that makes this plant particularly hard to control. The middles of the rhizomes contain mechanisms to prevent water loss and to resist breakage, and are very resistant to fire. Rhizomes have the potential to penetrate up to 1.2m into the soil, and seed production of cogongrass typically tops 3,000 seeds per plant (Bryson and Carter 1993).

Seeds are generally dispersed fairly close to the parent plant, but some seed may be dispersed as far as 24 miles away over open country (Daneshgar et al. 2008). Flowering in this species has been reported from March to May, in the fall after a frost, or even all year in warmer climates such as in Florida (Bryson and Carter 1993). In Alabama and Mississippi, 98% germination was reported within 1 week of harvest, with seed viability lasting at least 1 year (MacDonald 2004).

Origins and Distribution: Cogongrass was introduced to the United States many times, both accidentally and intentionally. The earliest record of the plant is from Mobile, Alabama in 1912, when cogongrass was introduced as packing material in boxes from Japan. It has also been introduced intentionally as forage, erosion control, and as an ornamental crop (Brewer 2008; Dozier et al. 1998). The species can be found throughout Mississippi and Florida and in scattered infestations in Georgia, Louisiana, South Carolina, Texas, and Virginia. Currently the species is confined to the southeastern United States. It can be found growing along roadsides, in open fields, in forests, and up to the edge of standing water (MacDonald 2004).

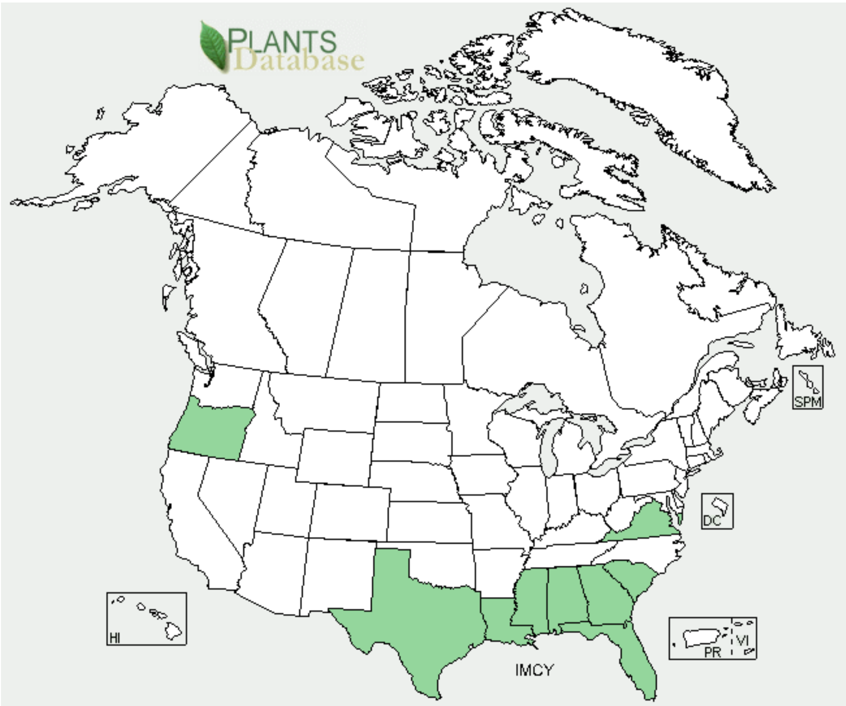

Figure 6. Distribution of Cogongrass.

Probability of Future Expansion: There is major concern in the southeastern United States, as the species has exhibited significant expansion throughout the region. Vectors include movement by equipment and contaminated soil, by wind-dispersed seeds, and by expanding colonies. Additionally, some varieties are still sold as ornamentals. Cogongrass has many competitive advantages that aid its spread. Allelopathic substances have been found in cogongrass, and the extensive rhizome and root system has been reported to inflict physical injury (resulting in infection and disease) in neighboring competing plants (Brewer 2008; Bryson and Carter 1993; Daneshgar et al. 2008). Fire conditions in cogongrass habitats reach much higher heights and temperatures, killing other plants and juvenile trees (Daneshgar et al. 2008). Plant hardiness may restrict northern expansion. Military disturbance has been shown to increase populations at a local scale (Yager et al. 2009).

Control Technologies: To successfully manage cogongrass, the rhizomes must be destroyed. To date, the most effective method for controlling cogongrass is with herbicides. Glyphosate (Roundup) and imazapyr (Arsenal) have shown the greatest efficacy and can be applied in the fall or in the spring followed by a second application in the fall. A spring application of glyphosate can provide up to 90% control through the summer months; however, control drops below 40% by the following spring. Spring applications of imazapyr can provide 80% to 90% control through to the following spring. Due to the soil activity of imazapyr, treated areas will be free from vegetation for 6 months to 1 year after treatment (Byrd 2007; MacDonald et al. 2006; MacDonald 2004). Multiple applications of either glyphosate or imazapyr are required to control regrowth from rhizomes (Dozier et al. 1998; Willard et al. 1996). Refer to herbicide labels for information regarding rates, adjuvants, application techniques, and use restrictions.

Currently, there are no biological agents used to control cogongrass. However, two fungal pathogens, *Bipolaris sacchari* and *Drechslera gigantea*, can cause foliar blight. Although further development of these two fungi is continuing, neither pathogen is host-specific and will not provide complete control of cogongrass (Yandoc et al. 2004, 2005). Mechanical techniques such as mowing and discing have been shown to reduce rhizome biomass, but multiple treatments are necessary to control regrowth (Willard et al. 1996). Mowing and discing can effectively be used as part of an integrated management plan. Discing followed by an imazapyr or glyphosate application can provide over 90% control (Dozier et al. 1998). Cogongrass may also be burned or mowed to remove older leaves and thatch, followed by herbicide application to actively growing younger leaves. This strategy can reduce rhizome biomass by initiating new growth, which also maximizes herbicide absorption to actively growing leaves (MacDonald et al. 2006). After any management strategy, infested areas should be planted with native species to prevent the reinfestation of cogongrass (Dozier et al. 1998).

Direct and Indirect Military Impacts: Cogongrass alters TES habitats; the resultant compliance issues may cause loss of valuable training lands if left uncontrolled. The plant interferes with Gopher tortoise habitat at Camp Shelby, MS (Yager et al. 2009). Invasion of the species into pine wire grass ecosystems favored by red cockaded wood peckers may be a future concern. Additional ecological impacts will presumably be caused as cogongrass causes significant losses in habitat-specialist plants (MacDonald 2004; Brewer 2008). Additionally, the plant presents a major fire hazard (Koger et al. 2004; Daneshgar et al. 2008; MacDonald 2004), which may directly impact training exercises.

Installations (9): Fort Hood*, Fort Sam Houston*, Fort Polk*, Fort Rucker*, Fort Gordon*, Fort Benning*, Fort Stewart*, Fort Jackson*, Fort Pickett*.

Sources:

Brewer, S. 2008. Declines in plant species richness and endemic plant species in longleaf pine savannas invaded by *Imperata cylindrica*. *Biological Invasions* 10:1257-1264.

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Daneshgar, P., S. Jose, A. Collins, and C. Ramsey. 2008. Cogongrass (*Imperata cylindrica*), an alien invasive grass, reduces survival and productivity of an establishing pine forest. *Forest Science* 54(6):579-587.

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Photo credits:

Figures 4, 5: Chris Evans, River to River CWMA, Bugwood.org

Figure 6: USDA Plants Database, plants.usda.gov

Diffuse knapweed *(Centaurea diffusa)*: Diffuse knapweed is an 8- to 40-in.-tall biennial or short-lived perennial species with a long tap root and many spiny seed heads. Growth form is a single stem with multiple branches; each branch is capable of supporting multiple flower clusters. Stems can grow from 50-80 cm tall, with many spreading branches and alternate leaves that can grow up to 20-cm long and 5-cm wide (Watson and Renney 1974). Flowers are generally white, but are occasionally purple or pink.

Figure 7. Diffuse Knapweed flowering head.

Figure 8. Diffuse Knapweed plant

<u>Reproductive Biology</u>: Diffuse knapweed can reproduce from its root-crown; however, its primary reproduction method is seed. Approximately 95% of seed germination occurs in April due to moderate temperatures and moist ground, with the remaining 5% occurring around September (Powell 1990). Seedlings develop into rosettes and bolt in early May after overwintering. Flowering occurs in July and August. Under range conditions, this species is capable of producing 400-900 seeds per seed head (up to 40,000 per sq m), with the ability to germinate under a variety of conditions varying from 7-34°C (Watson and Renney 1974). Seeds are dispersed when the plant breaks off at the base, with the many spreading branches of the plant giving it a tumbleweed appearance and mobility. The urn-shaped seed heads, which open gradually to disperse seeds slowly, also contribute to long distance distribution. Tumbleweeds can be transported by vehicles and similar vectors. Additionally, seeds can be transported via water, wildlife, contaminated seed, foot-traffic etc. (Roche and Roche 1991).

Origins and Distribution: Diffuse knapweed is native to the grassland steppes of southeastern Europe, and was introduced to the United States via contaminated alfalfa seeds and hay (Sheley 1998). The plant can thrive in semi-arid and arid conditions and a variety of altitudes (observed at 150-900m) which allow it to be a problem in the western United States, especially Washington, Montana, Idaho, Oregon, and California (Maddox 1982). Diffuse knapweed prefers open habitats to shaded areas and cannot tolerate cultivation or excessive moisture (Watson and Renney 1974). Since the plant was first reported in 1907, it has spread to more than 3.2 million acres across Washington, Idaho, Oregon, Montana, and British Columbia.

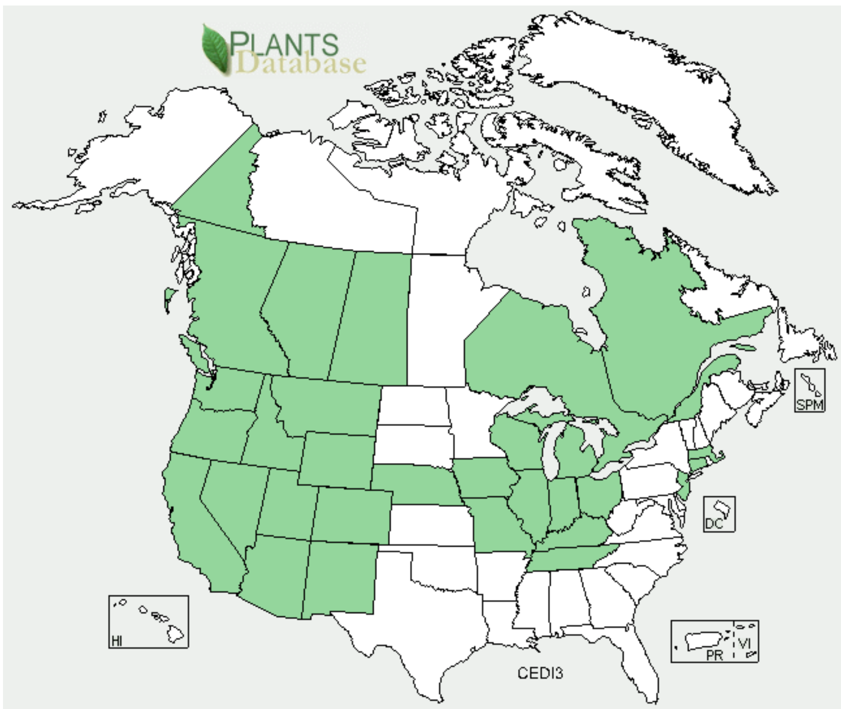

Figure 9. Distribution of Diffuse Knapweed.

Probability of Future Expansion: Currently found primarily in the western United States, diffuse knapweed is easily transported via many natural and anthropogenic vectors. Locally the plant makes aggressive, slow-paced increases in its frontage if uncontrolled. On larger geographic scales, the plant's ability to spread via transport on vehicles and through similar sources of contamination is a risk factor for spread, especially for the military as tactical vehicles are often transported across long distances. The competitive success is also due to prolific seed production, high seed viability, perennial reproductive cycles, and the absence of natural enemies (Lacey 1990). The plant may complete its life cycle in a single year, or maintain rosette form for many years before flowering, giving the plant the advantage of being well-established before control agents can effectively attack it. Additionally, because of the long tap root, the plant is able to extract more moisture from the ground and remain green longer than the native species. The allelopathic characteristics of the plant also allow it to form dense patches, and residual allelopathic chemicals (which are found in the leaves and ground upon decomposition) hinder regrowth of natural grass (Fletcher and Renney 1963). Additionally, a persistent seed bank once the plant is established could allow for new infestations.

Control Technologies: Diffuse knapweed can be controlled with herbicides, biological control agents and hand pulling. Picloram (Tordon), clopyralid (Transline), dicamba (Banvel), 2,4-D, clopyralid + 2,4-D (Curtail) and dicamba + 2,4-D are effective at controlling diffuse knapweed (Beck et al. 2004; Sheley et al. 1998). Apply picloram from rosette to early bolting, clopyralid or clopyralid + 2,4-D from mid-bolt to bud, dicamba or 2,4-D when weeds are actively growing, and dicamba + 2,4-D when weeds are in the rosette stage. Picloram, clopyralid, clopyralid + 2,4-D and dicamba can also be applied in the fall (Beck 2008; Beck et al. 2004). Dicamba and 2,4-D alone will provide only short-term control and therefore need to be applied annually until the seedbank is depleted (Sheley et al. 1998). Refer to herbicide labels for specific instructions regarding rates, adjuvants, application techniques, and use restrictions.

A number of biological control agents have also been used to control diffuse knapweed. They include seedhead-feeding flies (*Urophora affinis* and *U. quadrifasciata*), root beetles (*Sphenoptera jugoslavica*), and seedhead weevils (*Larinus minutus*). On many sites, suites of biological control agents have been released; however, *Larinus* appears to be the most effective (Sheley et al. 1998; Seastedt et al. 2007). Livestock such as goats, cattle, and sheep will feed on diffuse knapweed. Sheep will typically only feed on young, tender growth. Cattle have been shown to decrease seed set by 50% when grazed on diffuse knapweed that was bolting and 6–12 in. tall (Beck 2008).

Hand pulling is effective only when the entire plant (including the root crown) is removed prior to seed production; however, due to the amount of time and labor required, hand pulling may only be practical on small infestations (Sheley et al. 1998). Regardless of the control method used, diffuse knapweed requires long-term control to deplete the seed bank and successful management requires an integrated strategy including the establishment of competitive plants (Sheley et al. 1998). Research is ongoing to identify effective integrated management strategies for control of diffuse knapweed.

Direct and Indirect Military Impacts: The species is linked to increased soil erosion potential and wildlife habitat degradation (Roche and Roche 1991). These factors are important in training terms as the species favors the same open rangelands valued for maneuver training. Increased soil erosion and negative impacts on TES habitat will reduce training capacity.

Installations (10): Fort Lewis, Fort Hunter Liggett, Fort Irwin, Fort Bliss*, Fort Carson, Fort Leonard Wood*, Fort McCoy*, Atterbury Reserve Training Area*, Fort Knox*, Fort Campbell*.

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Photo credits:

Figure 7: Cindy Roche (Bugwood.org)

Figure 8: Richard Old, XID Services, Inc. (Bugwood.org)

Figure 9: USDA Plants Database (plants.usda.gov)

Garlic Mustard (*Alliaria petiolata*): Garlic mustard is a cool-season biennial forb that grows to 12–48 in. tall with a long taproot. It has large heart-shaped leaves and has very variable growth habits, as the plant is very phenotypically plastic and can adapt well to a wide range of habitats (Rodgers et al. 2008; Lewis et al. 2006). First year plants have a basal rosette of dark green leaves and grow to produce several flowering stalks in the second year. Flowers are white, terminal, tightly clustered, and four-petaled (Rodgers et al. 2008). The species is shade-tolerant, and is a rapidly spreading competitive exotic that is predominantly associated with woodlands and dense-shade, but can also grow on sunny sites (Meekins and McCarthy 1999). Allelopathic compounds, especially gluconsinulates, have been found in garlic mustard and possibly contribute to its invasive potential (McCarthy and Hanson 1998).

Figure 10. Flowering Garlic Mustard.

Figure 11. Garlic Mustard plant.

Reproductive Biology: Garlic mustard plants are self-compatible and very often self-pollinate before the flower is open, making selfing a dominant breeding system (Durka et al. 2005). Plants typically germinate early in the spring and form a rosette in the first year. After overwintering as a rosette, flowering stems develop in March or April (Meekins and McCarthy 2001; Durka et al. 2005). Almost all pollination results in viable seeds, and those seeds that do not germinate form seed banks that last for 10 years (Rodgers et al. 2008; Meekins and

McCarthy 2001). Garlic mustard stands are capable of producing more than 62,000 seeds per square meter, which are dispersed through a variety of methods, including on the fur of animals, through waterways, and by anthropogenic vectors (Lewis et al. 2006).

Origins and Distribution: Garlic mustard was first reported in North America on Long Island in 1868 and presently is listed as a noxious weed in 6 of the 34 continental states in which it is found (Meekins et al. 2001; Durka et al. 2005). Originally from Europe, garlic mustard's native range extends from as far south as Italy to as far north as Sweden. It has been suggested that colonists introduced garlic mustard as a medicinal herb (Meekins et al. 2001). High allelic diversity in the United States indicates that there were probably multiple introductions (Durka et al. 2005). Garlic mustard typically invades second growth forests, and can also be found along roadsides, floodplains, and on forest margins and openings (Meekins and McCarthy 1999). The plant does not require disturbance to invade an area, and has been known to invade high quality forests (Lewis et al. 2006).

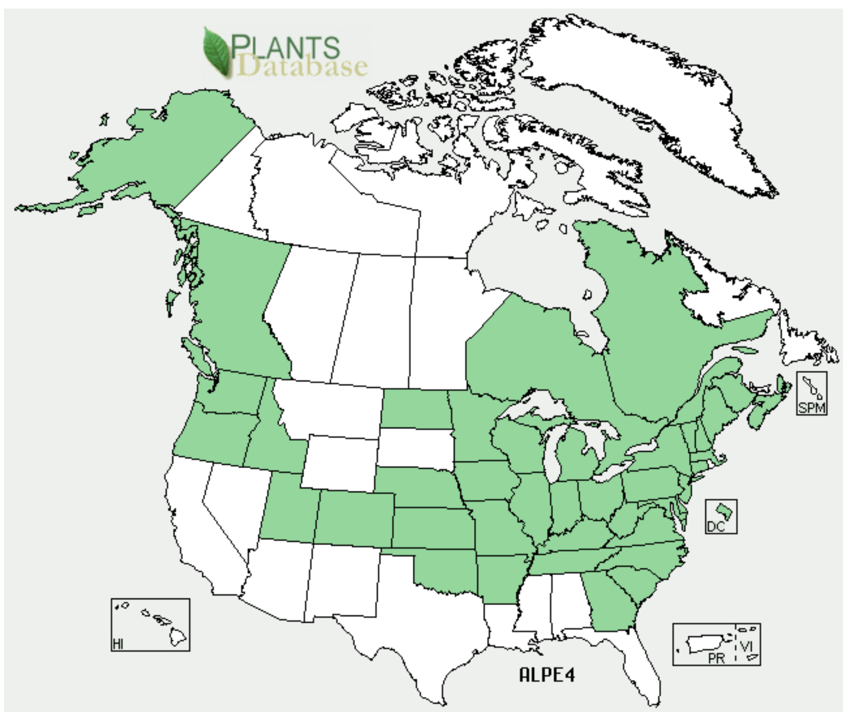

Figure 12. Distribution of Garlic Mustard.

Probability of Future Expansion: Garlic mustard is a difficult plant to control due to many life history characteristics that make it a particularly competitive invasive. In the United States, the estimated rate of expansion is 64,000 square kilometers per year (Rodgers et al. 2008). The phenology of the plant is also substantially different from native plants, resulting in garlic mustard growing earlier in the spring and acquiring large portions of the available nutrients before native plants are able to. Additionally, allelopathic compounds may interfere with native plant germination, ability to form symbiotic relationships, and overall growth (Meekins and Hanson 1998; Rodgers et al. 2008).

Control Technologies: Garlic mustard can be controlled with herbicides, hand pulling, cutting, or burning. The most commonly used herbicide for garlic mustard control is glyphosate (Roundup). Glyphosate can be applied in early spring or late fall but is non-selective so care should be taken to avoid non-target plants. Garlic mustard will continue to grow unless temperatures fall below 35°F or if there is snow cover; therefore, the safest time to apply glyphosate is when native plants are dormant but garlic mustard is still growing. Triclopyr (Garlon) is also effective at controlling garlic mustard in the early spring (Hoffman and Kearns 1997; Miller 2003; Nuzzo 1991). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Hand pulling garlic mustard can be effective for small infestations. Plants should be pulled before seed formation and the upper half of the root must be removed to prevent new flowering stalks from forming (Hoffman and Kearns 1997; Miller 2003). If cutting garlic mustard, cut flowering stalks close to the soil surface once flowering begins (Hoffman and Kearns 1997). Burning in early spring or fall can be effective but may require 3–5 years of treatment (Hoffman and Kearns 1997). Garlic mustard seeds can remain viable for up to 5 years; therefore, regardless of treatment used, areas should be monitored and new plants removed for at least 5 years (Hoffman and Kearns 1997). There are currently no biological control agents for garlic mustard, but research is ongoing to identify and develop new agents.

Direct and Indirect Military Impacts: Garlic mustard invades the understory of forests where it will displace many native plant species and disrupt plant-species interactions (Durka et al. 2005). Additionally, garlic mustard has been associated with an almost complete decline in the mycorrhizal status of native plants. The plant has been shown to impact butterfly populations by reducing egg success when laid on garlic mustard leaves instead of native leaves (USFWS 2004. As such, garlic mustard may also have other unknown impacts on other important pollinators. These negative impacts on plant communities and pollinators may produce TES concerns that impact Army lands in the future.

Installations (16): Fort Lewis*, Fort Carson, Fort Riley, Fort Leonard Wood*, Fort Chaffee*, Fort McCoy*, Atterbury Reserve Training Area*, Fort Knox, Fort Benning*, Fort Campbell*, Fort Stewart*, Fort Jackson*, Fort Bragg*, Fort A.P. Hill, Fort Pickett*, Fort Indiantown Gap*

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Photo credits:

Figures 10 and 11: Chris Evans, River to River CWMA (Bugwood.org)

Figure 12: USDA Plants Database (plants.usda.gov)

Giant Reed (*Arundo donax L.*): Giant reed is an invasive perennial grass that can grow over 20 ft in height and has leaves over 1-ft long, that are broad at the base and taper to a point (Stuhlman 1947). The panicles are long and plume-like. Nodes in the stem reinforce the plant at distances of approximately 8 in. (Spatz et al. 1997). Plants are often arrayed either in phalanx (tightly packed) to guerrilla (loosely packed) arrangements, with the plant exhibiting great amounts of plasticity in terms of where they fit in the continuum (Decruyenaere and Holt 2005). The plant can tolerate a wide variety of conditions, including high salinity, and can grow in many soil types from loose sand to heavy clays. It grows best in well-drained soils where abundant moisture is available (Mackenzie 2004). Giant reed has invaded riparian habitats across the coastal waters of the United States and spreads rapidly, displacing native vegetation and modifying several important physical and chemical site characteristics (Decruyenaere and Holt 2005).

Figure 13. Giant Reed stalk.

Figure 14. Giant Reed infestation.

Reproductive Biology: Giant reed primarily reproduces through rhizomes that root and sprout readily. Clonal growth predominates in this species, and although the plant has been observed to flower in California, the seeds are not viable (McWilliams 2004). Fragmented pieces of stems and rhizomes spread downstream during flooding, and it was reported that over 90% of the fragments sprouted if they contained a node (Boose and Holt 1998). Additionally, rhizome pieces have been reported to establish at any time of year. Spring and summer are the main growing seasons for giant reed, and shoot growth has been noticed primarily in March to August (Decruyenaere and Holt 2001). The rapid spread of the plant is also due to its high levels of photosynthesis (Decruyenaere and Holt 2005).

Origins and Distribution: Giant reed is thought to have originated in Asia and is now widespread throughout southern Europe, the Middle East, Australia, North Africa, and North and South America. Riparian zones in California have experienced severe invasive damage from giant reed since its introduction as a means of erosion control in drainage canals in the early 1800s (Boose and Holt 1998; Quinn and Holt 2008). Giant reed mainly invades riparian areas or wetlands but can become established anywhere where the water table is near or at ground level. Giant reed has been reported in prairie, meadows, sagebrush, and grasslands (McWilliams 2004).

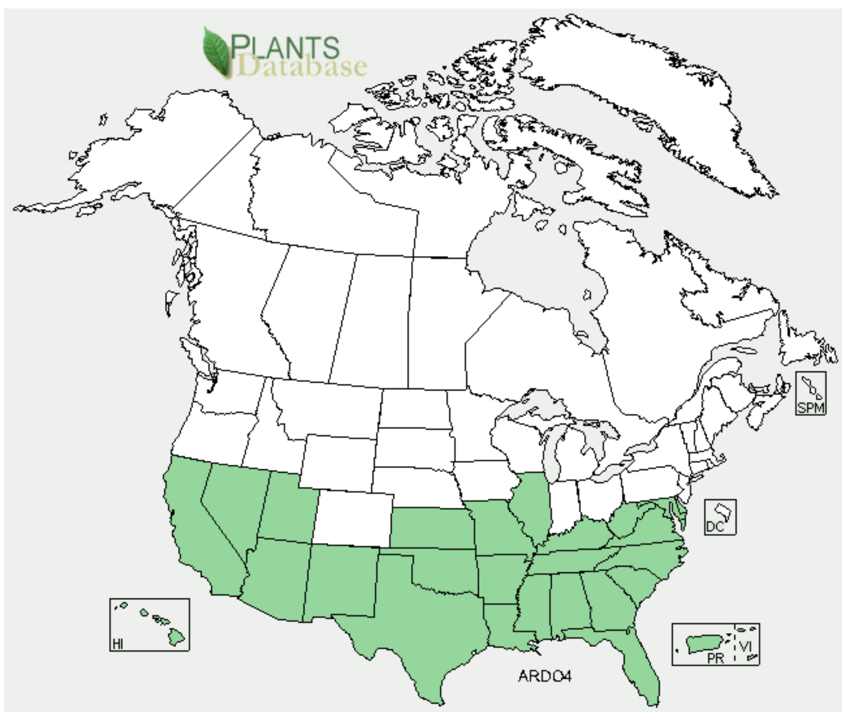

Figure 15. Distribution of Giant Reed.

Probability of Future Expansion: Giant reed has made inroads in Virginia, Kentucky, and other eastern states and has overtaken large areas of the American Southwest and northern Mexico. Rate of spread can be very aggressive, as illustrated by the species' invasion of Texas where it expanded its range from 20,000 to 60,000 acres between 2002 and 2007. Locally the species spreads through wet areas during flooding by means of rhizome dispersal. Disturbance and transport of vegetative propagules (Miller 2003) are responsible for spread across geographic regions. The prolific asexual reproduction of the plant makes giant reed particularly difficult to control (Boose and Holt 1998).

Control Technologies: Effective control of giant reed requires the rhizome and root mass to be killed. Foliar applications of the systemic herbicides glyphosate (Rodeo), imazapyr (Habitat) or imazamox (Clearcast) are currently the most effective means to control giant reed (BASF 2008; Bell 1997; Neill 2006; Vollmer et al. 2008). Herbicide applications should occur after flowering but prior to dormancy, typically between mid-August to early November (Bell 1997). Due to the extensive root and rhizome mass, repeat applications are necessary to control regrowth. Integrating herbicides with mechanical techniques such as mowing or cutting has also been shown to be effective. Giant reed can be cut, followed by a foliar herbicide application 3–6 weeks later when new plants have appeared. This technique uses less herbicide and allows for better coverage (Bell 1997). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Direct and Indirect Military Impacts: Given that the species is dominantly found in wet areas that are less favorable to training, impacts of giant reed on training lands are felt to be more ecological than training restrictive. Although undocumented, the plant’s growth form may cause line-of-sight issues with laser and similar training devices. Additionally, the plant can undermine flood control efforts, increase erosion, and alter wildlife habitat (Boose and Holt 1998; Fall et al. 2004).

Installations (18): Fort Hunter Liggett, Fort Irwin*, Fort Bliss, Fort Hood, Fort Sam Houston, Fort Sill*, Fort Riley*, Fort Leonard Wood*, Fort Chaffee*, Fort Polk*, Fort Campbell, Fort Knox*, Fort Rucker*, Fort Benning*, Fort Stewart*, Fort Jackson*, Fort Bragg*, Fort A.P. Hill*, Fort Pickett*.

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Photo credits:

Figure 13: David J. Moorhead, University of Georgia (Bugwood.org)

Figure 14: Chris Evans, River to River CWMA (Bugwood.org)

Figure 15: USDA Plants Database (plants.usda.gov)

Japanese honeysuckle (*Lonicera japonica*): Japanese honeysuckle is a twining, perennial, semi-evergreen woody vine that climbs other objects. It has been listed as one of the worst pest species in managed forests, and it often grows over and overshadows small trees, shrubs, and other herbaceous vegetation (Barden and Matthews 1980; Dillenburg et al. 1993). Japanese honeysuckle stems are green when pubescent and, upon reaching maturity, they are densely tangled with a reddish-brown hue, becoming woody and developing shredded bark. The stems can reach 105-226cm, with 2-3 branches per stem, which are 2-60cm long. The roots can reach 30-50cm when moisture is available, and up to 102cm on sites where moisture is scarce (Schierenbeck 2004). Japanese honeysuckle displays huge amounts of phenotypic plasticity in response to available resources, and the ability to climb objects or not to climb objects results in the plant's increased ability to place plant modules that take full advantage of conditions. Lateral runners of the root system aboveground also give Japanese honeysuckle the advantage of further expansion and exploitation of new habitat for more of the twining stems (Williams and Timmins 1998).

Figure 16. Flowering Japanese Honeysuckle.

Figure 17. Japanese Honeysuckle along fencing.

<u>Reproductive Biology</u>: Japanese honeysuckle may grow vegetatively through the extensive root system and its lateral runners or through seed production. Most Japanese honeysuckle seed germinates in late February through the first weeks of April, and appear to only be limited by water availability, heavy frost, and temperatures necessary for stratification (Fowler and Larson 2004; Regehr and Frey 1988). Germination occurs best in disturbed areas, such as areas in forests where light gaps occur. Japanese honeysuckle flowers from mid-May to mid-June and is pollinated diurnally and nocturnally by hawkmoths and bees (Miyake and Yahara 1998). Flowers typically open just before dusk, and wilt after about 3 days. Additional flowering can

occur in September (Pair 1994). Dispersal of seeds occurs through the variety of bird and mammal species that eat the fruit, with this immediate dispersal technique greatly facilitating the typically low seed viability of Japanese honeysuckle. Local growth occurs through the lateral runners, which can grow up to 15m away from the parent root in a single growing season (Williams et al. 2001).

Origin and Distribution: Japanese honeysuckle is native to the thickets and hills of Japan, Korea, and China, and there is evidence that this plant has been used medicinally in that region since the Tang dynasty in 659 AD (Schierenbeck 2004). It has been distributed as an ornamental and continues to be a popular component in tea. Japanese honeysuckle was introduced to the United States in 1806 by William Kerr, a collector and gardener (Schierenbeck 2004; Williams and Timmins 1998). It escaped from cultivation in 1882, and with additional spread propagated by animal dispersal, ornamental cultivation, and further plantings for land stabilization, Japanese honeysuckle reached its current distribution in 42 states. Japanese honeysuckle grows well in fertile soils and full sunlight, and can be found on road cuts, abandoned fields, fences, woodlots, and crevices in walls (Schierenbeck 2004).

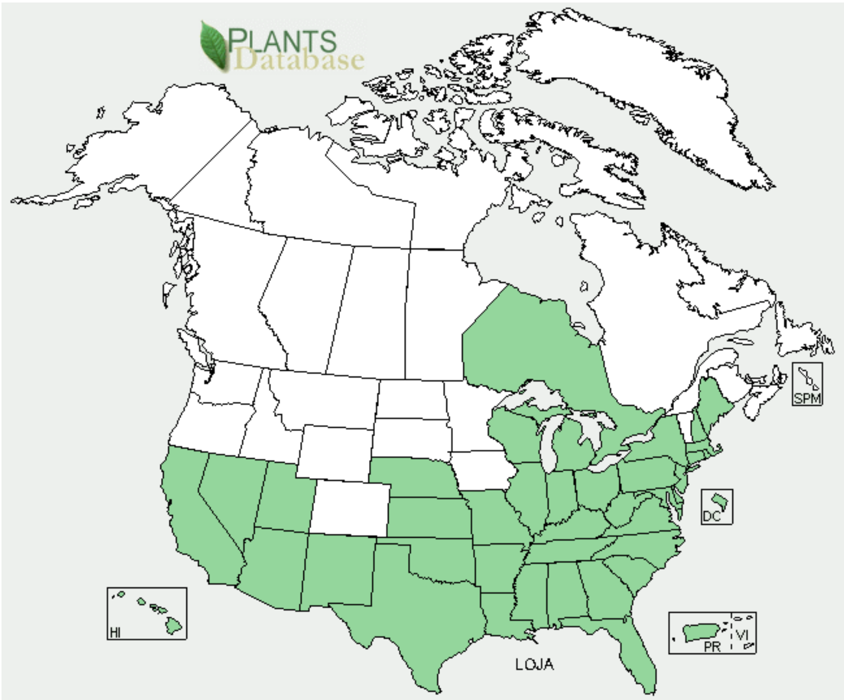

Figure 18. Distribution of Japanese Honeysuckle.

Probability of Future Expansion: Japanese honeysuckle is a prolific seed producer, and the lateral roots also assist in gaining new ground. The plant is capable of reaching up to 15m high on the object it climbs, and can grow a mat of vegetation up to 2m deep on top of itself and the object. Fire can reduce aboveground biomass, but the plant survives and re-sprouts from the root stem (Barden and Matthews 1980). It suppresses ground vegetation and foliage production of forest vegetation, which leads to further dominance by Japanese honeysuckle (Fowler and Larson 2004). Varieties of the plant are still available commercially as ornamentals. Additionally, there are no known pests or diseases that affect Japanese honeysuckle.

Control Technologies: Japanese honeysuckle can be controlled with herbicides such as glyphosate (Roundup) or 2,4-D + triclopyr (Crossbow). These herbicides should be applied in the fall before a hard freeze (Nyboer 1992). Japanese honeysuckle plots treated with glyphosate in October showed excellent control 30 months after treatment with no recovery (Regehr and Frey 1988). By applying later in the year, non-target vegetation is dormant thereby minimizing damage. Due to the dense growth of Japanese honeysuckle, retreatment may be necessary to remove plants that may have been missed with initial herbicide applications (Nyboer 1992). Japanese honeysuckle may also be cut at the soil surface and the cut stem treated with glyphosate or triclopyr (Garlon). This treatment can be applied from July to October and is safe to surrounding plants (Miller 2004). Yeiser (1999) also reported a June application of metsulfuron (Escort) provided good control of Japanese honeysuckle for three growing seasons in bottomland hardwood stands. Refer to herbicide labels for information regarding, application rates, adjuvants, application techniques, and use restrictions.

Burning may also be used to control Japanese honeysuckle. Spring burns can reduce honeysuckle crown volume and coverage. Repeated fires can reduce honeysuckle by 50%. Herbicides can also be applied after fire has reduced plant coverage, resulting in less herbicide being used (Miller 2004; Nyboer 1992). Although mowing and grazing can reduce the spread of honeysuckle, these control measures are not effective (Nyboer 1992). There are currently no known biological control agents for Japanese honeysuckle.

Direct and Indirect Military Impacts: Japanese honeysuckle reduces native vegetation and severely alters species composition, and it has become a serious pest of young pine plantations, where it completely smothers the new trees (Pair 1994). Through additional root competition, Japanese honeysuckle further reduces the biomass and productivity of young trees (Surrette and Stephen 2008). This poses serious threats to commercial forestry. Additionally, training exercises can be affected and materials can be damaged if Japanese honeysuckle is disregarded when it is actually covering a small tree or a large stump.

Installations (20): Fort Hunter Liggett*, Fort Irwin*, Fort Bliss*, Fort Sam Houston, Fort Hood*, Fort Sill*, Fort Riley, Fort Leonard Wood*, Fort Chaffee, Fort Polk, Fort Rucker, Fort McCoy*, Atterbury Reserve Training Area*, Fort Campbell, Fort Jackson, Fort Knox, Fort Bragg, Fort A.P. Hill, Fort Pickett, Fort Drum*.

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Photo credits:

Figure 16: Chuck Bargeron, University of Georgia (Bugwood.org)

Figure 17: Charles T. Bryson, USDA Agricultural Research Service (Bugwood.org)

Figure 18: USDA Plants Database (plants.usda.gov)

Kudzu (*Pueraria montana*): Kudzu is a woody perennial vine, with the ability to grow extremely quickly in a variety of environments. It is deciduous and capable of growing stems as long as 20-30m in a single growing season, with a very well established root system. Stem elongation rates are estimated to be anywhere from 3–19 cm per day (Foresth and Innis 2004). Kudzu is capable of creating multiple canopy layers and is a high-climbing, highly aggressive vine that can decimate full grown tree stands. Three-leaflet leaves are alternately spread around the plant, and are hairy and green on both sides. Flowers occur mostly in short-stalked clusters, have a purple hue, and can grow up to 2.5cm across (Guertin et al. 2008). As a structural parasite, kudzu can climb surrounding objects (i.e., plants or man-made structures) to reach higher levels of light, out competing surrounding vegetation (Foresth and Innis 2004).

Figure 19. Flowering Kudzu.

Figure 20. Severe Kudzu infestation.

Reproductive Biology: Kudzu is known to reproduce primarily vegetatively, which takes place when stems come in contact with the ground and form roots from the stem nodes (Guertin et al. 2008). Sexual reproduction is possible in kudzu, and percentages of seedlings surviving past germination vary according to geographical location, with more seedlings germinating in populations in the southern regions of the United States. Sexually, kudzu is also pollinator limited (Foresth and Innis 2004). Additionally, seed germination is limited by the fact that seed scarification is necessary. Frequent rooting can occur mostly through vines that come in contact with the ground or from segments of stem that senesce, resulting in much higher levels of asexual reproduction. Spread of the plant in an area primarily occurs through the extensive vines and carbon fixation.

Origins and Distribution: Kudzu was first introduced into the United States in 1876, from the Centennial Exposition in Philadelphia, PA. Since that time, deliberate plantings of kudzu in the southern United States were to control erosion, as a commercial source of fiber, and to provide fodder for cattle. These many and varied introductions give the plant and its respective populations a high level of genetic diversity (Foresth and Innis 2004; Guertin et al. 2008). It was listed as a federally noxious weed in the late 1970s and has continued to spread due to of a lack of natural pathogens or predators. The plant is found in a variety of places, including untended fields, forests, roadsides, and pastures. It has a wide range of altitudes that it can grow in (up to 2000m), and can perform well in all soil types except in poor sandy soils or poorly drained clay soils (Guertin et al. 2008). The most severe infestations of kudzu occur in the southeastern continental United States, especially Alabama, Georgia, and Mississippi.

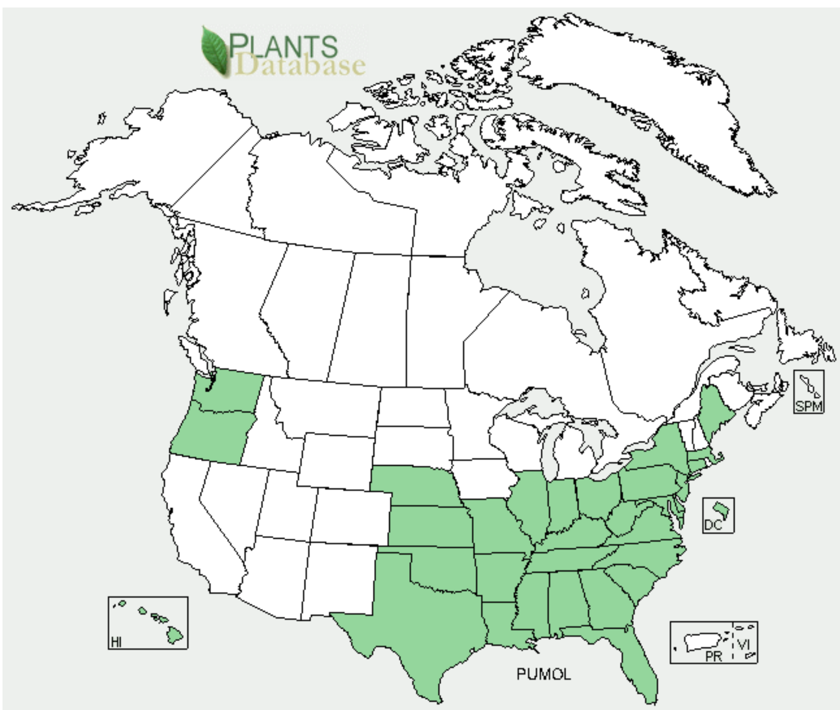

Figure 21. Distribution of Kudzu.

Probability of Future Expansion: Kudzu's large tuberous roots and its ability to quickly refoliate after disturbance, make it extremely hard to exterminate. Eradication becomes more difficult with time as the plant develops large roots that sore stach, which makes them more resilient to control. The current estimate of kudzu spread within the Eastern United States is 50,000 hectares a year (Foresth and Innis 2004). Anticipated changes in the Eastern United States include higher temperatures, higher carbon dioxide (CO_2) levels, and increased habitat fragmentation. Kudzu's growth rate responds positively to these changes, and these effects will favor Kudzu's aggressive vegetative reproduction characteristics.

Control Technologies: Kudzu can be controlled with herbicides, grazing, or mowing. Four herbicides that control kudzu are picloram (Tordon), metsulfuron (Escort), triclopyr (Garlon) and clopyralid (Transline). Picloram should be applied from June through September when plants are actively growing, and the last application of the year should be made 1 month prior to the first frost. Higher application rates are required for plants that have been established for more than 10 years, and rainfall is necessary within 2–5 days after herbicide application to carry herbicide into the upper soil layer to achieve acceptable control. Picloram is the most effective of all aforementioned herbicides since it can be taken up via the foliage as well as the roots. Metsulfuron, triclopyr, and clopyralid may require 2–10 annual applications and work best on kudzu that is less than 10 years old. These herbicides should be applied after midsummer during flowering, and regrowth should be expected. Other herbicides that can be used include imazapyr (Arsenal) and fosamine (Krenite). Imazapyr should be applied after midsummer, and fosamine should be applied at the end of summer (Harrington et al. 2003; Miller 1996). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Kudzu can also be controlled with livestock grazing, and studies have found that grazing for 2–4 years can eliminate kudzu stands (Miller and Boyd 1983; Luginbuhl et al. 1996). Cattle, goats, and sheep have all been shown to be efficacious and vines should be cut from trees and other structures so that livestock can reach foliage (Miller 1996). Although mowing can be difficult and labor intensive, it can control kudzu if done on a monthly basis for at least 2 years. Kudzu tubers and roots must be depleted for mowing to be successful. Both grazing and mowing may only be practical for small infestations.

There are currently no insect biological control agents to control kudzu; however, three pathogens have been studied (Boyette et al. 2002; Frye et al. 2007; Weaver and Lyn 2007). Of the three, only the fungal pathogen *Myrothecium verrucarua* has shown the greatest potential. *M. verrucarua* infects kudzu stems and leaves, and field tests demonstrated 100 percent control could be achieved within 14 days (Boyette et al. 2002). *M. verrucaria* has also been successfully integrated with herbicides and is currently being developed as a bioherbicide (Weaver and Lyn 2007).

<u>Direct and Indirect Military Impacts</u>: Kudzu forms large, lush green tangles of foliage that cover large areas of ground and extend high into tree tops. This sprawling growth form can directly impact the training mission in that vegetative cover interferes with dismounted troop movements and training equipment that requires a direct line of sight. Foliage can easily cover tree stumps, ditches, and other obstacles causing damage or injury to equipment or personnel traversing the area. Indirectly, kudzu can impact the training mission through its ability to damage sensitive habitat and degrade installation infrastructure. Kudzu’s aggressive growth form will dominate habitats, altering soils, plant structure, and species composition. Additionally, kudzu vines and foliage can engulf parking lots, buildings, power lines, and other infrastructure. The plant forms dense vegetative mats that trap moisture and may accelerate the deterioration of concrete and masonry. The vines can also interfere with the operation of power lines and similar structures.

<u>Installations (19)</u>: Fort Lewis*, Fort Sill*, Fort Riley*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee*, Fort Polk, Atterbury Reserve Training Area*, Fort Knox, Fort Campbell, Fort Rucker, Fort Benning*, Fort Stewart¸ Fort Jackson*, Fort Bragg*, Fort A.P. Hill, Fort Pickett, Fort Indiantown Gap.

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Photo credits:

Figure 19: Forest and Kim Starr, U.S. Geological Survey (Bugwood.org)

Figure 20: Robert L. Anderson, USDA Forest Service (Bugwood.org)

Figure 21: USDA Plant Database (plants.usda.gov)

Leafy Spurge (*Euphorbia esula*): Leafy spurge is an erect, branching, perennial shrub 2–3.5-ft tall, with smooth stems and yellow flower. Stems occur from a vertical root that can extend many feet underground, and are typically found in clusters rather than single stalks. Clusters resemble flax and are conspicuous because of the bluish-green leaves and yellowish-green flowers (Hanson 1934). Additionally, the plants are easily recognized by their many long and narrow leaves, 2-3 in. long and 0.25-in. wide (Higgins and Ames 1965). The yellowish-green clusters are not the actual flowers of the plant and typically appear 2–3 before actual flowering (Lym 1998). It is one of the first plants to emerge in the spring, and its highly efficient root system consists of coarse and fine roots, which occupy a large volume of soil (Lym 1998; Hanson and Rudd 1933).

Figure 22. Leafy Spurge cluster.

Figure 23. Invading Leafy Spurge on a roadside.

<u>Reproductive Biology</u>: Leafy spurge is capable of reproducing asexually and sexually (Bakke 1936). Leafy spurge reproduces through seeds that have a high germination rate. Seeds are easily transportable by water and wildlife and can stay viable for years, complicating eradication. As the seed capsules ripen, pressure causes them to break open, launching seeds as far as 20 ft from the parent plant (Hanson 1934; Higgins and Ames 1965). Seeds become ripe in early to mid-June and may continue into September. Asexually, the plant can regenerate from root fragments as small as 0.5-in. long and 0.125-in. wide (Hanson and Rudd 1933). Leafy spurge has a very well developed and

highly efficient root system, which begins to branch into many large, woody branches near the surface. The roots are able to reach depths lower than competing plants, and the greatest depth recorded was 15 ft, 8 in. (Hanson and Rudd 1933; Bakke 1936). The roots may also extend laterally, with the greatest recorded spread of 3.5 ft from the parent plant. Particularly extraneous root buds may also become new plants (Best et al. 1980). Additionally, the plant tends to display evidence of allelopathic advantages (Steenhagen and Zimdahl 1979).

Origin and Distribution: Samples from across North America have shown that leafy spurge is not a single species but an aggregate of closely related variants, suggesting that multiple strains were imported at different times from many different regions of its native range (such as Europe and Asia) in grass, cereal seed, or ship ballast. Since its introduction, the plant has become a serious management problem, particularly for the north and central plains states. It occurs in fields, pastures, roadsides, abandoned fields, rangelands, prairies, streams, ditches, and other waste places (Steenhagen and Zimdahl 1979). States with the greatest infestations include Colorado, Idaho, Minnesota, Montana, Nebraska, North Dakota, Oregon, South Dakota, Wisconsin and Wyoming.

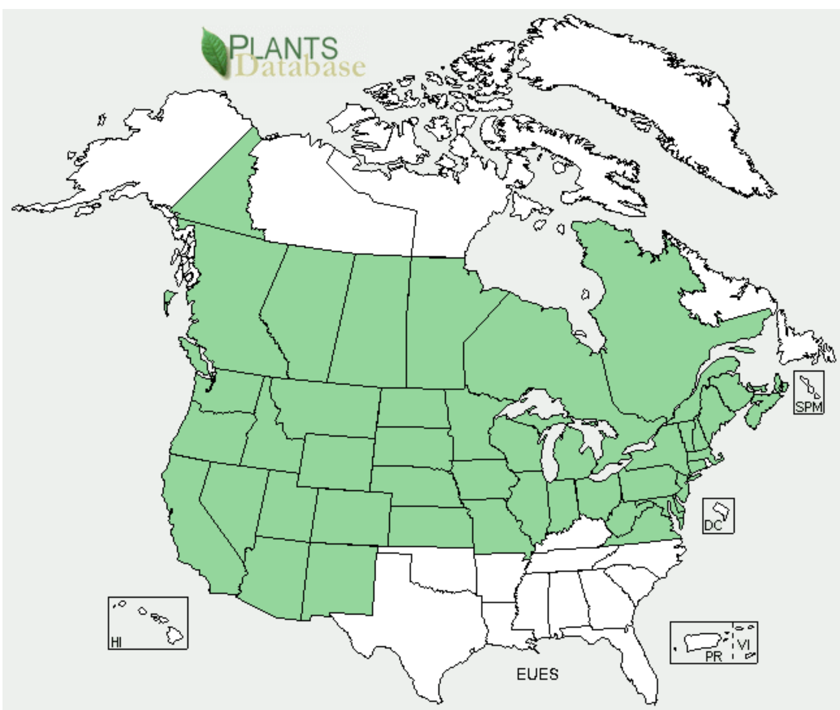

Figure 24. Distribution of Leafy Spurge.

Probability of Future Expansion: The plant’s highly efficient and extensively networked root system greatly contributes to its spread, and increased cultivation and plowing of the weeds only leads to further colonization because of the spread of root fragments (Hanson 1934). Additionally, the roots are very effective storage mechanisms and can store food supplies for the plant to overwinter (Leistritz et al. 1992). The allelopathic effects of the plant make it difficult to introduce other species as controls, and its ability to sustain itself despite repeated herbicide treatments shows that eradication is difficult (Leistritz et al. 2004). The lack of natural enemies gives the plant yet another competitive edge, and the plant's ability to germinate and grow in a variety of soil types means that its spread is completely independent of topographical constraints (Lym 1998). Leafy spurge has been reported to double in acreage every 10 years for the past 100 years (USDA-ARS 2002a).

Control Technologies: Leafy spurge can be controlled with herbicides, grazing, or biological control agents (Joshi 2008; Walker et al. 1994; USDA-ARS 2002b). A number of herbicides can effectively control leafy spurge and include picloram (Tordon), picloram + 2,4-D, dicamba (Banvel), imazapic (Plateau), glyphosate (Roundup), or glyphosate + 2,4-D (Campaign) but, no single treatment will eradicate leafy spurge. Timing of herbicide application is important and varies for each herbicide. Picloram and picloram + 2,4-D should be applied during the true flower growth stage or fall regrowth. Picloram after 1 year and picloram + 2,4-D after four annual treatments can provide 85–95% control. Dicamba applied during the true flower growth stage or fall regrowth can provide 95% control after three annual treatments. Imazapic should be applied in early to mid-September and provides 90% control 1 year after treatment. Glyphosate can control leafy spurge by 80–90% when applied after 1 July to actively growing plants. Glyphosate + 2,4-D should be applied at seed set or when plants are actively growing in the fall. Glyphosate + 2,4-D can provide 95% control when applied in late June followed by application of picloram + 2,4-D the following year (USDA-ARS 2002b). Refer to herbicide labels for information regarding application rates, adjuvants, application techniques, and use restrictions.

Grazing of sheep and goats has also been used to control leafy spurge. Goats have demonstrated a higher preference for leafy spurge than sheep and are more effective (Walker et al. 1994). Grazing should begin in early spring when leafy spurge first emerges, but if grazing is stopped, leafy spurge can regrow from roots (Landgraf et al. 1984; Bowes and Thomas 1978). Of the biological control insects, flea beetles (*Aphthona spp.*) have been the most successful (Lym 1998). Flea beetles can reduce root and stem density but may not establish well at all release sites (Kirby et al. 2000). A gall midge (*Scurgia esulae*) which causes stem tip galls can reduce seed production and has been most successful near wooded areas (Lym 1998). Biological control agents can take many years to cause a significant reduction in leafy spurge.

Integrated pest management plans have been successful in controlling leafy spurge populations. Flea beetles have been used with herbicides and grazing and can provide better control than either agent used alone (Joshi 2008; Lym 2005). Although burning alone does not reduce leafy spurge density, it can be integrated with herbicides and flea beetles. Burning can open the canopy by removing thatch and therefore improve herbicide coverage. Removing thatch will also allow flea beetles to lay eggs on the soil surface instead of plant thatch increasing larval survival (Lym 2005). The best strategy for controlling leafy spurge is an integrated one. Research is ongoing to identify effective integrated management strategies for control of leafy spurge.

<u>Direct and Indirect Military Impacts</u>: Leafy spurge can out-compete native species, thereby altering habitat and soil erosion/nutrient properties. Expansion leads to a decline in native plants, resulting in increased biodiversity loss, causing runoff and erosion (Leistritz et al. 2004). These changes can directly reduce training through less stable soils and indirectly reduce training by reducing habitat for grassland TES.

<u>Installations (11)</u>: Fort Lewis¸ Fort Hunter Liggett*, Fort Irwin*, Fort Bliss, Fort Carson, Fort Riley*, Fort Leonard Wood*, Fort McCoy*, Fort A.P. Hill*, Fort Indiantown Gap, Fort Drum.

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Photo credits:

Figure 22: Chris Evans River to River CWMA (Bugwood.org)

Figure 23: Leslie J. Mehrhoff, University of Connecticut (Bugwood.org)

Figure 24: USDA Plants Database (plants.usda.gov)

Multiflora Rose (*Rosa multiflora*): Multiflora rose is a short, thorny, diffusely branched, perennial shrub with numerous arching canes arising from the crown. It is easy to distinguish from other wild roses because of its large size and its numerous thorns (Doll 2006). Individual plants can reach up to 6.5m in diameter and 3m in height in full sunlight. In shady conditions, canes can grow on trees and may reach lengths of 6m or more. The stems are green to reddish in color and the leaves of the plant are pinnately compound, with 5 to 11 leaflets. After the plants have become established, multiple stems arise from the root crown, and physical removal of the plant is difficult because the entire root crown needs to be removed, which could mean digging an excess of 8 in. into the soil. In open areas, multiflora rose grows as isolated plants, but it also grows in dense, impenetrable thickets in partially shaded areas and on sloping sites. Individual plants may live indefinitely, and can tolerate a wide range of soil conditions, although they perform best in undisturbed areas.

Figure 25. Flowering Multiflora Rose.

Figure 26. Multiflora Rose at edge of forest.

<u>Reproductive Biology</u>: Multiflora rose flowers from late May to June and is pollinated by insects. Flowers are typically white to whitish-pink, with five flowers per panicle. Once pollinated, fruits become bright red hips that contain seven to eight seeds. The potential seed output for a single plant is 500,000 seeds annually, with a single winter season being sufficient to break dormancy (Amrine 2002). Seeds tend to fall close to the parent plant, but can also be dispersed by birds and mammals. In the soil, seeds can stay viable for 10 to 20 years. Multiflora rose also reproduces asexually by suckering and layering once the tips of the canes grow long enough to touch the soil.

Origin and Distribution: Multiflora rose was introduced to the United States in the late 1800s as an ornamental plant, but in the mid 1900s it was used for conservation benefits as a "living fence" (Doll 2006). West Virginia alone planted more than 14 million plants during that time. The total area of infestation is estimated to be over 45 million acres across the eastern United States, and 38 states have reported its presence. Multiflora rose is best adapted to undisturbed areas, such as roadsides, old fields, pastures, fence rows, right-of-ways, stream banks, recreational lands, and forest edges. It does particularly well on steep hillsides and is most productive in sunny areas with well-drained soils (Munger 2002). It is moderately winter hardy, but its northern distribution is limited by tolerance to extreme cold temperatures. Multiflora rose grows as isolated plants in open areas, but in sloping sites or shading areas, it grows as dense thickets.

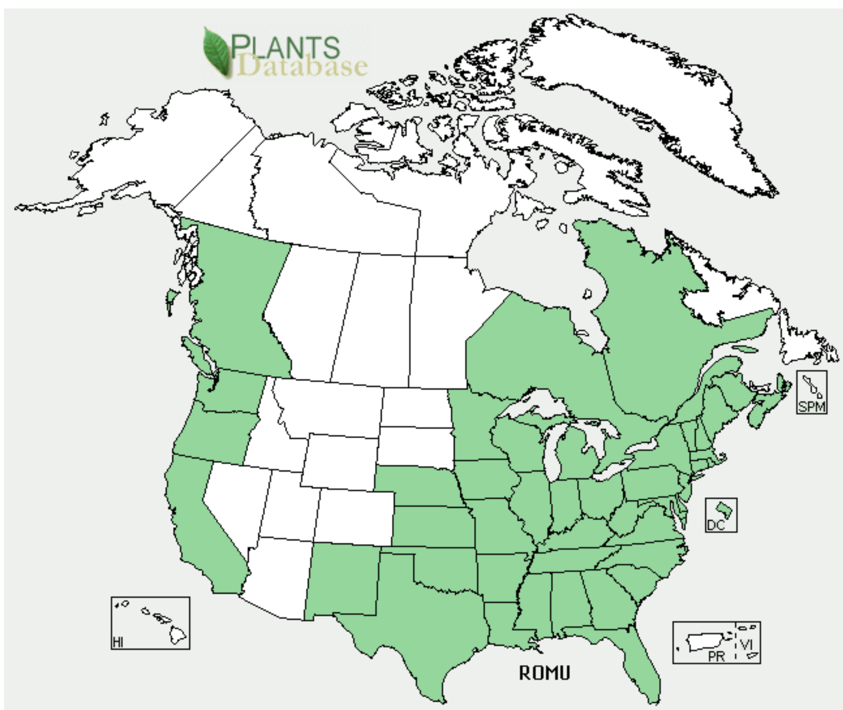

Figure 27. Distribution of Multiflora Rose.

Probability of Future Expansion: The plant's tough woody root system and its prolific seed production greatly contribute to its spread, and increased spread will only continue if traffic of seeds via wildlife or through anthropogenic vectors is not monitored. The roots can cause serious eradication problems, and the long-lived seeds can remain dormant for many years before germinating, which will only allow re-infestation. The plant's ability to germinate and grow in a variety of soil types means that its spread is typically independent of topographical constraints, excluding northern limitations due to winter temperatures.

Control Technologies: Multiflora rose can be controlled with herbicides, grazing, mowing, and biological control agents. A number of herbicides are effective, including dicamba (Banvel), fosamine (Krenite), glyphosate (Roundup), imazapyr (Arsenal), metsulfuron (Escort), tebuthiuron (Spike), and 2,4-D + triclopyr (Crossbow). Dicamba can be applied as either a foliar application after full leaf out in early spring or as a basal bark treatment when the plant is dormant (late December to early April). Basal bark treatments can be more effective than foliar applications; however, do not apply when snow or water may interfere with proper application. Fosamine should be applied as a foliar spray during July through September. Apply glyphosate as a foliar spray when plants are fully leafed-out during the bud to bloom stage; later applications can be made at 30-day intervals. Glyphosate treatments can provide near-complete, season-long control. Imazapyr and metsulfuron can also provide near-complete, season-long control when applied as a foliar spray from early spring through late summer. Tebuthiuron can be applied to the soil at any time when soils are not saturated or frozen, but rainfall is required to activate the pellets. 2,4-D + triclopyr can either be applied as a foliar spray when plants are actively growing or as a basal bark treatment when plants are dormant. If using a thinline basal bark treatment, apply early spring to early summer (Lingenfelter and Curran 1995; Loux et al. 2005; Szafoni 1990). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Grazing by sheep and goats can also be used to control multiflora rose. Goats may be more effective because they are not discouraged by the thorns of multiflora rose and can debark woody shrubs. Grazing should occur for two or more seasons to provide acceptable control (Lingenfelter and Curran 1995). Mowing three to six times per season for more than a year can also control multiflora rose, but can be labor intensive and expensive (Lingenfelter and Curran 1995).

The viral pathogen rose rosette disease (RDD) has been shown to kill multiflora rose within 2–5 years after infection. The RDD virus is spread by mites or by grafting infected shoots onto healthy plants (Epstein et al. 1997). Three insects have also been identified as causing injury to multiflora rose. The rose seed chalcid (*Megastigmus aculeatud* var. *nigroflavus*) destroys seeds, the tortricid hip borer (*Grapolita packerdi*) consumes floral parts, and the raspberry cane borer (*Oberea bimaculata*) kills the stems. None of these insects occur in large enough numbers to greatly impact multiflora rose populations and further research is needed to fully develop these biological control agents (Hindal and Wong 1988).

Direct and Indirect Military Impacts: Multiflora rose encroachment is considered a threat to the loss training land. (DA 2007). The major impact to the mission from the presence of multiflora rose on an installation is to dismounted troop movements. Because the plant grows in large, dense thickets, possesses large recurved thorns, and can grow to heights of up to 6m, movement through a stand of multiflora rose is very difficult. The thorns can rip both flesh and clothing as troops navigate through an area infested with multiflora rose. In addition, the long and extremely stout canes of multiflora rose can wrap around parts of vehicles, such as drive shafts, brake lines, and wheels, which could result in damage to vehicles.

Installations (22): Fort Lewis*, Fort Bliss*, Fort Hunter Liggett*, Fort Irwin*, Fort Riley*, Fort Sill*, Fort Leonard Wood *, Fort Chaffee*, Fort Polk, Fort McCoy*, Atterbury Reserve Training Area*, Fort Knox, Fort Campbell, Fort Rucker*, Fort Benning*, Fort Stewart*, Fort Jackson*, Fort A.P. Hill, Fort Pickett*, Fort Bragg*, Fort Indiantown Gap, Fort Drum*.

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Photo credits:

Figure 25: Leslie J. Mehrhoff, University of Connecticut (Bugwood.org)

Figure 26: James H. Miller, USDA Forest Service (Bugwood.org)

Figure 27: USDA Plants Database (plants.usda.gov)

Purple Loosestrife (*Lythrum Salicaria*): Purple loosestrife is an herbaceous, semi-woody, perennial, that is considered invasive in the United States and Canada. It is a self-incompatible, tristylous herb that grows best on neutral to acidic soils and typically performs better with disturbance (Ketterer and Abrahamson 2006). Established plants are up to 2-m tall with 30-50 stems forming large crowns, supported by a very strong rootstock. Flowers are at the top of the spike, which is usually 0.5–1 m long. Petals are usually magenta, but can range from white to pink to even deep purple and red (Mullin 1998). Purple loosestrife usually forms large, monotypic stands that displace many other native species and cause changes in organic matter distribution (Malecki et al. 1993; Fickbohm and Zhu 2006).

Figure 28. Purple Loosestrife flowers.

Figure 29. Purple Loosestrife in wetland habitat.

Reproductive Biology: The plant reproduces almost exclusively by seed, and a single mature plant can produce more than 2.5 million seeds annually. Flowering typically occurs in July-August for 6 weeks, with seeds maturing after an additional 6-8 weeks (Olsson and Agren 2002). Seeds have a 90% germination rate and can remain in the seed bank for 3 years (Ketterer and Abrahamson 2006; Chun et. al. 2007).

Origin and Distribution: Native to Eurasia, the first recorded instance of purple loosestrife in the United States was in 1814. It was originally introduced through ship ballast and wildlife, but was also cultivated for its ornamental and pharmacological values (Farnsworth and Ellis 2001). It can be found in 43 of the 48 contiguous states, and can be found mostly in wetlands, lakes, riversides, fens, and seashores (Anderson et al. 2006; Olsson and Angren 2002). Growth is reported at a rate of 115,000 hectares per year, and 24 states have listed purple loosestrife as a pest (Shadel and Molofsky 2002).

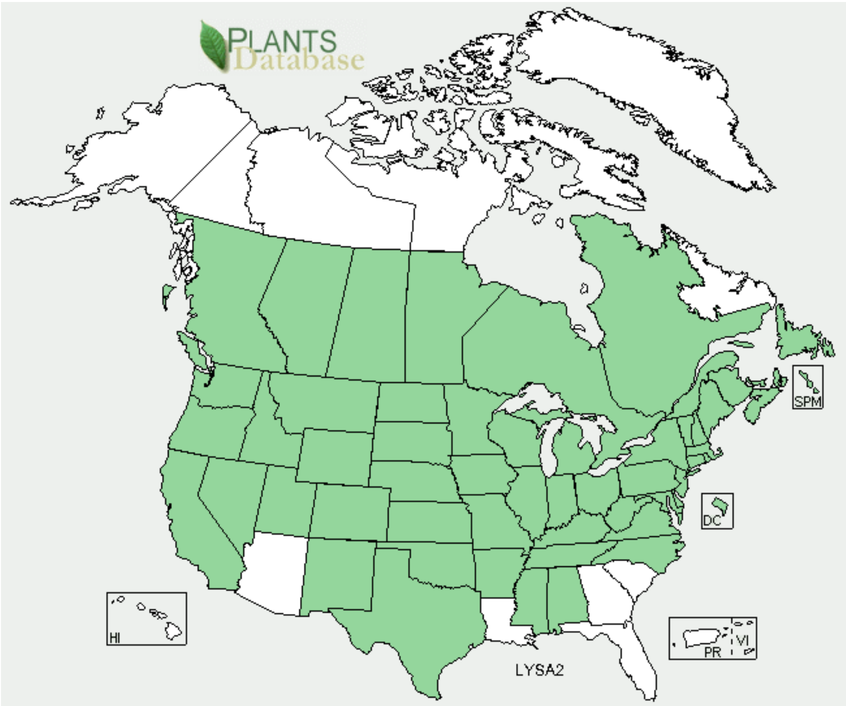

Figure 30 - Distribution of Purple Loosestrife

<u>Probability of Future Expansion</u>: Purpose loosestrife control efforts have been met with limited success due to the large and long-lived seed bank (Chun et al. 2007). The plant’s tolerance of a wide variety of soil-nutrient conditions also contributes to further expansion, as the plant is also able to outcompete many native plants (Denoth and Myers 2007). Additionally, purple loosestrife consumes significantly more water than native plants, alters nitrogen transformations, and dedicates more of its resources toward vegetative growth, resulting in a stronger competitor and a tougher plant (Fickbohm and Zhu 2006). The plant’s spread over large geographic areas is possible through water routes and vehicle traffic (Malecki et al. 1993). Current estimates are that purple loosestrife affects over 190,000 hectares of marshes, wetlands, riparian zones, and pastures in the United States every year (Minnesota Sea Grant 2009). Varieties of purple loosestrife are also still commercially available in some locations, and even if the plant is advertised

as a sterile hybrid, studies have shown that the hybrids readily cross with wild varieties, and produce viable seed (Mullin 1998).

Control Technologies: Purple loosestrife may be controlled by herbicides, hand pulling, and biological control agents. Herbicides that have shown activity on purple loosestrife include glyphosate (Rodeo), 2,4-D, triclopyr (Renovate 3), and imazapyr (Habitat). Glyphosate should be applied from the early to late bloom stage, 2,4-D when plants are actively growing or until early bloom, triclopyr from bud to mid-bloom and imazapyr to actively growing plants. Glyphosate and imazapyr are nonselective herbicides; therefore, spray to non-target vegetation should be avoided (Mullins 1998; Knezevic et al. 2004). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Hand pulling can be effective on 1–2 year old plants because of their small root systems. Hand-pulled plant material should be dried or burned and soil disturbance should be kept to a minimum to avoid re-infestation (Mullin 1998). *Galerucella pusilla*, *G. calmariensis, Hylobius transversovittatus*, and *Nanophyes marmoratus* are four biological control agents that have been released. *G. pusilla* and *calmariensis* are beetles that feed on foliage and buds, reducing seed production and stunting plant growth. *H. transversovittatus* larvae feed on roots, whereas adults feed on the foliage, and *N. marmoratus* feeds on shoot tips, flower buds, and immature seed capsules (Blossey and Schroeder 1995; Blossey et al. 1994a, b). Biological control agents will not eradicate purple loosestrife, but can suppress loosestrife populations. If eradication is the ultimate goal, then biological control agents should not be used alone (Mullin 1998).

Direct and Indirect Military Impacts: Establishment of purple loosestrife has impeded the water flow in irrigation systems, and large monotypic stands jeopardize TES wetland species (Malecki et al. 1993). Also attributable to invasion of purple loosestrife is the reduction of wetland pasture and hay meadows, as livestock find the plant to be less palatable than native grasses (Klips and Penalosa 2003).

Installations (15): Fort Lewis, Fort Hunter Liggett, Fort Irwin*, Fort Carson*, Fort Bliss*, Fort Riley*, Fort Sill*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee*, Fort McCoy*, Fort Knox, Fort Campbell*, Atterbury Reserve Training Area*.

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Photo credits:

Figure 28: Norman E. Rees, USDA Agricultural Research Science (Bugwood.org)

Figure 29: John D. Byrd, Mississippi State University (Bugwood.org)

Figure 30: USDA Plants Database (plants.usda.gov)

Russian Knapweed (*Acroptilon repens*): Russian knapweed is a long-lived, highly aggressive, creeping perennial weed that infests thousands of acres of rangeland and pastures in the United States (Benz 1999; Goslee 2003). Mature forbs are fibrous, and the coarse spines and stems limit access to the plant, preventing grazing (Roche and Roche 1988). Stems originate from a basal rosette of leaves, which can be 2-4 in. long. The leaves are oblong, with pink and purple flowers that gradually turn straw colored upon maturity and seed dispersal. The extensive root system includes a taproot that can grow up to 6 m into the ground (Grant 2003). Russian knapweed typically forms a monoculture, and has been known to exude allelopathic compounds (Benz 1999).

Figure 31. Flowers of Russian Knapweed

Figure 32. Russian Knapweed along a roadside.

Reproductive Biology: Russian knapweed growth begins in the spring as soon as the ground does not freeze. Russian knapweed produces by seed and by its adventitious creeping root system (Grant et al. 2003). The plants bolt in May and June, with flowering occurring in July and continuing until September or October. Russian knapweed manages resources well and will produce seeds only when it is advantageous to do so. A single plant is capable of producing up to 1,200 seeds that are capable of germinating for up to 3 years (Maddox et al. 1985). Seed dispersal most often occurs via infested hay. Both the shoots and the roots of Russian knapweed produce allelopathic compounds (Grant et al. 2003).

Origin and Distribution: Russian knapweed is native to southern Russia and Asia and was introduced to the United States in the early 1900s, with the earliest record between 1910 and 1914 in California. It was most probably introduced as a contaminant of alfalfa seed and occasionally of sugarbeet seed (Maddox 1985).

Russian knapweed did not significantly spread until about 1928, however, most likely due to its widespread distribution in infested hay. Invasion occurs most often on soils with high clay content, low June precipitation, low elevation, and high December temperatures (Goslee et al. 2003). Significant infestations of Russian knapweed have been reported in 21 states, and this plant is chiefly a problem in the western region of the United States, especially in the Rocky Mountain areas and further west (Laufenberg et al. 2005). Russian knapweed thrives in disturbed areas and spreads along railroads and roadsides (Roche and Roche 1988). Current estimates indicate that Russian knapweed can be found on more than 1 million acres in the contiguous United States (Zouhar 2001).

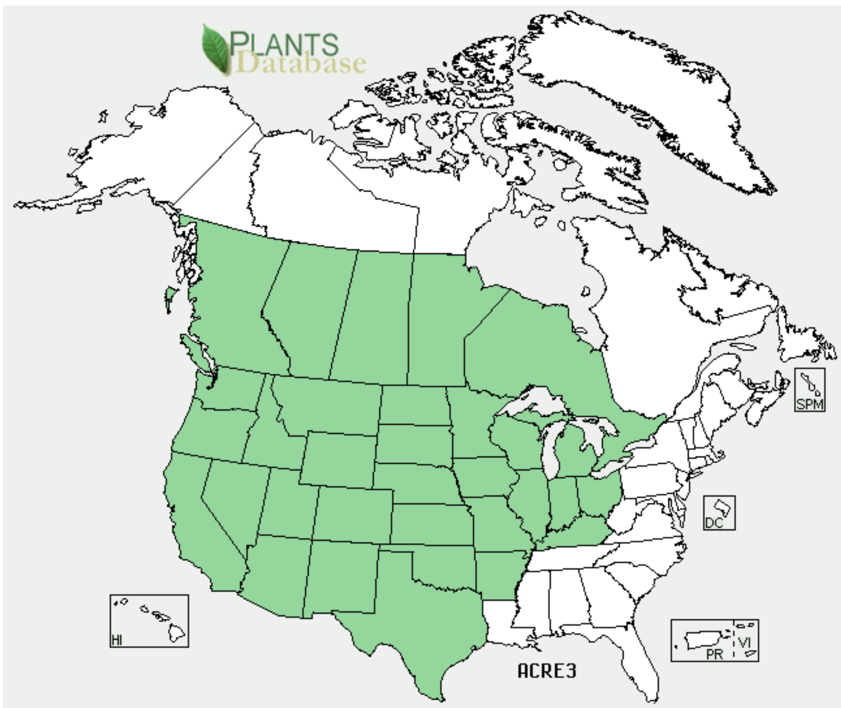

Figure 33. Distribution of Russian Knapweed.

Probability of Future Expansion: Russian knapweed causes a large shift in species composition on a site, and further expansion is encouraged when monocultures of the weed are formed – thereby removing competing plants (Laufenberg 2005). Russian knapweed can adapt very quickly to disturbances and environmental changes, and the production of allelopathic compounds also gives the weed a germination and growth advantage over native grasses. The plant’s extensive root system is capable of reaching water when other plants cannot, and can reproduce vegetatively when it is not advantageous for the plant to produce seeds (Morris et al. 2006). Infestations of Russian knapweed have been reported to survive over 75 years. Monocultures are poorly controlled by grazing as the plant is unpalatable to livestock, and in large amounts can be toxic to horses (Zouhar 2001). The current estimated rate of expansion is as high as 11% in some western states (Whitson 1999).

Control Technologies: Russian knapweed is difficult to control and currently the best option is herbicides. Herbicides that are effective include clopyralid (Transline), clopyralid + 2,4-D (Curtail), picloram (Tordon), or dicamba (Vanquish). Clopyralid should be applied from the bud to mid-flower stage of growth or to fall regrowth, whereas clopyralid + 2,4-D should be applied during the bud to bloom stage. Apply picloram or dicamba to actively growing plants in the spring. Picloram can also be applied to fall regrowth (Beck et al. 2004). Refer to herbicide labels for specific instructions regarding rates, adjuvants, application techniques, and use restrictions.

Currently, the only biological control agent that has been released is the gall-inducing nematode, *Subanguina picridis*. The nematode has been shown to reduce flowering and biomass, but infections have been inconsistent. In addition, the nematode does not move and therefore requires large-scale propagation and redistribution. Several other agents currently being evaluated include two flower gall flies *Urophora kasachstanica* and *U. xanthippe* (Coombs et al. 2004). Goats will graze on Russian knapweed; however, long-term impacts are unknown (Jacobs and Denny 2006).

Direct and Indirect Military Impacts: Russian knapweed can contribute to soil erosion and alteration of species composition in both plant and small mammal populations (Goslee et al. 2003; Maddox et al. 1985). These factors can be of concern directly to maneuver training and similar land use as well as loss of TES habitats.

Installations (15): Fort Lewis*, Fort Hunter Liggett, Fort Irwin, Fort Bliss, Fort Carson¸ Fort Riley, Fort Sill*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee*, Fort McCoy*, Fort Knox, Fort Campbell*, Atterbury Reserve Training Area*.

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Figure 31 and 32: Steve Dewey, Utah State University (Bugwood.org)

Figure 33: USDA Plants Database (plants.usda.gov)

Russian Thistle (*Salsola tragus*): Russian thistle is an annual herb with erect and profusely stemmed branches with red striations, and alternate leaves. Upon reaching maturity, the plant resembles a spherical bush between 10- and 100-cm tall (Shillinger and Young 2000). The root system of Russian thistle can grow to 5 m in diameter, or 2 m in length, and extracts water more successfully than native species (Schillinger and Young 2000). The flowers do not appear to have petals but rather wing-like appendages that resemble petals. At maturity the plant is dislodged from the ground and moved by the wind, scattering seeds (Mosyakin 1996).

Figure 34. Russian Thistle flowers.

Figure 35. Russian Thistle herb.

Reproductive Biology: Russian thistle produces primarily by seeds. The plants grow initially in March and April and are flowering by June. Beginning in August, seeds are produced (Schillinger 2007). Seeds are grayish brown and have a thin seed coat. Seeds remain viable for up to 1 year, and need only loose soil and temperatures between 28 and 110°F to germinate (Mosyakin 1996; Young et al. 2008). Large Russian thistle plants are capable of producing up to 100,000 seeds (Young 2008). Russian thistle is self-fertile but can also out-cross and is pollinated via wind. Upon reaching maturity, Russian thistle breaks off at the stem base and is propelled by the wind, scattering seeds as they tumble. Transport of the weed can also occur through transport through contaminated animal bedding in rail cars, contamination of agricultural seed, and by becoming attached to vehicles or passing animals and humans (Ayres et al. 2009).

Origin and Distribution: Russian thistle is native to Eurasia, from eastern Russian to southeast Siberia and northeast China to northern Africa. The plant is an invasive species in all 48 of the contiguous United States (Ayres 2009). It was introduced as a contaminant of flax seed, and it typically invades sandy soils in disturbed areas. It can be found in waste areas, roadsides, fields, and has infested an estimated 41 million ha throughout the western United States (Schillinger 2007; Young 2008).

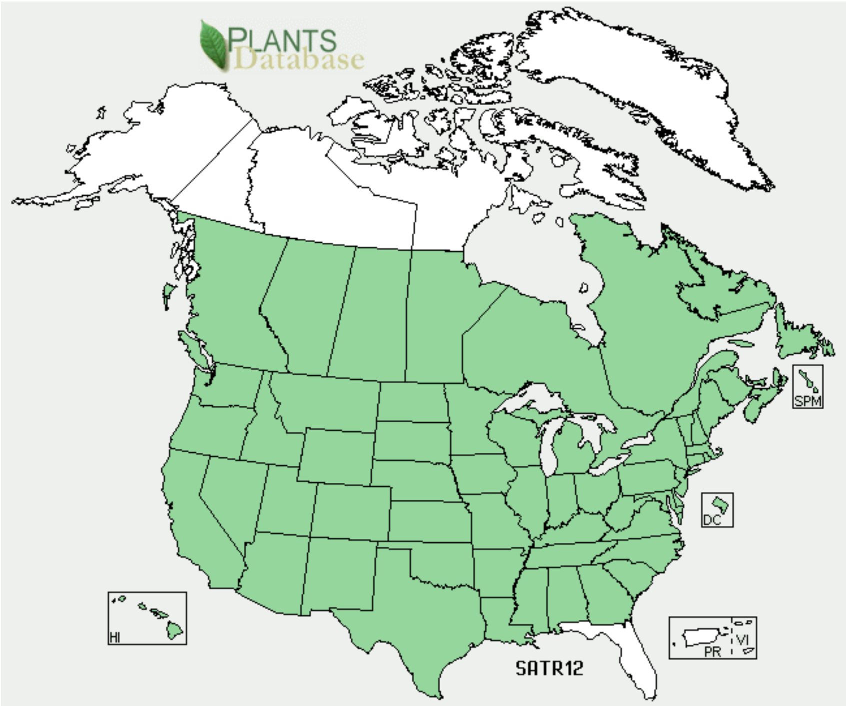

Figure 36 - Distribution of Russian Thistle

Probability of Future Expansion: Russian thistle seed is extensively dispersed by the tumbling of the weed. Additionally, with such prolific seed production and a wide range of temperatures that are conducive to germination, Russian thistle can grow in a variety of conditions. The extensive root system can reach water where other native grasses cannot, and Russian thistle is self-fertile. Russian thistle aids in spreading fires, and colonizes a burn site within 1-3 years, becoming completely dominant after only 2 years (Howard 1992). Varieties of the plant are still available and used for hay and silage, as well as holiday ornamentals and disturbed-site rehabilitation (Bare 1979).

Control Technologies: Russian thistle can be controlled with a number of herbicides. Preemergent herbicides should be applied to soil before weeds appear to obtain best activity. These herbicides include bromacil (Hyvar), chlorsulfuron (Telar), hexazinone (Velpar), imazapyr (Arsenal), simazine (Princep), and sulfometuron (Oust). If applying a postemergent herbicide, treat immature plants before the plant produces spines and becomes hardened. Treating early will prevent seed production and thus reduce the spread of Russian thistle. Post-emergent herbicides include dicamba (Banvel), glufosinate (Finale), glyphosate (Roundup), and paraquat (Gramoxone). If new seedlings emerge in the treated area due to irrigation or rain occurrence following application, additional herbicide applications will be necessary (CDFA 2009; Orloff et al. 2008). Some populations of Russian thistle have become resistant to acetolactate synthase (ALS) inhibiting herbicides such as sulfometuron and chlorsulfuron (Peterson 1999; Orloff et al. 2008). Rotate between herbicides of various modes of action or use an integrated management strategy to prevent further herbicide resistance (CDFA 2009). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Mowing can be used to control very young plants but should never be done after seed set. Older plants will recover after mowing by forming axial branches below the cut level (CDFA 2009). Several biological control agents have been released in an effort to control Russian thistle. *Coleophora parthenica* is a moth that feeds inside the stem, causing minimal damage to the growth of the plant; therefore, it is considered an ineffective control agent. *C. klimeschiella* is a moth that feeds on leaves. Young plants usually die when heavily infested with *C. klimeschiella*, but the impact on older plants is unknown. Although *C. klimeschiella* does impact young plants, it is considered an ineffective control agent because the moth is heavily attacked by native predators and parasitoids, preventing it from populating to densities required to control Russian thistle (Coombs et al. 2004). The most recent and promising agent is the blister mite, *Aceria salsolae*, which feeds on meristematic tissues resulting in stunted plant growth. The mite has been shown to reduce the size of Russian thistle plants by 66% under artificial conditions (Smith 2005). Several other biological control agents are also under investigation and include stem-boring and seed feeding caterpillars as well as two weevils (Orloff et al. 2008).

<u>Direct and Indirect Military Impacts</u>: Russian thistle is a road hazard while tumbling, with the potential to startle drivers and cause traffic accidents. The tumbling weeds can become caught against fences and other obstructions, potentially causing damage to military range equipment and other property.

Biodiversity is also threatened, as Russian thistle exploits resources more effectively than native species.

Installations (25): Atterbury Reserve Training Area*, Fort A.P. Hill*, Fort Benning*, Fort Bliss, Fort Bragg*, Fort Campbell*, Fort Carson, Fort Chaffee*, Fort Drum, Fort Hood*, Fort Hunter Liggett, Fort Indiantown Gap, Fort Irwin, Fort Jackson*, Fort Knox*, Fort Leonard Wood*, Fort Lewis, Fort McCoy*, Fort Pickett*, Fort Polk*, Fort Riley, Fort Rucker*, Fort Sam Houston*, Fort Sill, Fort Stewart*.

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Figures 34 and 35: Forest and Kim Starr, U.S. Geological Survey (Bugwood.org)

Figure 36: USDA Plants Database (plants.usda.gov)

Scotch Broom (*Cytisus scoparius*): Scotch broom is a perennial, nitrogen fixing, leguminous shrub that is capable of forming extremely dense stands. The plant grows between 3- and 9-ft tall and is tolerant of most soil conditions. However, Scotch broom grows best in dry sandy soils in full sunlight. Flowers are yellow and pea-like and form in axillary clusters, with 1-2 flowers per cluster (Zouhar 2005). The branches are long and slender with small leaves. The plant is capable of rapidly forming dense and monotypic stands and outcompeting native vegetation (Prevosto et al. 2006). Given its ability to dominate communities, it does have the potential to degrade TES habitat and/or contribute to long term soil stability (USDA NRCS 2004; Parker et al. 1998; Mountjoy 1979).

Figure 37. Flowering Scotch Broom

Figure 38. Scotch Broom shrub.

<u>Reproductive Biology</u>: Scotch broom is a prolific seeder with a single plant able to produce 60 seed pods, each containing 6-8 seeds, each year. The seeds can remain viable in the seed bank for 5–80 years. Primary reproduction occurs sexually and may be facilitated by allelopathic strategies based on the plants ability to produce an alkaloid substance. Flowers will trip open when pollinated, and the plant has an almost obligatory relationship with bees in the western states (Parker 1997). Dispersal occurs in a catapult fashion as the seed pods dry and snap apart. Seeds may also be dispersed along waterways and by vehicle transport (Zouhar 2005).

<u>Origin and Distribution</u>: Scotch broom is native to Europe and can be found from Ireland to Ukraine, and Spain to Sweden (Simpson et al. 2005). The plant was first introduced as an ornamental by settlers of the Pacific Northwest, and the first recorded introduction was collected from Seattle in 1888 (Parker 1997). It was reintroduced later as erosion control and stabilization by the Soil Conservation Service of the U.S.

Department of Agriculture (Caldwell 2006; Bossard and Rejmanek 1994). Scotch broom has been naturalized in California, and it has invaded areas such as open grasslands and fir forests. Additionally, the plant occurs often in disturbed urban areas (Haubensak and Parker 2004).

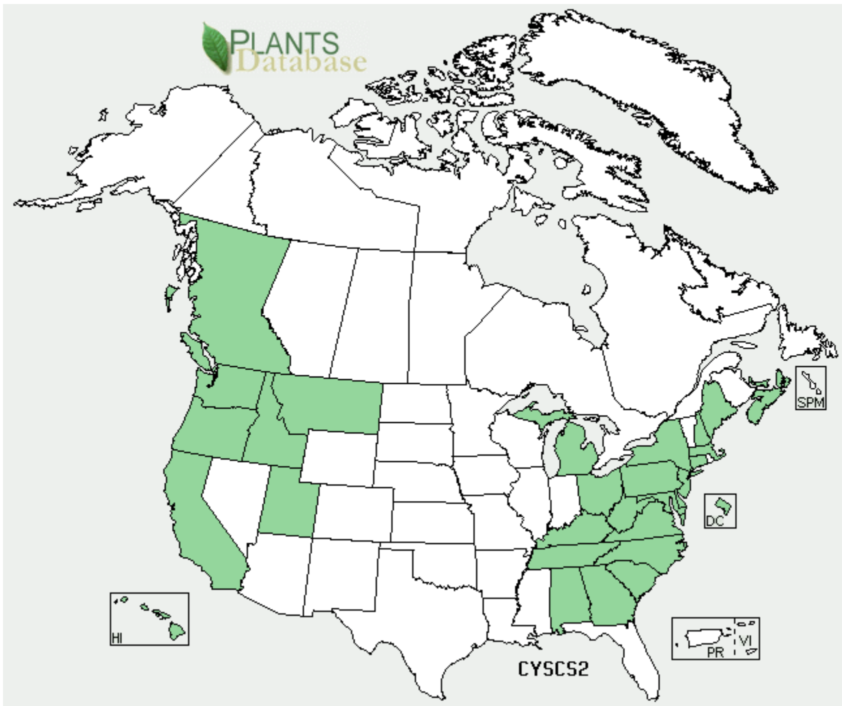

Figure 39 - Distribution of Scotch Broom

Probability of Future Expansion: Localized spread is possible through many vectors including a pod-dispersal mechanism that can eject seed up to 20 ft from the plant, ants, livestock, wildlife, and water (Parker et al. 1998). The plant can be spread across larger geographical areas via human vectors such as cars, heavy equipment, and foot traffic. Characteristics of Scotch broom that enable rapid growth include a long-lasting seed bank, large amounts of early seed production, rapid growth, and an ability to recolonize an area after disturbances (Prevosto et al. 2006). Scotch broom alters fuel structure and fire regimes, and studies suggest that it is well adapted to postfire germination (Zouhar 2005). The allelopathic abilities and the plant's ability to fix nitrogen could reduce native plant growth because of the unavailability of nutrients (Haubensak and Parker 2004; Fogarty and Facelli 1999).

Control Technologies: Scotch broom can be controlled with herbicides, hand pulling, cutting, and grazing. Herbicides shown to be effective include 2,4-D, triclopyr (Garlon), 2,4-D + triclopyr (Crossbow), fluroxypyr (Starane), hexazinone (Velpar), picloram (Tordon) and atrazine (Atrazine) (Hoshovsky 1991; Peterson and Prasad 1988). Atrazine and hexazinone are effective on germinating or very young seedlings, but not on established broom plants (Hoshovsky 1991). All other herbicides should be applied when Scotch broom is in full leaf. Results have been best when plants are in the seed head stage of growth during late summer or early fall. Herbicides should not be sprayed when Scotch broom is in full flower (Matthews 1960). When using any of the above=listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Hand pulling young shrubs can be effective if the entire root system is removed. This should be done before Scotch broom produces seeds and can disturb the soil (Hoshovsky 1991). Cutting can also be effective but must be done more than once to exhaust underground food reserves or during times when the plant is most stressed (Hoshovsky 1991; Ussery and Krannitz 1998). Scotch broom is consumed by both sheep and goats; however, goats are more effective because they strip bark from the stems. Seedlings can appear after mature shrubs die; therefore, good grazing management must be employed to prevent re-establishment (Holst et al. 2004). Insect biological control agents that have been released have not been able to reduce established populations of Scotch broom. Several new potential control agents have been shown to feed on Scotch broom, but research is still needed to develop these agents (Hoshovsky 1991).

<u>Direct and Indirect Military Impacts</u>: Given its ability to dominate plant communities, Scotch broom does provide for the potential to degrade TES habitat and/or contribute to long-term soil stability (USDA NRCS 2001; Parker et al. 1998; Mountjoy 1979). All of the impacts have the potential to reduce training capacity, although (unlike the lespedezas and knapweeds) Scotch broom is more niche specific, which translates to less installation lands potentially affected. Where Scotch broom is found, however, it forms dense stands that have significant impacts on native vegetation and have been reported to provide habitat for feral pigs (Caldwell 2006; Simpson et al. 2005).

<u>Installations (14)</u>: Fort Lewis, Fort Hunter Liggett, Fort Irwin*, Fort Knox*, Fort Campbell*, Fort Rucker*, Fort Drum*, Fort Benning*, Fort Stewart*, Fort Jackson*, Fort Bragg*, Fort A.P. Hill*, Fort Pickett, Fort Indiantown Gap*.

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Photo credits:

Figure 37: Utah State Univ. Archive, Utah State University (Bugwood.org)

Figure 38: Steve Dewey, Utah State University (Bugwood.org)

Figure 39: USDA Plants Database (plants.usda.gov)

Sericea Lespedeza (*Lespedeza cuneata*): Sericea lespedeza, also known as Chinese lespedeza, is a perennial, upright, semi-woody, nitrogen-fixing forb reaching 3–6 ft in height with slender stems and small whitish-yellow flowers. It is deep-rooted and well adapted to acidic soils, making it well known throughout the Southern and Midwestern United States as a highly invasive plant of grasslands (Brandon et al. 2004). Sericea lespedeza is the only widely adapted, perennial, warm-season plant legume with seed still commercially available for forage, wildlife habitat improvement, and conservation in the southeastern regions of the Nation (Peterson et al. 2003). This is due to its performing very well in harsh environments where other plants cannot become established (Brandon et al. 2004).

Figure 40. Flower of Sericea Lespedeza.

Figure 41. Sericea Lespedeza in an open field.

Reproductive Biology: Sericea lespedeza reproduces primarily via seed and is capable of producing as much as 670 kg of seed/hectare annually (Smith and Knapp 2001). The growth rate was reported to be insensitive to changes in the seed bank; seeds are also capable of remaining viable in soil for more than 30 years. Due to a large tap root (up to 1 m), the plant can survive extreme drought conditions. Seeds are spread via many methods, including wildlife, water, farm equipment, and transportation of infested hay or grasses (Schutzenhofer and Knight 2007). In areas with little management, the plant spreads mostly by wildlife such as birds or deer. It is thought that sericea lespedeza has the potential to be allelopathic due to the tannins in the foliage, but this has yet to be studied extensively.

Origin and Distribution: Sericea lespedeza is native to Asia and was originally introduced to the United States from Japan in 1896. It has been used as a forage crop and for soil conservation, being planted in areas such as strip mines, road banks, and other areas that have been significantly disturbed. It was used heavily in the 1930s and 1940s for conservation, and has since been spread to over 30 states (Drake et al. 2003). It slowly invades less managed pastures and grasslands after being planted in an area (Smith and Knapp 2001).

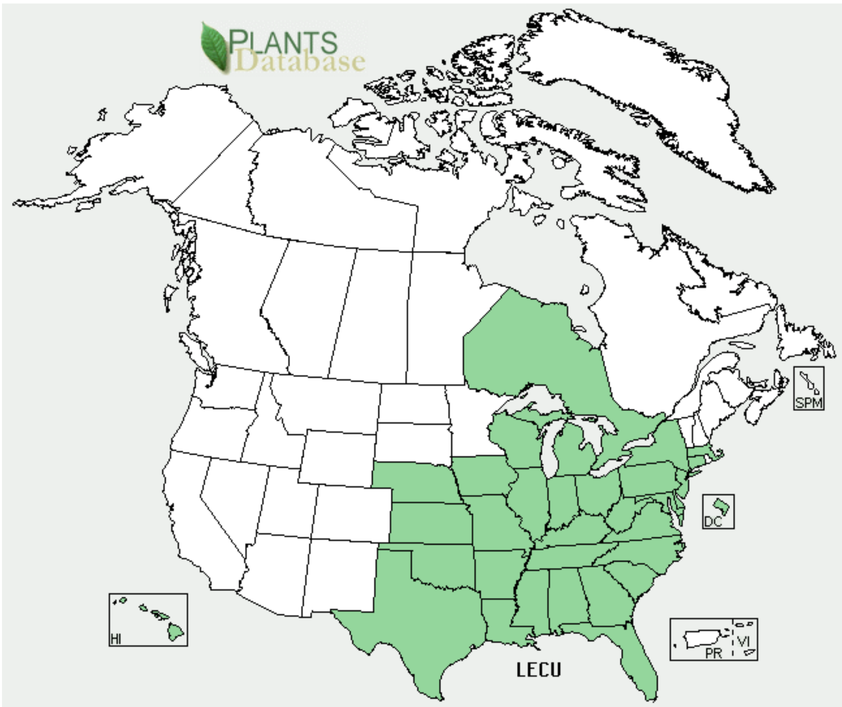

Figure 42. Distribution of Sericea Lespedeza.

Probability of Future Expansion: Spread of this plant seems inevitable because of its numerous introductions and re-introductions, and because of the many vectors through which it is spread. The long-lived seed bank contributes to the plant’s spread or to re-colonization of an area. Suspected allelopathic tendencies also contribute to its spread and can reduce germination of big bluestem, indiangrass, and Kentucky bluegrass (Drake et al. 2003). Additionally, the plant’s response to human control mechanisms is poorly understood; as such, the plant could benefit from mowing or burning (Brandon et al. 2004). The plant can be spread across larger geographical areas via human vectors such as cars, heavy equipment, and foot traffic, and is still commercially available as an ornamental, forage crop, or for habitat improvement.

Control Technologies: Sericea lespedeza can be controlled with herbicides, mowing, or an integrated strategy. Herbicides that are effective include triclopyr (Garlon), fluroxypyr (Starane), glyphosate (Roundup), and metsulfuron (Escort). Triclopyr and fluroxypyr can be applied at the simple or branched-stem stage of growth, but applying at the branched-stem stage can provide the most effective, consistent, and long-term control (Koger et al. 2002). Glyphosate should be applied during flowering which typically occurs from early August to mid-September (Yonce and Skroch 1989). Metsulfuron can also be applied at the flowering stage of growth; however, control has been reported to be variable (Altom et al. 1992; Koger et al. 2002). Due to the prolific seed production and extended seed dormancy of sericea lespedeza, repeat applications may be necessary to control new seedlings. Refer to herbicide labels for information regarding application rates, adjuvants, application techniques, and use restrictions.

Mowing plants multiple times a year for 2–3 years can reduce vigor and control further spread. Sericea lespedeza should be mowed each time plants reach 12–18 in. in height, especially late in the growing season when plants are transferring carbohydrates to the roots (Stevens 2002; Vermeire et al. 1998). Grazing by goats may be used to reduce seed production as long as plants are kept below 3–4 in., but grazing will not prevent the plant from spreading (Ohlenbusch and Bidwell 2007). Burning in the spring can increase seed germination and stimulate resprouting and cannot control sericea lespedeza if used alone (Stevens 2002). Integrated management techniques such as mowing, grazing, and burning with herbicide can provide effective control of sericea lespedeza. Examples include mowing in June or July followed by herbicide application in July or September or burning (to encourage seed germination) followed by herbicide application (Vermeire et al. 1998). Contact local extension offices for the latest integrated management strategies.

Direct and Indirect Military Impacts: The plant’s ability to reduce diversity, exclude native species, and form monocultures directly results in habitat degradation, and potentially affects sensitive or endangered species. Sericea lespedeza has been proven to reduce grass production in native tallgrass prairie by 92% (Schutzenhofer and Knight 2007). The species directly competes with at least one TES plant (*Rhuz michauxii*) on an installation (Personal Communication, 20 May 2008: Army O. Hayne, VAARNG-FM-E).

Installations (19): Fort Riley*, Fort Sill*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee, Fort Polk¸ Fort McCoy*, Atterbury Reserve Training Area*, Fort Knox, Fort Campbell, Fort Rucker*, Fort Benning, Fort Stewart*, Fort Jackson, Fort Bragg, Fort A.P. Hill, Fort Pickett, Fort Drum*.

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Photo credits:

Figure 40: Dan Tenaglia, Missouriplants.com (Bugwood.org)

Figure 41: Chuck Bargeron, University of Georgia (Bugwood.org)

Figure 42: USDA Plants Database (plants.usda.gov)

Shrubby Lespedeza (*Lespedeza bicolor*): Shrubby lespedeza, also known as bushclover, is a perennial, multi-branched, leguminous shrub that is a very shade-tolerant and can grow from 3- to 10-ft tall (Vogel 1974). The plant has three-leaflet leaves (where the lower surface is lighter than the top surface), a strong rootcrown, and purple and white flowers that resemble pea-flowers. The stems are branched and arching and can reach up to 1 in. in diameter. Fine hairs cover the stems, which appear gray (Miller 2003). Shrubby lespedeza is nitrogen-fixing and can grow in a wide range of soils, from acidic, nutrient-poor soils to sandy soils. It has the ability to spread in areas of medium to dense under-story and can easily become dominant in disturbed areas (EFTC 2009).

Figure 43. Shrubby Lespedeza flower.

Figure 44. Sericea Lespedeza stand.

Reproductive Biology: Shrubby lespedeza reproduces by seed, but can also re-sprout from root crowns if the plant is damaged by mowing or burning (Miller 2003). The lifespan of the plant is relatively short, but growth is very rapid. Shrubby lespedeza flowers from June to September in 6-in. clusters that grow from the upper leaves. The seeds and fruits form from August to March. Seeds are small and black and are enclosed in gray pods with hair-like tips (Miller 2003; EFTC 2009). Seeds are dispersed by animals and by movement of infested hay and can remain viable for up to 20 years (Duke University 2005).

Origins and Distribution: Shrubby lespedeza was introduced to North America in the 1800s from Japan, and was originally planted for soil conservation, erosion control, and wildlife cover or forage (EFTC 2009; Haugen and Fitch 1955). Shrubby lespedeza can be found in fields, parks, forests, meadows, waterways, swamps, prairies, and rights-of-way (Miller 2003).

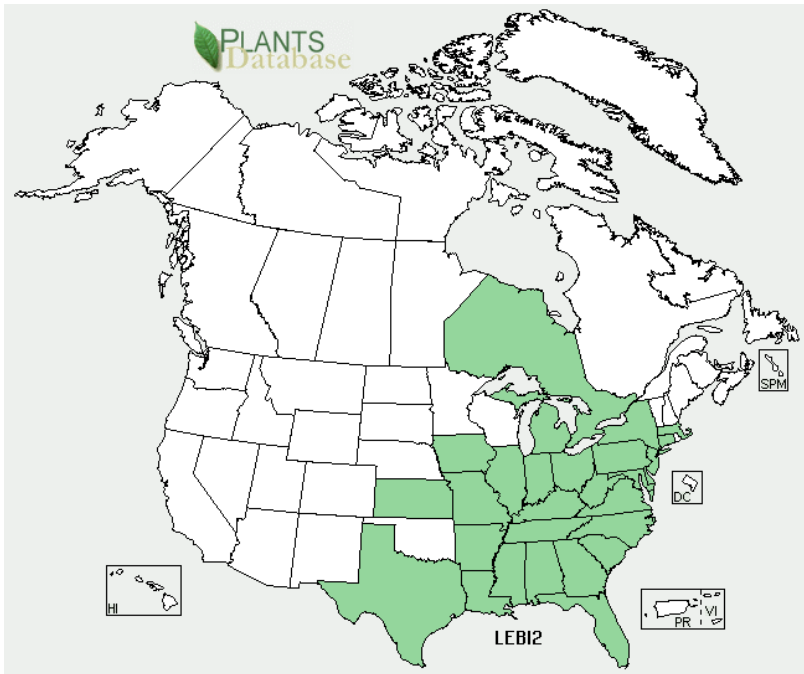

Figure 45. Distribution of Shrubby Lespedeza.

Probability of Future Expansion: Shrubby lespedeza is still being planted as “wildlife habitat”, especially for quail, for erosion control, and in some areas it is still thought desirable for ornamental use (Miller 2003). Additionally, seeds are easily transported by wildlife and human activities and can remain viable in the seed bank for long periods of time; perhaps 20 years (Vogel 1974). These factors suggest high potential for spread across both localized and larger scale geographic areas.

Control Technologies: Shrubby lespedeza can be controlled with the following herbicides: triclopyr (Garlon), metsulfuron (Escort), clopyralid (Transline), glyphosate (Roundup), or hexazinone (Velpar). All herbicides listed above should be applied between July and September. Mowing shrubby lespedeza 1–3 months before herbicide application can improve control (Miller 2003).

Prescribed burning enhances its spread. Mowing may retard growth but will not kill the plant. No biological controls are approved for the species. Some native insect species are beginning to adapt and use the plant; which may someday lead to future biological control methods (Duke University 2005).

Direct and Indirect Military Impacts: Due to its ability to dominate in near monoculture densities and exclude native species, it has the potential to degrade TES habitat. The species can also impede tree regeneration, thus impacting both maneuver lands and forested lands.

Installations (17): Fort Riley*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee, Fort Polk*, Atterbury Reserve Training Area*, Fort Knox*, Fort Campbell, Fort Rucker*, Fort Benning*, Fort Stewart, Fort Jackson*, Fort Bragg*, Fort A.P. Hill*, Fort Pickett, Fort Indiantown Gap*.

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Photo credits:

Figure 43: James H. Miller, USDA Forest Service (Bugwood.org)

Figure 44: Chris Evans, River to River CWMA (Bugwood.org)

Figure 45: USDA Plants Database, plants.usda.gov

Spotted Knapweed (*Centaurea maculosa*): Spotted knapweed is a biennial or short-lived perennial with branched stems growing to 4-5 feet in height. It is capable of living up to 9 years. The plant is a deeply tap-rooted, rosette-forming plant that prefers well-drained, light-textured soils. (Sheley et al. 1998) It can be found in open forests and prairies in a majority of the United States. While disturbance allows for more rapid invasion, spotted knapweed can also invade well-managed rangelands. It competes poorly with well-established grass populations in moist areas, but in seasonally dry areas its taproot allows access to water deeper in the soils than shallow rooted species. The species can invade an area and exist in small colonies for an extended period of time, then rapidly dominate an area when conditions are suitable. The plant exudes allelopathic substances that inhibit the establishment of competing plant species (Fletcher and Renney 1963).

Figure 46. Flower of Spotted Knapweed.

Figure 47. Spotted Knapweed in arid habitat.

<u>Reproductive Biology</u>: Spotted knapweed primarily reproduces by seed, although it can re-establish from root-crown buds after disturbance. Germination usually occurs in early spring or fall, depending on moisture availability. Seedlings develop into rosettes, which produce floral stems after overwintering. Continued growth occurs in June, flowering in July, and seed

dispersal in August (Story et al. 2001). The flower heads open after maturation, and movement of the stem (through wind, animal contact, or otherwise) propels the seeds up to 1 m from the parent plant. Seed production ranges from 5,000 to 40,000 seeds per square meter (Sheley et al. 1998). The seeds can remain in the seed bank for 5–8 years. Spotted knapweed can also produce vegetatively by sending out a number of lateral shoots that can grow up to 3 cm away from the parent plant, although they never become detached from the parent root stock (Lacey et al. 1989).

Origins and Distribution: Spotted knapweed was first collected in North America in British Columbia in 1893. It came either as a contaminant of alfalfa seed from Asia Minor or from hybrid alfalfa seeds from Germany (Maddox 1982). Originally, spotted knapweed grew aggressively in the forest steppe zone in Europe. It has been observed in precipitation zones ranging from 20 to 200 cm annually, and at elevations ranging from 578 to over 3040 m (Sheley et al. 1998). According to a 2001 estimate, the plant is reported to have invaded 2.8 million ha in the western United States, of which 68% occurs in Montana (Story et al. 2001).

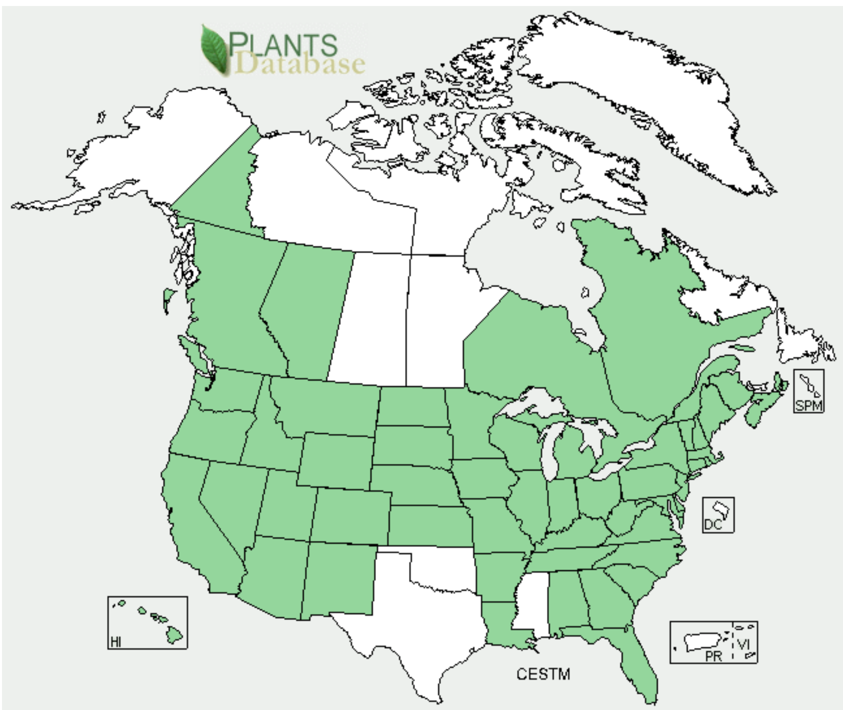

Figure 48. Distribution of Spotted Knapweed.

Probability of Future Expansion: Spotted knapweed is a successful invasive species because of its opportunistic germination patterns, such as remaining in the seed bank until appropriate conditions are present, being able to respond rapidly to sunlight, and being able to tolerate different moisture levels and depth (Spears et al. 1980). A small portion of knapweed that can remain in the seed bank can also outlive the phytotoxic residual periods of pesticides such as picloram (Davis et al. 1993). Competitiveness is also attributed to prolific seed production, the absence of natural enemies, high seed viability, and multiple reproductive methods such as seed dispersal or lateral shoots (Fletcher and Renney 1963; Tyser and Key 1988). Spotted knapweed continues to spread at a high rate across rangeland in the United States and Canada; estimates range as high as a 27% increase in acreage per year. This aggressive spread has made the species a top concern for many state, federal, and private conservation groups/agencies.

Control Technologies: Spotted knapweed can be effectively controlled with herbicides, biological control agents, and hand pulling (Sheley et al. 1998). The herbicides picloram (Tordon), clopyralid (Transline), dicamba (Banvel), 2,4-D, dicamba + 2,4-D, and clopyralid + 2,4-D (Curtail) have all been shown to effectively control spotted knapweed (Sheley et al. 1998; Sheley et al. 2004. Picloram can provide 100% control for 3–5 years when applied during the bud stage of growth (Davis 1990). Clopyralid and clopyralid + 2,4-D applied at the bolt or bud stage can provide control similar to picloram. Herbicides such as dicamba + 2,4-D should also be applied at the bolt to bud stage, whereas dicamba and 2,4-D alone should be applied at the bud stage. Dicamba and 2,4-D alone will provide only short-term control and therefore need to be applied annually until the seedbank is depleted (Sheley et al. 1998; Sheley et al. 2000). Refer to herbicide labels for information regarding application rates, adjuvants, application techniques, and use restrictions.

A number of biological control agents have been shown to reduce spotted knapweed populations. These agents include seedhead feeding flies (*Urophora affinis* and *U. quadrifasciata*), seedhead moths (*Metzneria paucipundctella*), root moths (*Agapeta zoegana*), root weevils (*Cyphocleonus achates*), and seedhead weevils (*Larinus spp*.). At many sites, both seedhead and root feeding insects have been released with success (Sheley et al. 1998; Seastedt et al. 2007). Sheep and goats can also be used to control spotted knapweed, with sheep preferring to graze on young growth. Grazing alone has been shown to provide better control than 2,4-D. After 2,4-D is applied in the spring to remove adult plants, sheep grazing can be used to control seedlings. This integrated strategy has been shown to reduce spotted knapweed rosette density 5 years after treatment (Sheley et al. 1998; Sheley et al. 2004).

Hand pulling is only effective when the entire plant (including the root crown) is removed prior to seed production but, due to the amount of time and labor required, hand pulling may be practical only on small infestations (Sheley et al. 1998). Spotted knapweed requires long-term control to deplete the seedbank, and successful management requires an integrated strategy including the establishment of competitive plants (Sheley et al. 1998). Research is ongoing to identify effective integrated management strategies for control of spotted knapweed.

Direct and Indirect Military Impacts: Given the species growth form and ability to exclude favorable competition, increased levels of soil erosion and other negative impacts have the potential to affect training. Additionally, the species ability to form dense monocultures that exclude native plants is a direct threat to biodiversity. On Fort McCoy, it infringes on karner blue butterfly (a TES) habitat (Westbrook et al. 2005).

Installations (19): Fort Lewis, Fort Hunter Liggett, Fort Irwin¸ Fort Bliss, Fort Carson*, Fort Riley*, Fort Leonard Wood, Fort Chaffee, Fort Polk*, Fort McCoy, Atterbury Reserve Training Area*, Fort Campbell¸ Fort Rucker*, Fort Benning*, Fort Stewart*, Fort A.P. Hill¸ Fort Pickett, Fort Indiantown Gap, Fort Drum.

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Photo credits:

Figure 46: Marisa Williams, University of Arkansas, Fayetteville (Bugwood.org)

Figure 47: L.L. Berry (Bugwood.org)

Figure 48: USDA Plants Database (plants.usda.gov)

Tall Fescue (*Schenodorus phoenix*): Tall fescue is a robust, long-lived, deep-rooted bunchgrass. The plant stands 2-4 ft in height, and has flat leaves 4-18 in. long with erect panicles 15-32 cm long. The panicles terminate in broad loose spikelets (Miller 2003; Hitchcock and Chase 1971). The leaf blades are thin, and the stem is stout and unbranched. The leaves appear dark green in winter, but the color of the leaf can range from yellowish green to deep green depending on available nitrogen in the soil (Rayburn 1993). Flowers are greenish-white, becoming purple. Tall fescue is wind-pollinated and has been used for perennial forage and conservation turf, and forms dense mono-typic stands. Allelopathy has been suggested as a potential mechanism for the success, as evidenced by the increased and very aggressive expansion when infested with endophytes. The endophytes produce alkaloids and enhance the plant's resistance to herbivores and competition (Orr et al. 2005).

Figure 49. Tall Fescue spikelets.

Figure 50. Tall Fescue stand

Reproductive Biology: Tall fescue reproduces vegetatively through rhizomes and sexually through seeds. The rhizomes and extensive rooting system of tall fescue are often cited as a key factor in its ability to adapt to and grow in a variety of environments (Sleper and Buckner 1995). The plant grows from March to June, becoming dormant in the summer to avoid the heat, and growing again in the fall to winter. The panicles are erect and nodding at the tips and spread in the spring before narrowing in the summer (Miller 2003). Seeds are spindle-shaped, granular, and 3–5 mm long, with 5-7 seeds per spikelet.

Origin and Distribution: Tall fescue was introduced to the United States from Europe in the mid-1800s, for turf, forage, stabilization, and wildlife food plots (Orr et al. 2005; Duble 2010). In the 1930s, the ecotype called “Kentucky-31” was discovered and further cultivated for its turf quality (Miller 2003). Kentucky-31 was released for sale in 1942 and is still available for purchase today. Tall fescue can typically be found along roadsides and in meadows, waste places, forest edges, and old fields. It appears to grow best in deep moist soils with medium to heavy texture, but has the ability to grow in a variety of environments (Rayburn 1993).

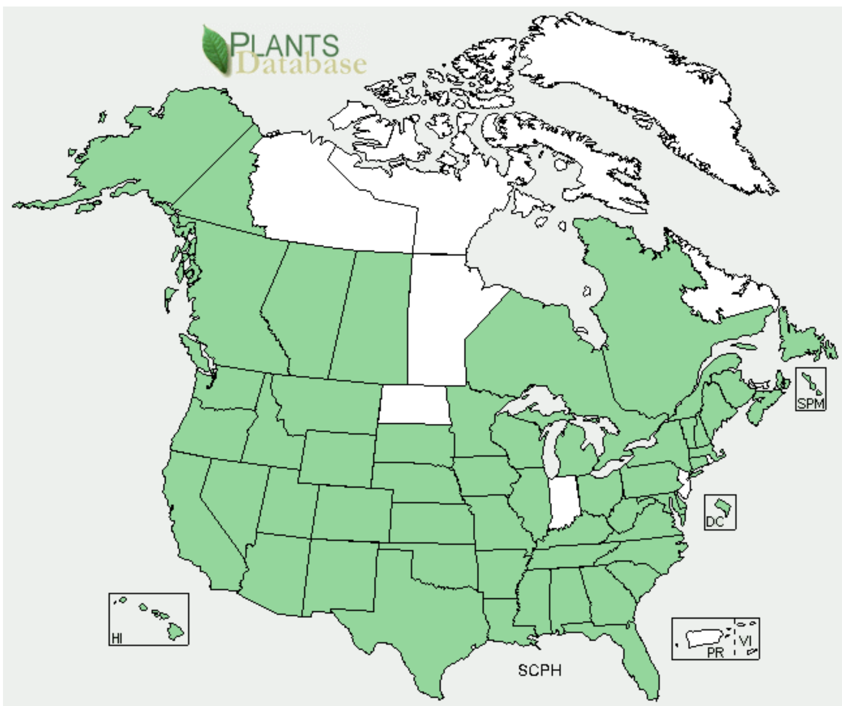

Figure 51. Distribution of Tall Fescue.

Probability for Future Expansion: Tall fescue is cold tolerant, drought tolerant, and is more competitive than most weeds, with the ability to thrive on a wider range of soils. Due to its excellent seed production and lack of native herbivores, tall fescue has the ability to form denser monotypic stands that can inhibit the growth of native tree seedlings (Orr et al. 2005). The cultivar Kentucky-31 is still available for purchase, which increases the spread and probability of tall fescue escaping management (Rayburn 1993). The rooting system of tall fescue also allows it to out-compete native grasses. An endophytic fungus grows on a larger portion of the tall fescue population and increases tall fescue’s invasiveness by enhancing its resistance to herbivores, pathogens, drought, and competition (Orr et. al 2005).

Control Technologies: Tall fescue can be controlled with a number of herbicides that include glyphosate (Roundup), glufosinate (Finale), paraquat (Gramaxone), imazapyr (Arsenal), imazapic (Plateau), sethoxydim (Poast), quizalofop (Assure), clethodim (Select), chlorsulfuron (Telar), and metsulfuron (Escort) (Smith 1989; Dernoeden 1990; Miller 2003). Glyphosate can be applied either in the spring or fall and has been shown to reduce tall fescue cover to less than 12% (Washburn and Barnes 2000). Imazapyr and imazapic should be applied in the spring (Miller 2003), whereas paraquat, glufosinate, sethoxydim, chlorsulfuron, and metsulfuron are more effective when applied in the fall (Smith 1989; Dernoeden 1990). Quizalofop and clethodim should be applied to young, actively growing plants. When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Burning can also be used to control tall fescue and should take place in spring while plants are actively growing. Burns must be repeated to inhibit tall fescue and encourage native grass establishment (Miller 2003; Probasco and Bjugstad 1977). Currently no biological control agents have been released to control tall fescue.

Direct and Indirect Military Impacts: Tall fescue can cause disease in cattle and wildlife, including aborted fetuses, tall fescue toxicosis, fescue foot, and summer fescue toxicosis. These diseases only occur with tall fescue that has been infected with the endophyte. As an aggressive competitor, it can reduce biodiversity, damage TES habitat, and contribute to long-term soil stability. The species can also impede tree regeneration, thus impacting both maneuver lands and forested lands.

Installations (20): Fort Lewis, Fort Hunter Liggett, Fort Irwin*, Fort Carson*, Fort Bliss¸ Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee¸ Fort Polk, Fort McCoy, Fort Campbell¸ Fort Rucker*, Fort Stewart*, Fort Bragg*, Fort Jackson*, Fort A.P. Hill*, Fort Pickett*, Fort Indiantown Gap*, Fort Drum.

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Photo credits:

Figures 49 and 50: Ted Bodner, Southern Weed Science Society (Bugwood.org)
Figure 51: USDA Plants Database (plants.usda.gov)

Tamarisk (*Tamarix* spp.): Tamarisk, also known as salt cedar, is a deciduous shrub or small tree, with some types of tamarisk growing 12-15 ft high. It proliferates very quickly and has dominated most floodplain ecosystems in the western United States. It is reported as the second worst invasive species in the country (Stromberg et al. 2009). The name salt cedar refers to the leaves that resemble those of cedar trees and the salty exudate that collects on the foliage (Birken and Cooper 2006). Tamarisk species are phreatophytes, with deep tap roots to reach the water table. Additionally, tamarisk species are also facultative halophytes and are capable of tolerating salt concentrations in the soil that are much higher than normal (Birken and Cooper 2006; DiTomaso 1998). Tamarisk has small pink five-petaled flowers with pale green foliage, and reddish-brown stems (Pearce and Smith 2003).

Figure 52. Flowering Tamarisk.

Figure 53. Tamarisk along a rail line.

Reproductive Biology: Tamarisk is self-compatible, and in one year a fully mature plant can produce half a million seeds (Pearce and Smith 2003). Tamarisk seeds are very small, have small tufts of hair to aid in dispersal, and have a high initial viability, but must find suitable conditions for germination within a few weeks of dispersal. Flowering can occur from April to October, and seed dispersal typically occurs from late summer to early fall by water and wind (DiTomaso 1998; Pearce and Smith 2003). Further expansion is possible by stump sprouts, layering, and woody fragment propagation. Ideally, seedlings can grow up to 4 m in a single growing season and, upon reaching maturity, are very tolerant of mechanical injury (Mortenson 2008).

Origin and Distribution: Tamarisk is native to southern Europe and the eastern Mediterranean region. Eight species of tamarisk were introduced to the United States in 1823 by nurserymen, and in the 1920s they escaped cultivation (DiTomaso 1998, Whitcraft 2007). Tamarisk can thrive in a wide range of conditions and elevations, from below sea level to over 2000 m (Kennedy and Hobbie 2004). Initially, tamarisks were planted to create windbreaks, stabilize the soil, and as ornamentals (Shafroth and Briggs 2008). Tamarisk is found in at least 23 states. In the western United States, it is reported as the third most frequently occurring woody plant (Sogge et al. 2008; Whitcraft et al. 2007).

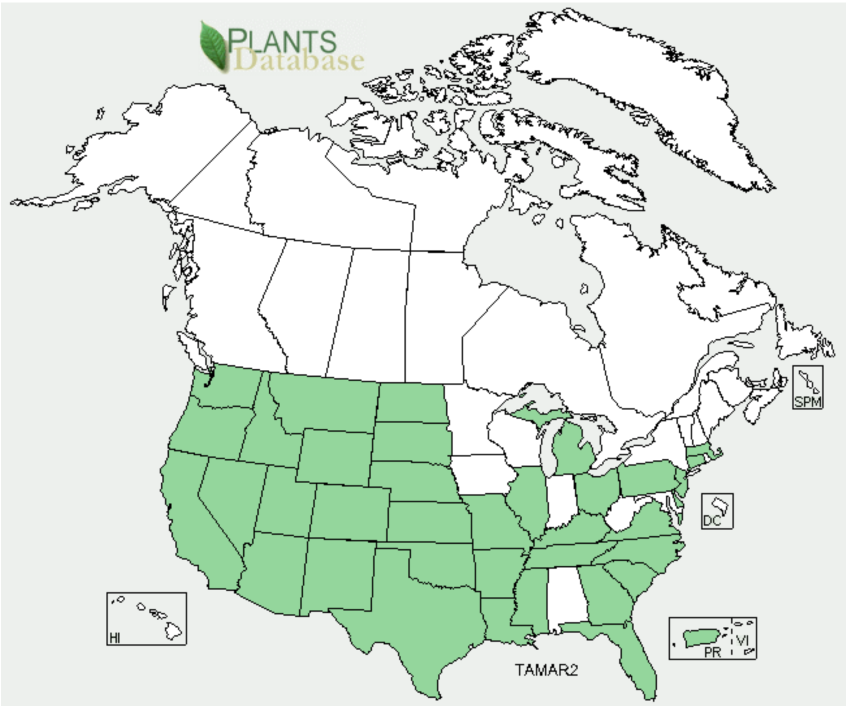

Figure 54. Distribution of Tamarisk.

<u>Probability of Future Expansion</u>: Many factors contribute to tamarisk's success in the United States, including its extensive root system that can reach further for water than the native vegetation, its ability to tolerate highly saline conditions, its tolerance of mechanical injury and a variety of other stress conditions, and its use of wind and water for seed dispersal (DiTomaso 2003; Kennedy and Hobbie 2004). Additionally, tamarisk thrives in floodplain regions; as the river recedes, tamarisk advances into the streambed, continuing forward movement until the stream flow is severely reduced or completely blocked (DiTomaso 2003). High seed production over many months and rapid germination of seeds also contribute to the tamarisk's further spread (Birken and Cooper 2006).

Control Technologies: Saltcedar can be controlled with herbicides, biological control agents, root plowing, or an integrated management strategy. Three herbicides effective for controlling saltcedar are imazapyr (Habitat), triclopyr (Garlon), and glyphosate (Rodeo). Imazapyr alone or in combination with glyphosate is effective as a foliar or aerial spray when applied in August or September (Duncan and McDaniel 1998). To avoid resprouting, treated plants should not be burned or bulldozed for two growing seasons (Carpenter 2003). Glyphosate or triclopyr alone are most effective as cut stump applications in the late fall or winter. Saltcedar should be cut as close to the root crown as possible and herbicide should be applied to the entire cambium ring immediately after cutting. Girdling, or cutting through the cambium around the entire tree circumference with herbicide applied to the frill is effective on large trees (Sudbrock 1993). When using any of the above listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions. Also, note that saltcedar in or near water must be treated with a herbicide formulation labeled for aquatic sites.

The saltcedar leaf beetle (*Diorhabda elongata*) has been released as a biological control agent for saltcedar. Adult and larval stages feed on foliage. In addition, third instar larvae scrape the bark of small twigs, killing more foliage than they consume. Defoliation of saltcedar will result in stem dieback, but resprouting may occur from the base. Initial results indicate that the leaf beetle can defoliate up to 162 ha the third year after release (Coombs et al. 2004). Root plowing can provide 90% control of saltcedar in the field. Root plows should be set to 12–18 in. below the soil surface to cut saltcedar below the root crown. Plowing during hot dry weather can help dry the cut roots, increasing the effectiveness of this control method (Grub et al. 2006). Control methods such as burning or mowing have been shown to be ineffective due to resprouting during the first growing season after treatment (Duncan 1994), but integrating control methods has been successful for saltcedar management. Examples include combining herbicides with grazing, burning, and/or root plowing (Richards and Whitesides 2006; Howard et al. 1983; Carpenter 2003).

Direct and Indirect Military Impacts: Tamarisk often forms dense monospecific stands, which displace native vegetation and reduced plant species diversity. Additionally, the salty exudate that gathers on the leaves of tamarisk collects as litter and inhibits germination of other plants (Harms and Hiebert 2006). Up to 35% more water is taken up by tamarisk relative to the amount of water consumed by native species, which results in higher evapotranspiration rates, reduced water quality, increased fire hazard, and changes in erosion and flooding patterns (Pearce and Smith 2003; Shafroth and Briggs 2008).

Installations (16): Fort Hunter Liggett*, Fort Irwin, Fort Carson, Fort Bliss*, Fort Hood*, Fort Sam Houston*, Fort Sill*, Fort Riley, Fort Chaffee*, Fort Polk*, Fort Benning*, Fort Stewart*, Fort Jackson*, Fort Bragg*, Fort A.P. Hill*, Fort Pickett*.

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Photo credits:

Figure 52: Steve Dewey, Utah State University (Bugwood.org)

Figure 53: Tom Heutte, USDA Forest Service (Bugwood.org)

Figure 54: USDA Plants Database (plants.usda.gov)

Wild Parsnip (*Pastinaca sativa*): Wild parsnip is a tall, stout, herbaceous biennial with a thick, long taproot (Averill and DiTomaso 2007). The plant can also behave as a perennial, dying after the production of flowers and seeds. Wild parsnip has a pinnately compound stem with leaves that are divided into at least five leaflets, with a distinct odor. The flowers are yellow, with no bracts and small sepals. The root can grow up to 1.5-m deep, and, when wild parsnip flowers, it produces a flower stalk that can reach up to 5 ft in height (Averill and DiTomaso 2007; Hendrix and Trapp 1992). Human exposure to the plant results in burns and blisters due to the furanocoumarins, toxins in the plant, that deter herbivory (Berenbaum 1991; Hendrix and Trapp 1992). Wild parsnip grows in a variety of densities and distributions.

Figure 55. Flowering Wild Parsnip.

Figure 56. Wild Parsnip stand.

Reproductive Biology: Wild parsnip reproduces only through seed and is andromonoecious, with individual plants having both hermaphroditic and male flowers (Averill and DiTomaso 2007; Thompson 1978). Growth begins in the spring and lasts until late autumn, with the plant losing most of its leaves during winter. Growth resumes in March, and the plants bolt in April. In the first summer, the plant develops a rosette and a long taproot, sending up a flowering stalk the following spring (Zangerl 1986). The flowers of wild parsnip are clustered together in yellow umbels, and seeds typically over-winter and germinate the following spring (Thompson and Price 1977; Baskin and Baskin

1979). Wild parsnip is considered to have a long growing and germination season, as germination can continue until October.

Origin and Distribution: Wild parsnip is native to Europe and temperate Asia and can be found either naturally or naturalized in most countries. It has been reported in 45 of the 50 states, and is predominantly found in the eastern regions of the country (Averill and DiTomaso 2007). Wild parsnip was introduced to the United States in 1609 and was used for several herbal remedies. The cultivated parsnip escaped and reverted to its wild form. It grows in a variety of conditions but is most commonly found in waste areas, old fields, and along roadsides and railroad embankments (Hendrix and Trapp 1992).

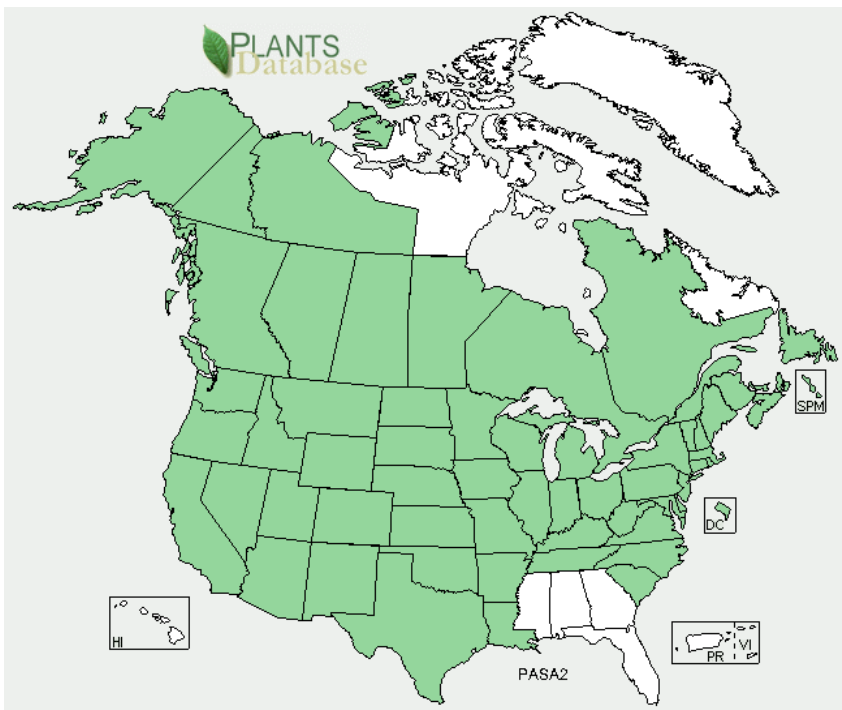

Figure 57. Distribution of Wild Parsnip.

Probability of Future Expansion: Wild parsnip either responds well or is controlled by mowing. Poorly timed mowing results in increased number of seedlings and reduced rate of mortality (Averill and DiTomaso 2007). Additionally, late mowing can result in wild parsnip using the mowing equipment as a dispersal mechanism. Furanocoumarins, toxins in the plant, defend against a wide range of herbivores. The plant’s long tap root is also capable of storing resources if conditions become unfavorable (Zangerl 1986).

Control Technologies: Wild parsnip can be controlled with herbicides such as 2,4-D, glyphosate (Roundup) or metsulfuron

(Escort). Herbicides should be applied in spring or fall when plants are in the rosette stage of growth, whereas treating plants in the bolting or flowering stage may be less successful. 2,4-D and metsulfuron have been shown to provide good to excellent control of wild parsnip with minimal damage to desirable grasses, whereas glyphosate is generally non-selective (Doll and Renz 2007). Refer to herbicide labels for information regarding application rates, adjuvants, application techniques, and use restrictions.

Wild parsnip can also be controlled by severing the root below the root crown. This can be done by hand pulling or with a shovel. To prevent severe skin irritation, care should be taken to avoid contact with plant tissues (Doll and Renz 2007). Burning is not recommended as it allows wild parsnip rosettes to develop rapidly (Eckardt 2006). Mowing has resulted in an increased abundance of wild parsnip by permitting light to reach short, young plants that could not be removed by the mower. Mowing also removes competitive species such as Canada goldenrod, allowing wild parsnip to dominate (Kline 1986). The parsnip webworm (*Depressaria pastinacella*) has been known to injure individual parsnip plants but has not been known to kill entire patches of this weed species; therefore, it is unlikely to be a good biocontrol agent for wild parsnip (Eckardt 2006).

<u>Direct and Indirect Military Impacts</u>: Human contact with wild parsnip causes blisters on exposed skin (Hendrix and Trapp 1992), which is a potential health hazard for Soldiers.

<u>Installations (21)</u>: Fort Lewis, Fort Hunter Liggett, Fort Irwin, Fort Carson*, Fort Bliss*, Fort Riley*, Fort Sill*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort Chaffee*, Fort Polk*, Fort McCoy, Atterbury Reserve Training Area¸ Fort Campbell, Fort Jackson*, Fort Bragg*, Fort A.P. Hill¸ Fort Pickett*, Fort Indiantown Gap¸ Fort Drum.

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Photo credits:

Figure 55: Joy Viola, retired, Northeastern University (Bugwood.org)

Figure 56: Steve Dewey, Utah State University (Bugwood.org)

Figure 57: USDA Plants Database (plants.usda.gov)

Yellow Starthistle (*Centaurea solstitialis*): Yellow starthistle is an annual of the aster family (Asteraceae) and grows to 1.5–3 ft in height (Joley et al. 1992). In the spring, plants bolt, producing a few to several branched, erect stems, each with a terminal flower head. Stems and leaves are covered with cottony wool. Basal leaves are 2–3 in. long and deeply lobed. Upper leaves are short and narrow with few lobes. Flowers are bright yellow with tubular florets and sharp spines surrounding the base. Once established, seedlings and growing plants are highly competitive for soil nutrients and space and can monopolize soil moisture, significantly reducing soil moisture reserves to depths greater than 6 ft (Gerlach et al. 1998). At low densities, yellow starthistle grows faster and roots grow to greater soil depths than cheatgrass (Sheley and Larson 1997). In contrast, at high densities, soil moisture can be depleted from all depths in the soil profile. Thus, high densities of yellow starthistle can produce growth conditions that simulate drought in grassland ecosystems.

Figure 58. Yellow Starthistle flower.

Figure 59. Flowering Yellow Starthistle plant.

Reproductive **B**iology: In contrast with many other invasive Asteraceae, yellow starthistle reproduces exclusively by seed. Flowering typically begins May and continues through September and sometimes much later. There are very low levels of self-fertilization in the species (Sun and Ritland 1998). Each seed head can produce from 35 to 80 or more seeds. Although seed dispersal can take place to a limited degree by wind (maximum dispersal distance of 16 ft), over 90% of seed falls within 2 ft of the parent plant (Roché 1991). Thus, ingestion by migratory birds and anthropogenic influences such as moving vehicles, livestock, and contaminated crop seed or hay are responsible for long distance spread of the seeds (Roché 1992). Germiability and germination rates are typically high (>90%) and are correlated with rainfall events (Benefield et al. 2001). Following germination, robust root growth follows. Seedlings that typically germinate in late fall to early winter grow as basal rosettes, with shoot, flower, and seed formation occurring during the following growing season. As with other Asteraceae, it has been suggested that yellow starthistle might release allelopathic compounds (Merrill and Stevens 1985; Stevens and Merrill 1985), but it remains unclear if that phenomenon exists with this species.

Origins and Distribution: Yellow starthistle is of Eurasian origin and, more specifically, the Mediterranean region. It was first introduced in California in the 1880s through thistle-contaminated alfalfa seed. However, based on genetic analysis of multiple populations, the species probably had more than one introduction from different parent populations (Sun 1997). Although now found in 41 states (USDA 2006) as well as the prairie provinces of Canada, the largest, contiguous infestations of yellow starthistle are in California (estimated at 15 million acres), Idaho, Oregon, and Washington. Distribution within these states is spotty. It is generally most common below 7,000 ft in elevation and is less commonly encountered in desert, high mountain, and moist coastal sites. Yellow starthistle is typically found in full sunlight and deep, well-drained soils. The species has a wide range of moisture tolerance and can exist in areas of annual precipitation ranging from 10 to 60 in.

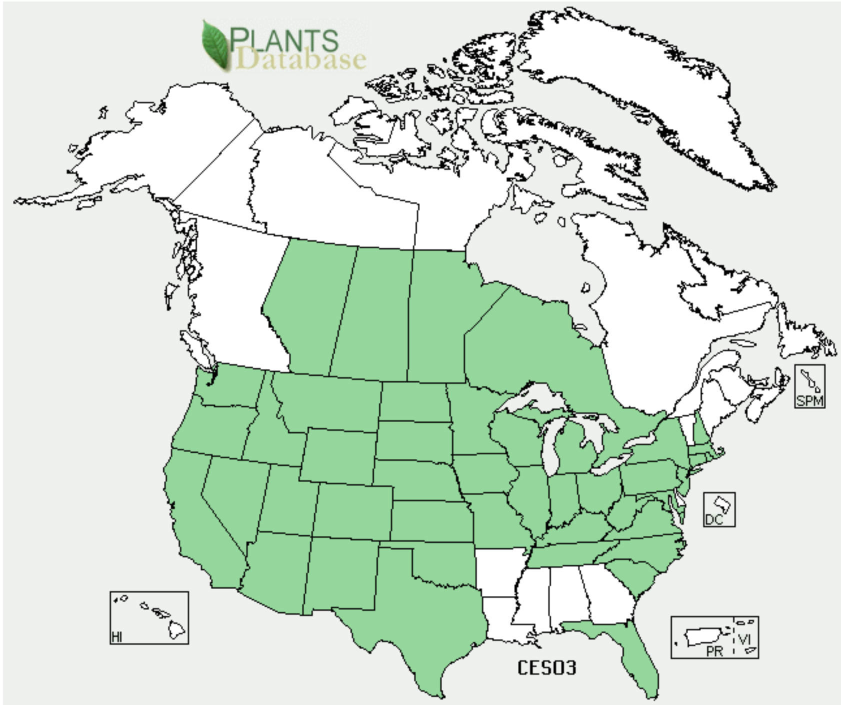

Figure 60. Distribution of Yellow Starthistle.

Probability of Future Expansion: Currently, yellow starthistle is mostly a problem on rangeland sites in California, Oregon, Washington, and Idaho; where it is estimated to spread at the rate of about 6% per year (Wilson et al. 2003). The potential for allelopathic abilities and long-term dispersal via animal ingestion or anthropogenic influences also increase the rate of spread.

Control Technologies: Yellow starthistle may require three or more years of intensive management to significantly reduce the population (DiTomaso 2000). There are a number of control methods that can be used on yellow starthistle alone or in combination. In most cases an integrated management plan is the most successful. A summary of control techniques are discussed below; however, an excellent resource for managers is the Yellow Starthistle Management Guide by DiTomaso et al. (2006b).

A number of pre- and postemergent herbicides can be used to control yellow starthistle, including 2,4-D (Weedar), aminopyralid (Milestone), chlorsulfuron (Telar), clopyralid (Transline), dicamba (Banvel), glyphosate (Roundup), imazapyr (Arsenal), metsulfuron (Escort), picloram (Tordon), and triclopyr (Garlon). A detailed summary of each herbicide can be found in DiTomaso et al. 2006b. In general, preemergent herbicides should be applied before seeds have germinated. Once yellow starthistle plants have emerged, it is best to apply both a preemergent herbicide to control future germinating seeds and a postemergent herbicide to control plants that have already emerged. One herbicide that has pre- and postemergent activity and is very effective is clopyralid. Clopyralid should be applied when yellow starthistle is in the early rosette stage, typically between January and March (DiTomaso et al. 2006b). Drought stress can reduce the efficacy of any herbicide applied, so plants should not be treated under those conditions (DiTomaso et al. 2006b). When using any of the above-listed herbicides, follow all label instructions regarding rate, adjuvants, application technique, and use restrictions.

Small patches of yellow starthistle can be removed by hand pulling. This should be done after plants have bolted but before seeds are produced. To avoid plant recovery, remove all aboveground stem material (DiTomaso et al. 2006b). In some areas, tillage in early summer can also be used to control yellow starthistle as long as the roots are detached from the shoots and it is done prior to seed production (DiTomaso et al. 2006b). Mowing should be done at the spiny to early flowering stage (DiTomaso et al. 2006). Cattle, sheep, and goats have all been shown to graze on yellow starthistle. Cattle and sheep will typically stop consuming yellow starthistle once spiny seedheads are produced, whereas goats will continue grazing into the flowering stage. Grazing is best if used as part of an integrated management program (DiTomaso et al. 2006b; Thomsen et al. 1993). Burning should be done for 2 or more years in early to mid-summer when yellow starthistle is in the early flowering stage but has not yet produced seeds. Germination of seeds can be increased after the first year’s burn, which can aid in depleting the seedbank but also means yellow starthistle must be controlled by some means the year after burn (DiTomaso et al. 2006a; DiTomaso et al. 2006b).

There are a number of biological control agents that have been released to control yellow starthistle. Of these, only three have a widespread distribution, the hairy weevil (*Eustenopus villosus*), the bud weevil (*Bangasternus orientalis*) and a gall fly (*Urophora sirunaseva*). These insects attack flower heads and ultimately reduce seed production. The hairy weevil and the false peacock fly (*Chaetorellia australis*) cause the most seed destruction and are the most abundant of the biological control insects. The false peacock fly was accidentally introduced and is not an approved agent. The Mediterranean rust fungus (*Puccinia jaceae* var. *solstitialis*) is the only pathogen that has been approved for release. The rust causes plant stress by attacking the stems and leaves of yellow starthistle rosettes and early bolts. This stress reduces the number of seed heads and seeds produced. An additional biological control insect being evaluated is the rosette weevil (Ceratapion bassicorne). This insect reduces the number of seed heads and causes shorter plants, but has not yet been approved for release (Coombs et al. 2004; DiTomaso et al. 2006b).

Integrated management plans are the most effective means of controlling yellow starthistle. Examples include herbicides combined with burning, biological control agents, or grazing. Contact local extension agents for guidance on integrated management plans as the sequence of methods used is important. It is also important to include follow-up treatments in long-term management plans to prevent new seedling recruitment and establishment (DiTomaso et al. 2006a).

Direct and Indirect Military Impacts: The thorny spines on the bracts that surround the flower and seed heads of starthistle interfere with military training; its sharp spines can be a serious impediment for Soldier movement. As an exotic ecologic disrupter, it out-competes native plants, reducing biodiversity and TES or other species habitat.

Installations (19): Fort Lewis¸ Fort Hunter Liggett, Fort Irwin, Fort Carson*, Fort Bliss*, Fort Riley*, Fort Sill*, Fort Hood*, Fort Sam Houston*, Fort Leonard Wood*, Fort McCoy*, Atterbury Reserve Training Area*, Fort Campbell*, Fort Jackson*, Fort Bragg*, Fort A.P. Hill*, Fort Pickett*, Fort Indiantown Gap*, Fort Drum*.

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Photo credits:

Figure 58: Peggy Greb, USDA Agricultural Research Service (Bugwood.org)

Figure 59: Charles Turner, USDA Agricultural Research Service (Bugwood.org)

Figure 60: USDA Plants Database (plants.usda.gov)

APPENDIX D

ACRONYMS

Term	Spellout
AR	Army Regulation
ALS	acetolactate synthase
CECW	Directorate of Civil Works, U.S. Army Corps of Engineers
CERL	Construction Engineering Research Laboratory
CONUS	Continental United States
CWMA	Cooperative Weed Management Areas
DA	Department of the Army
DoD	Department of Defense
EO	Executive Order
ERDC	Engineer Research and Development Center
HQUSACE	Headquarters, U.S. Army Corps of Engineers
POC	point of contact
PWTB	Public Works Technical Bulletin
RDD	rose rosette disease
SI	Systeme International de Unites (International System of Units)
TES	Threatened and endangered species
USACE	U.S. Army Corps of Engineers
USDA	U.S. Department of Agriculture
USDA-ARL	USDA Agricultural Research Service
USFWS	U.S. Fish and Wildlife Service