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Animal and Plant Health Inspection Service

Field release of the stem gall weevil *Rhinusa pilosa* (Coleoptera: Curculionidae) for classical biological control of yellow toadflax (*Linaria vulgaris*) (Plantaginaceae) in the contiguous United States.

Environmental Assessment, December 2017

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I. Purpose and Need for the Proposed Action

The U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS), Plant Protection and Quarantine (PPQ), Pest Permitting Branch (PPB) is proposing to issue permits for release of a stem gall weevil, *Rhinusa pilosa* (Gyllenhal) (Coleoptera: Curculionidae). The agent would be used for the biological control of yellow toadflax (*Linaria vulgaris* Mill.) (Plantaginaceae) in the contiguous United States.

This environmental assessment[1] (EA) has been prepared, consistent with USDA, APHIS' National Environmental Policy Act of 1969 (NEPA) implementing procedures (Title 7 of the Code of Federal Regulations (CFR), part 372). It examines the potential effects on the quality of the human environment that may be associated with the release of *R. pilosa* to control infestations of yellow toadflax within the contiguous United States. This EA considers the potential effects of the proposed action and its alternatives, including no action. Notice of this EA was made available in the Federal Register on October 2, 2017 for a 30-day public comment period. The comment period was extended for an additional 15 days to November 16, 2017. One comment was received on the EA by the close of the extended comment period. This comment is addressed in Appendix 5 of this EA.

APHIS has the authority to regulate biological control organisms under the Plant Protection Act of 2000 (Title IV of Pub. L. 106–224). Applicants who wish to study and release biological control organisms into the United States must receive PPQ Form 526 permits for such activities. The PPB received a permit application requesting environmental release of a stem gall weevil, *R. pilosa,* from Europe, and the PPB is proposing to issue permits for this action. Before permits are issued, the PPB must analyze the potential impacts of the release of this agent into the contiguous United States.

The applicant's purpose for releasing *R. pilosa* is to reduce the severity of infestations of invasive yellow toadflax in the contiguous United States. Yellow toadflax was introduced to northeastern North America in the 1600s, and has since spread throughout the United States from these initial New England infestations. Yellow toadflax is considered a serious invasive plant in pastures and crops, particularly on the northern Prairies of North

[1] Regulations implementing the National Environmental Policy Act of 1969 (42 United States Code 4321 et seq.) provide that an environmental assessment "shall include brief discussions of the need for the proposal, of alternatives as required by section 102(2)(E), of the environmental impacts of the proposed action and alternatives, and a listing of agencies and persons consulted." 40 CFR § 1508.9.

America. Yellow toadflax invading pastures and rangelands displaces valued forage species and may be avoided by cattle if they find it unpalatable. In the United States, yellow toadflax infestations have caused economically significant losses to peppermint producers, mainly because chemical control is generally incompatible with production cropping practices (Volenberg et al., 1999). A 1992 report estimated that yellow toadflax infested 7,000 of the 18,000 total acres of cultivated mint in Wisconsin, with 3,000 acres considered moderately to severely infested with the weed (Eagen et al., 1992).

Existing options for management of yellow toadflax are expensive, temporary, ineffective, and can have nontarget impacts. Yellow toadflax is difficult to control using chemical, mechanical, cultural, and existing biological control practices. For these reasons, the applicant has a need to release *R. pilosa*, a host-specific, biological control organism for the control of yellow toadflax, into the environment. The ultimate goals of the proposed action are to reduce the vigor/competitiveness, population density, and spread of yellow toadflax using *R. pilosa*, either alone or in concert with other compatible biological control agents or methods of control.

II. Alternatives

This section will explain the two alternatives available to the PPB—no action and issuance of permits for environmental release of *R. pilosa*. Although the PPB's alternatives are limited to a decision on whether to issue permits for release of *R. pilosa*, other methods available for control of yellow toadflax are also described. These control methods are not decisions to be made by the PPB, and their use is likely to continue whether or not permits are issued for environmental release of *R. pilosa*, depending on the efficacy of *R. pilosa* to control yellow toadflax. These are methods presently being used to control yellow toadflax by public and private concerns.

A third alternative was considered, but will not be analyzed further. Under this third alternative, the PPB would have issued permits for the field release of *R. pilosa*; however, the permits would contain special provisions or requirements concerning release procedures or mitigating measures. No issues have been raised that would indicate special provisions or requirements are necessary.

A. No Action

Under the no action alternative, the PPB would not issue permits for the field release of *R. pilosa* for the control of yellow toadflax. The release of this biological control agent would not take place. The following methods

are presently being used to control yellow toadflax; these methods will continue under the "No Action" alternative and will likely continue even if permits are issued for release of *R. pilosa*, depending on the efficacy of the organism to control yellow toadflax.

1. Chemical Control

Herbicidal control of yellow toadflax requires high application rates and repeated treatments (Jacobs and Sing, 2006). Use of the herbicides picloram, metsulfuron, chlorsulfuron, or imazapic can result in a short term, local reduction of yellow toadflax populations (Jacobs and Sing, 2006).

2. Mechanical Control

Pulling, plowing, or cultivating young yellow toadflax plants in small infestations before they go to seed will provide control, even eradication, if done consistently for several years (Kadrmas and Johnson, undated). The site will need to be revisited frequently to completely eradicate toadflaxes. Mowing is not recommended because it stimulates more plants to grow from the roots. However, mowing can be used to promote uniform growth of the toadflax plants and improve the effectiveness of herbicide applications (Kadrmas and Johnson, undated). Burning is also not recommended because it does not kill the roots and may stimulate shoot growth. Propane torches can be used to kill stalks before they seed, preventing seed production (Kadrmas and Johnson, undated).

3. Cultural Control

Introducing and maintaining competitive plants thwarts yellow toadflax from establishing on rangelands. Healthy rangeland plant communities can remain free of yellow toadflax if they provide season-long cover to prevent the establishment of toadflax seedlings (Kadrmas and Johnson, undated).

4. Biological Control

In the United States, there are six insects released for biological control of yellow toadflax. The beetle *Brachypterolus pulicarius* (Nitidulidae) was accidentally released pre-1919 from European origin. This beetle can cause high seed reduction but has a minimal impact. The noctuid moth *Calophasia lunula* causes defoliation of toadflax plants. The larvae of the moth *Eteobalea serratella* (Cosmopterigidae) cause damage to the root system of toadflax plants. Larvae of the weevil *Rhinusa* (*Gymnetron*) *antirrhini* (Curculionidae) attack immature seeds inside the seed capsules, but this agent has had minimal impact on yellow toadflax. Adults of the weevil *Gymnetron linariae* (Curculionidae) attack the shoots of yellow toadflax while larvae develop in galls formed on the roots and rhizomes. Adults of the weevil *Mecinus janthinus* (Curculionidae) feed on leaves and stems of yellow toadflax, and larvae mine yellow toadflax stems.

B. Issue Permits for Environmental Release of *R. pilosa*

Under this alternative, the PPB would issue permits for the field release of the stem gall weevil, *R. pilosa*, for the control of yellow toadflax. These permits would contain no special provisions or requirements concerning release procedures or mitigating measures.

Biological Control Agent Information

1. Taxonomy

Common name: none
Scientific name: *Rhinusa pilosa* (Gyllenhal)
Synonyms: none

Phylum: Arthropoda
Class: Insecta
Order: Coleoptera
Family: Curculionidae
Subfamily: Mecininae
Tribe: Mecinini
Genus: *Rhinusa*

2. Description of *R. pilosa*

The adult body length of *R. pilosa* is 3.2 to 4.3 millimeters; the color of the body is black, and it is densely covered with bristled blackish hairs; the rostrum (snout or beak) is slightly curved, and is roughly punctured at the end in the male, while in the female the rostrum is nearly smooth at the end. The elytra (hardened forewings) are twice as long as wide, with nine ridges or grooves, and they are roughly punctured (Figure 1).

Figure 1. *Rhinusa pilosa* (Gyllenhal). (From De Clerck-Floate et al., 2012)

3. Geographical Range of *R. pilosa*

a. Native Range

In the literature, *R. pilosa* is reported to be widely distributed in central, eastern and south-eastern Europe, Russia, Holland, Denmark, and southern Sweden (Lucht, 1987), as well as in Algeria and Tunisia in North Africa (Lohse and Tischler, 1983). According to Hoffmann (1958), *R. pilosa* is a

rare species in France, southern Germany, Denmark, Greece, and Algeria. However, the geographical distribution of *R. pilosa* known from the literature is doubtful because records have seldom distinguished between *R. pilosa* and a similar species, *R. brondelii*. Confirmed localities indicate that the geographical range of *R. pilosa* extends from Sweden in the north to France in the south-west and Serbia in the south-east. *Rhinusa pilosa* is a rare species in western Europe.

b. Expected Attainable Range of *R. pilosa* in North America

The native range of *R. pilosa* extends over a wide range of climatic conditions in Eurasia; the weevil is therefore is expected to be well-adapted to climatic conditions in most of the invasive range of yellow toadflax in the United States and Canada. The population proposed for introduction into North America originates from north and east of Belgrade in Serbia.

3. Life History of *R. pilosa*

Rhinusa pilosa is a univoltine (one generation per year) shoot-galling species that overwinters as an adult. A gall is an abnormal growth of the plant, caused in this case, by *R. pilosa* egg laying (oviposition) into the plant stem. Based on emergence data obtained under both controlled (laboratory) and field conditions, adult emergence occurs relatively early in the growing season (i.e., March-May), coinciding with the spring burst of shoot growth from tap roots. Post-hibernated adults feed 3 to 5 days on yellow toadflax shoots and foliage before mating begins. Oviposition follows approximately 10 days later, loosely timed to occur from April to May, depending on environmental conditions. Field observations indicate that *R. pilosa* adults disperse among yellow toadflax patches during the spring oviposition period.

Gall development is complete approximately 8 to 10 days after oviposition under laboratory conditions, corresponding with emergence of the first larval instar (Barnewall, 2011). *Rhinusa pilosa* has a total of three larval instars that feed and continue development on host tissues within the developed galls. Pupation is also completed within the gall.

Adults that have emerged from the pupal shell remain within the gall for 10 to 15 days while they continue to feed on host tissues before escaping via holes chewed through the gall's outer surface. Larval/pupal development lasts about 45 to 50 days; the estimated time for complete development from egg to adult emergence from the gall is 55 to 65 days. After emergence, the adults feed externally on host stems for about 10 days. Thereafter, the adults hide in litter or cracks in the soil during the day. Summer dormancy is interrupted by occasional feeding, mainly in the evening and at night. In late autumn, adults feed shortly before going into diapause within soil or leaf litter.

Galls caused by *R. pilosa* ovipositing into the stems of yellow toadflax are globular, round, or oblong, green structures that are positioned between the middle and tip of the host stem (Figure 2). Average dimensions of these galls are 9.1 ± 0.5 millimeters (mm) wide by 18.7 ± 1.7 mm long (Barnewall, 2011). Although galls typically contain multiple *R. pilosa* larvae per gall, sometimes galls only contain one larva. On average each gall produced 2.4 adults during laboratory and field studies conducted in Serbia. The highest number of *R. pilosa* adults to emerge from a single gall was 17 (De Clerck-Floate et al., 2012).

Figure 2. *Rhinusa pilosa* galls on yellow toadflax, A. field; B. field cage test; C. laboratory test with yellow toadflax from North America. (from De Clerck-Floate et al., 2012).

III. Affected Environment

A. Taxonomy of Yellow Toadflax

Yellow toadflax was described by the Scottish botanist, Philip Miller in 1768 (Sutton, 1988). Its current scientific name is *Linaria vulgaris* Mill., as verified through the Integrated Taxonomic Information System (ITIS) on-line database (http://www.itis.gov). The availability and application of molecular techniques in published and ongoing studies has significantly improved understanding of the taxonomy and genetics of the toadflaxes and their various biotypes and hybrids.

Considerable hybridization among yellow toadflax (*Linaria vulgaris*), Dalmation toadflax (*L. dalmatica* (L.) Mill.), and broomleaf toadflax (*L. genistifolia* (L.) Mill.) may make strict species boundaries unclear and make positive identification difficult, especially in the very few locations where two or more of these species co-exist (Pauchard et al., 2003). The probability of hybridization within *Linaria* is thought to be fairly high (Sutton, 1988). Hybrids of *L. dalmatica* and *L. vulgaris*, *L.*

dalmatica and another toadflax species, *L. euxina* Velen. (Bruun, 1937; Chater et al., 1972), and *L. dalmatica* (L.) Mill. and *L. genistifolia* (L.) Mill. ssp. *genistifolia* (Docherty, 1982) have been produced under laboratory conditions. *Linaria vulgaris*, *L. dalmatica*, and *L. genistifolia* growing throughout North America are highly variable in shape, size, and sometimes color.

Naturally occurring hybridization between *L. vulgaris* and *L. dalmatica* has not been historically recorded (Olsson, 1974; 1975), although hybrids have been described from central Romania, and cannot be excluded as occurring in nature, according to Sutton (1988). Field observations suggest that hybrid forms of *L. vulgaris* x *L. dalmatica* crosses may occur frequently throughout the western United States. Molecular diagnostic techniques have confirmed the occurrence of hybridization between *L. vulgaris* and *L. dalmatica* from samples field collected at sites in Montana (Fleischmann et al., 2007; Ward et. al., 2009), Idaho, and Colorado. Narrow-leaved forms of *L. dalmatica* may therefore actually be hybrids of *L. dalmatica* and *L. vulgaris*. Naturally occurring hybridization between *L. vulgaris* and *L. repens* (L.) Mill. (striped toadflax) is common. This hybrid, described as *Linaria* x *sepium* J.G. Allman from the British Islands in 1843, was subsequently discovered near St. John's, Newfoundland and Labrador, Canada in the early 1900s (Saner et al., 1995). Field collected hybrids have also occurred between *L. vulgaris* and *L. arvensis* (L.) Desf. (= *L.* x *heribaudii* Camus) in south central France; between *L. vulgaris* and *L. supina* (L.) Chaz. (= *L.* x *cornubiensis* Druce) in southwestern Britain; and between *L. vulgaris* and *L. angustissima* Borbás (= *L.* x *oligotricha* Borbás) (Sutton, 1988).

Non-native ornamental toadflaxes that have escaped cultivation in North America include *L. purpurea* (L.) Mill. (purple toadflax), *L. maroccana* Hook f. (Morocco toadflax), *L. bipartita* (Vent.) Willd. (clovenlip toadflax), and *L. pinifolia* (Poir.) Thell. (pineneedle toadflax). These species are easy to distinguish from the target weed as the escaped ornamentals typically have red to purple flowers, compared to the yellow blossoms of *L. vulgaris*. *Nuttalanthus canadensis* (L.) D.A. Sutton (Canada or oldfield toadflax) is a native North American species now considered a more distant relative of *L. dalmatica*, *L. vulgaris*, and *L. genistifolia*, that can also be distinguished by flower color from the exotic, weedy toadflaxes, with its purple to blue vs. yellow flowers (CDFA, Undated).

The appropriate taxonomic placement for yellow toadflax is as follows:

Kingdom: Plantae
Division: Magnoliophyta

Class: Magnoliopsida (Dicots) Subclass: Asterideae
Order: Lamiales
Family: Plantaginaceae (= Veronicaceae)
Tribe: Antirrhineae
Genus: *Linaria*

Scientific name: *Linaria vulgaris* Mill.
Synonyms: *Linaria linaria* (L.) Karst.; *Linaria acutiloba* Fisch. ex Rchb.
Common names: yellow toadflax; butter-and-eggs; eggs and bacon; common toadflax; toad-flax; Jacob's ladder; flaxweed; ramsted; wild snapdragon; perennial snapdragon; common linaria; rabbit-flower; imprudent lawyer; *linaire commune*; *linaire*; *gueule de lion*; *gueule de lion des champs*; *lin des crapauds*; *muflier sauvange*; *pain de beurre*; *pisse de chien.*

Taxonomically verified voucher specimens of *Linaria vulgaris* from Canadian national collections are deposited with the Agriculture and Agri-Food Canada (AAFC; formerly Department of Agriculture) Herbarium, and the Canadian Museum of Nature, both in Ottawa, Canada (Saner et al., 1995), and with AAFC, Lethbridge Research Centre, Lethbridge, Alberta, Canada, and the Entomology Museum, Montana State University, Bozeman, Montana (Hennessey, 1996) for material used in host specificity testing of *R. pilosa.*

B. Areas Affected by Yellow Toadflax

1. Native Range of Yellow Toadflax

Yellow toadflax is native to most of Europe and northern Asia, from the United Kingdom south to Spain in the west, and east to eastern Siberia and western China. Its native range encompasses Western Asia (Turkey), Siberia (Russian Federation – Eastern Siberia and Western Siberia), the Soviet Far East, China (Gansu, Henan, Jiangsu, Shaanxi, Shandong, and Xinjiang Provinces), Northern Europe (Denmark, Finland, Ireland, Norway, Sweden, and the United Kingdom), Middle Europe (Austria, Belgium, former Czechoslovakia, Germany, Hungary, Netherlands, Poland, and Switzerland), East Europe (Belarus, Moldova, the European part of the Russian Federation, and Ukraine), Southeastern Europe (Albania, Bulgaria, Greece, Italy, Romania, and former Yugoslavia) and Southwestern Europe (France, including Corsica, and Spain) (Sutton, 1988; Tutin et al., 1972; USDA-ARS, 2012).

2. Introduced Range of Yellow Toadflax

Yellow toadflax has become naturalized in temperate regions including North America, Chile, Guatemala, Jamaica, Australia, New Zealand, and South Africa (IUCN-ISSG, 2012; USDA-ARS, 2012).

Yellow toadflax occurs throughout the contiguous United States, and in every province and territory of Canada except for Nunavut and Labrador

(Figure 3) (USDA-NRCS, 2012; Saner et al., 1995). Although it is most common in northeastern North America, it occurs in localized dense infestations in other parts of the continent. It is particularly problematic in annual crops of the Prairie provinces, the Peace River Lowland, and Aspen Park ecoregions of Canada (Leeson et al., 2005). The northern limit of yellow toadflax in North America ranges from 55-65° N (Saner et al., 1995), indicating the wide ecological amplitude, and therefore, high potential for further spread of this weed.

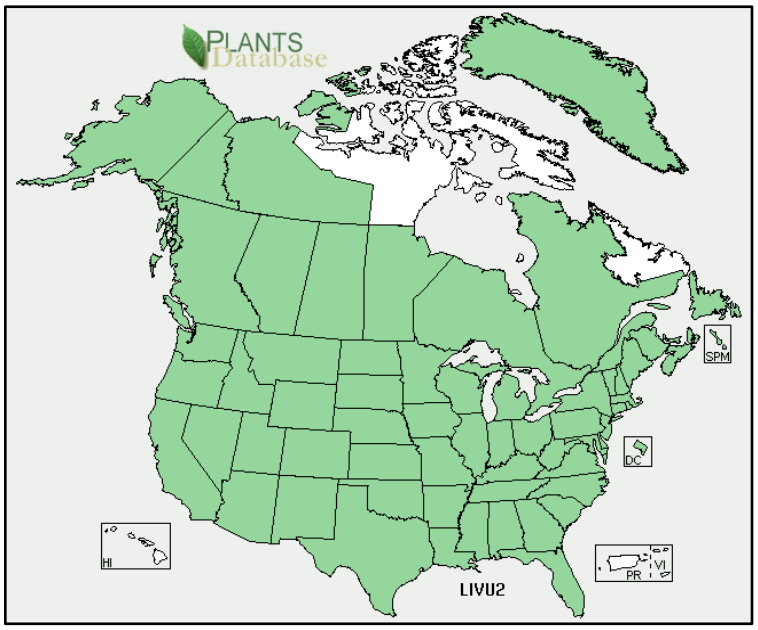

Figure 3. Green colored area comprises North American species distribution of yellow toadflax based on reported presence within states, provinces and territories. (USDA-NRCS, 2012).

3. Habitats Where Yellow Toadflax is Found in North America

Invasive exotic toadflaxes tolerate a broad range of climatic conditions and soil types (Saner et al., 1995; Vujnovic and Wein, 1997; De Clerck-Floate and Harris, 2002; McClay and De Clerck-Floate, 2002). Yellow toadflax has successfully established in North American rangelands, grasslands, scrub and shrublands, and to varying degrees in agricultural areas (croplands, fields, and pastures), riparian zones, and in ruderal zones, along roads, in dunes, and on disturbed and cultivated land (Alex, 1962; Coupland et al., 1963; Robocker, 1974; Darwent et al., 1975; Morishita, 1991; Jacobs and Sheley, 2003; Pauchard et al., 2003; D'Antonio et al., 2004; Sutton et al., 2007; Dodge and Fulé, 2008). Once established, invasive toadflaxes have the potential, like many weeds, to spread into adjacent non-disturbed areas (Zilke and Coupland, 1954).

Yellow toadflax is widespread in North America but is commonly associated with the relatively summer-moist, coarse soils found in the northwestern and north central United States, and the dark loamy (Chernozem) soils of the northern part of the Canadian Prairie provinces.

It occurs in agricultural communities in the western United States and Canada, and in disturbed areas in the northcentral and northeastern United States. Yellow toadflax is a common, naturalized weed of roadsides and poor soils. In North America, toadflaxes have been found at elevations exceeding 3,048 meters and yellow toadflax, in particular, has spread into high mountain valleys and parks, including forested rangelands. It can tolerate sub-arctic conditions (Saner et al., 1995).

C. Plants Related to Yellow Toadflax and Their Distribution

The genus *Linaria* Mill. was traditionally placed in the Scrophulariaceae (Figwort) family (Sutton, 1988). The traditional definition of the Scrophulariaceae family (i.e., Scrophulariaceae *sensu lato*) was based more on the traits that family members lacked, rather than on shared common traits (Olmstead and Reeves, 1995; Olmstead et al., 2000; Bremer et al., 2001; Olmstead et al., 2001; Tank et al., 2006). Revisions of the Scrophulariaceae based on molecular phylogenetic analyses indicated that *Linaria* would be more appropriately included within the expanded Plantaginaceae (Plantain) family (Albach et al., 2005; Olmstead et al., 2001; Ghebrehiwet et al., 2000). In some cases, *Linaria* may be listed under the plant family Veronicaceae, which is an older name for the Plantaginaceae family.

Most of the former Scrophulariaceae genera have been re-distributed among seven families representing independent lineages of the order Lamiales: 1) Scrophulariaceae *sensu stricto*; 2) Calceolariaceae; 3) Linderniaceae; 4) Orobanchaceae; 5) Plantaginaceae *sensu lato* [= Veronicaceae]; 6) Phrymaceae; and 7) Stilbaceae (Tank et al., 2006; Schäferhoff et al., 2010).

Scrophulariaceae *sensu stricto*. The revised Scrophulariaceae *sensu stricto* family is comprised of eight tribes: Aptosimeae, Buddlejeae, Hemimerideae, Leucophylleae, Limoselleae, Myoporeae, Scrophularieae and Teedieae (Oxelman et al., 2005). The Scrophulariaceae *sensu stricto* tribes are predominantly distributed in the southern hemisphere (four in southern Africa and one in Australia); only *Scrophularia* and *Verbascum* species, members of the Scrophulariaeae tribe, have radiated significantly in the northern hemisphere but they are entirely Old World in origin (Tank et al., 2006). Scrophulariaceae *sensu stricto* genera in the temperate North American flora (north of Mexico) include genera from the Buddlejeae (*Buddleja*) and Leucophylleae (*Capraria* and *Leucophyllum*) tribes; the northern-most extent of *Capraria's* and *Leucophyllum's* ranges reach only as far north as the southern United States (Tank et al., 2006).

Calceolariaceae. The Calceolariaceae family includes three genera:

Calceolaria, *Jovellana*, and *Parodittia* (Andersson, 2006). The approximately 270 *Calceolaria* species have a New World distribution that spans from Chile to Mexico. *Jovellana* species originate from both New Zealand and Chile, while *Parodittia* species are is known only from Peru (Andersson, 2006; Tank et al., 2006).

Linderniaceae. Linderniaceae family genera include *Craterostigma*, *Lindernia*, *Artanema*, *Picria*, *Torenia*, *Crepidorhopalon*, *Micranthemum*, and *Stemodiopsis*, although intergeneric relationships remain uncertain (Rahmanzadeh et al., 2005; Tank et al., 2006; Schäferhoff et al., 2010). Two genera, *Micranthemum* and *Lindernia*, contain species with southern United States distributions (Tank et al., 2006).

Orobanchaceae. Scrophulariaceae *sensu lato* species ranged from fully autotrophic (capable of self-nourishment), partially parasitic, to fully parasitic. All parasitic Scrophulariaceae *sensu lato* species are now placed within the Orobanchaceae (Wolfe et al., 2005; Schäferhoff et al., 2010).

Phrymaceae. Evidence is presently lacking to support the monophyly (descent from a common evolutionary ancestor or ancestral group) of a Phrymaceae family (Schäferhoff et al., 2010; Tank et al., 2006).

Stilbaceae. Schäferhoff et al. (2010) reports conflicting results of molecular analysis performed to explain the relatedness of the genera within the Stilbaceae family. The traditional circumscription of this family included six genera: *Stilbe*, *Campylostachys*, *Euthystachys*, *Eurylobium*, *Thesmophora*, and *Xeroplana* (Tank et al., 2006). These and additional genera were subsequently subdivided into tribes: Bowkerieae (*Anastrabe*, *Bowkeria*, and *Ixianthes*); Stilbeae (*Kogelberia, Stilbe*, *Retzia*, *Campylostachys*, *Euthystachys*, and *Nuxia*) and Hallerieae (*Charadophila* and *Halleria*) (Tank et al., 2006). All species are heath-like shrubs with a primarily South African distribution; only *Nuxia* has a wider range, reaching tropical Africa and the Arabian Penninsula (Schäferhoff et al., 2010).

Plantaginaceae *sensu lato*. The revised Plantaginaceae family is comprised of a range of life forms (herbs, shrubs and a few rooted aquatic species) with a cosmopolitan and mostly temperate zone distribution. The majority of the Scrophulariaceae *sensu lato* genera, including *Linaria*, have been transferred to the Plantaginaceae (Albach et al., 2005). Plantaginaceae tribes include Plantagineae, Veroniceae, Digitalideae, Globularieae, Sibthorpieae, Callitricheae, Antirrhineae, Russelieae, Cheloneae, Angelonieae, and Gratioleae (Tank et al., 2006; Estes and Small, 2008).

The genus *Linaria's* approximately 150 species (the largest genus in the

Antirrhineae tribe) have a Palaearctic (region comprising Eurasia north of the Himalayas, together with North Africa and the temperate part of the Arabian peninsula) distribution (Niketić and Tomović, 2008). Fourteen *Linaria* species (including the target species, *L. vulgaris*) and hybrids of *L. dalmatica* and *L. vulgaris* are recorded as established in North America (USDA-NRCS, 2012; Ward et al., 2009; Jeanneret and Schroeder, 1991). According to Wiersema and León (1999), six *Linaria* species are considered economically important ornamentals: *L. bipartitia*, *L. dalmatica*, *L. maroccana*, *L. pinifolia*, *L. purpurea*, and *L. reticulata* (Sm.) Desf.; some (e.g., *L. maroccana*) have escaped cultivation and become invasive to varying degrees in North America (Saner et al., 1990; USDA-NRCS, 2012). Species with a perennial life history, such as *L. maroccana* and *L. purpurea*, are most likely to serve as alternative nontarget hosts for toadflax biocontrol agents. Most known specialist herbivores (plant feeders) on toadflax are thought to be predominantly associated with short-lived perennials, although this degree of host persistence is not required for all known toadflax biological control agents (Hansen and Gassmann, 2002). *Linaria maroccana* is present in the northeast (Connecticut, Massachusetts, Maine, New Hampshire, New York, Virginia, West Virginia), southwest (Arizona), Pacific southwest (California), and northwest of the United States, and in the Canadian province of Prince Edward Island. *Linaria purpurea's* North American distribution is restricted to the west coast (British Columbia, Washington, and California).

The only native North American species (or varieties) conventionally placed within *Linaria* were reclassified in 1988, remaining in the tribe Antirrhineae but moved to a new genus, *Nuttallanthus* D.A. Sutton (Sutton, 1988). The three historically recognized native North American *Linaria* species or varieties referred to as the North American toadflaxes are now known as *Nuttallanthus canadensis* (formerly *Linaria canadensis*), *N. floridanus* (Chapman) D.A. Sutton (formerly *Linaria floridana*), and *N. texanus* (Scheele) D.A. Sutton (formerly *Linaria canadensis* var. *texana* and *Linaria texana*) (Crawford and Elisens, 2006). Both *N. canadensis*, also known by the common name oldfield toadflax, and *N. texanus* have extensive North American distributions (*N. canadensis*: In the United States it is found in the following states: Alabama, Arkansas, California, Connecticut, Delaware, Florida, Georgia, Iowa, Illinois, Indiana, Kansas, Louisiana, Massachusetts, Maryland, Maine, Michigan, Minnesota, Missouri, Mississippi, North Carolina, North Dakota, New Hampshire, New Jersey, New York, Ohio, Oklahoma, Oregon, Pennsylvania, Rhode Island, South Carolina, South Dakota, Tennessee, Texas, Virginia, Vermont, Washington, Wisconsin, and West Virginia, as well as the Washington, D.C. In contrast, *N. floridanus* has been reported only from the southeastern United States in Alabama, Georgia, Florida, and Mississippi.

Genera in the Antirrhineae tribe popular as ornamentals include *Antirrhinum*, *Cymbalaria*, *Lophospermum*, and *Maurandya*. Garden snapdragon (*Antirrhinum majus* L.) is a European perennial generally used as an ornamental annual in North America. Genera in the Plantaginaceae family used as ornamentals include *Bacopa*, *Chelone*, *Collinsia*, *Digitalis*, *Erinus*, *Globularia*, *Gratiola*, *Hebe*, *Limnophila*, *Penstemon*, *Russelia*, *Tetranema*, *Veronica*, and *Veronicastrum*. Plantaginaceae genera commonly encountered in areas where exotic toadflaxes occur in North America include *Collinsia* (blue-eyed Mary), *Penstemon* (beardstongue), and *Veronica* (speedwell).

As a result of widely sweeping and accepted systematic reorganization, no *Linaria* species are considered to be native to North America. Plant species that were used in testing the specificity of *R. pilosa* to yellow toadflax are listed in appendix 1.

IV. Environmental Consequences

A. No Action

1. Impact of Yellow Toadflax

a. Animals

Several secondary chemicals are present in *Linaria* spp. These include pharmacologically active flavonoids (e.g., linarin, acacetin, and quercetin), saponins, quinazoline alkaloids (e.g., vasicine) and iridoid glycosides (e.g., antirrhinoside) (Sing and Peterson, 2011; Saner et al., 1995; Vujnovic and Wein, 1997). Reports of occasional cases of mild poisoning from yellow toadflax exist for cattle, but were thought to be rare because cattle avoided toadflax (Mitich, 1993). Cattle would unlikely be able to consume enough toadflax through typical grazing activities to exceed toxicity thresholds (Sing and Peterson, 2011). Toadflax also has been reported to be mildly toxic to horses, but sheep and goats appear unaffected (Jacobs and Sing, 2006).

b. Plants

In Canada, yellow toadflax is considered a serious invasive plant in pastures and crops, particularly on the Prairies (Coupland et al., 1963; Harker et al., 1995; Baig et al., 1999), but also is a growing problem in the mountain rangelands of British Columbia (BCMFR-IAPP, 2012), where it displaces valued forage species and is avoided by grazing cattle. Studies of the impact of yellow toadflax infestations on crop production in Alberta have reported a 33 pecent seed yield loss in the forage species red fescue (*Festuca rubra* L.) (Darwent et al., 1975), and a 20 percent yield reduction in canola and wheat (O'Donovan and McClay, 1987; O'Donovan and

Newman, 1989). Exotic invasive *Linaria* spp. serve as alternate hosts for crop diseases including cucumber mosaic virus and broad bean wilt virus (Pariera Dinkins et al., 2007; Rist and Lorbeer, 1989).

Threats to native species and ecosystems, changes in biodiversity, and the health consequences attributed to invasive toadflaxes, or to any invasive plant, are difficult to quantify. The former Secretary of the Interior, Bruce Babbitt, reported that exotic plants are invading wildlands in the United States at a rate of roughly 700,000 hectares per year. In 2000, it was estimated that economic losses caused by exotic species were upwards of $137 billion annually in the United States alone (Pimentel et al., 2000). Exotic toadflaxes contribute to these losses as the total area of lands invaded by toadflax is extremely high in western North America, especially in fire-affected areas (Dodge and Fulé, 2008).

c. Human Health

Hruska (2003) determined that the pollen of yellow toadflax had an allergen index of 4.0 out of 10, and categorized it as a moderately allergenic species.

d. Beneficial Uses

The beneficial uses of yellow toadflax are briefly discussed in Mitich (1993) and Saner et al. (1995). Yellow toadflax is characterized as a prized ornamental intentionally introduced to North America in the 1600s. Yellow toadflax was also used historically in folk medicinal preparations, ingested to treat edema, jaundice, liver diseases, and skin problems, and applied topically to hemorrhoids, skin eruptions, sores, and ulcers.

This species also was described as valuable in veterinary applications, to treat cattle unable to ruminate and functioned as an insecticide in animal bedding. Yellow toadflax also has been described as a plant source of yellow textile dye and as a fly poison when boiled in milk.

Linaria species are thought to have little value as forage plants because they are generally believed to be unpalatable to livestock (Lajeunesse, 1999). However, while livestock and wildlife may not favor toadflax, they are capable of browsing on it, and can adapt to using toadflax as a significant food source as circumstances dictate (Jacobs and Sing, 2006; De Clerck-Floate et al., 2012). Toadflax infestations may, however, originate from seeds eliminated by livestock and wildlife (Reed, 1970).

Yellow toadflax is tolerant to heavy metals and has been used in the reclamation of mined areas, sites despoiled by heavy metal-laden sewage sludge, and abandoned gravel pit slopes (Long, 1974; Heagy and Cavers, 1980).

2. Impact from Use of Other Control Methods

The continued use of chemical, mechanical, cultural, and biological controls at current levels would be a result if the "no action" alternative is chosen. These environmental consequences may occur even with the implementation of the biological control alternative, depending on the efficacy of *R. pilosa* to reduce yellow toadflax populations in the contiguous United States.

a. Chemical Control

Chemical control of yellow toadflax can be erratic because of the genetic variability of the plants, and their ability to grow in many sites and climates (Kadrmas and Johnson, undated). Because seeds can remain in the soil for a long time, it may require treatment with herbicides for 12 or more years to completely eradicate toadflaxes from an area (Kadrmas and Johnson, undated). Also, herbicides can have impacts on non-target plant species.

b. Mechanical Control

Pulling or cultivating of yellow toadflax plants can be effective but is tedious and time-consuming, and the entire lateral root system must be removed or plants will regrow. This type of control is not recommended for medium to large stands of yellow toadflax. Mowing and burning do not kill the roots and may stimulate shoot growth.

b. Cultural Control

Maintaining competitive plants thwarts toadflaxes, but healthy rangeland must be consistently monitored and spot herbicide treatments of invasive toadflaxes are necessary. This method is most effective after toadflaxes have been removed from the environment by other methods.

d. Biological Control

Many of the agents previously released either have negligible impact, are affected by predators or pathogens, or have failed to establish (Jacobs and Sing, 2006). However, all of the agents released can contribute to the overall erosion of yellow toadflax's fitness and should be considered when an integrated pest mangament approach is used to control this weed (Jacobs and Sing, 2006).

B. Issue Permits for Environmental Release of *R. pilosa*

1. Impact of *R. pilosa* on Nontarget Plants

Host specificity of *R. pilosa* to yellow toadflax has been demonstrated

through scientific literature, field observations, and host specificity testing. If the the candidate biological control agent only attacks one or a few closely related plant species, it is considered to be very host-specific. Host specificity is an essential trait for a biological control organism proposed for environmental release.

a. Scientific Literature

Host records from the scientific literature are questionable because they do not adequately distinguish *R. pilosa* form the similar *R. brondelii*. According to Hoffmann (1958), *R. pilosa* is reported as causing galls on several *Linaria* species, but in central and southeastern Europe it mainly occurs on *L. vulgaris*. The reported alternative hosts are *Chaenorrhinum minus* (L.) Lange, *Linaria repens*, *L. purpurea*, *L. simplex* (Willd.) DC., 1805, *L. reflexa* (L.) Desf. (Hoffman, 1958; Lohse and Tischler, 1983) and *L. gharbensis* Batt. & Pit. in North Africa (Mimeur, 1949). However, it is likely that at least some host records of *R. pilosa* in the western Palearctic region belong to yet undescribed weevil species.

Using molecular techniques, Hernández-Vera et al. (2010) found that the species from the *R. pilosa* group have diverged from each other most likely before the early Miocene period. Even if only minor morphological differences can be found in the species of the *R. pilosa* group, all these taxa should be treated as very old and relict species, and thus should be expected to be very specific to the host plant with which they are associated. Hernández-Vera et al. (2010) also demonstrated the existence of highly specific host associations between the toadflax seed-feeding weevils in the *R. antirrhini* species complex, but species are very similar in appearance to one another.

b. Field Observations

In the Balkan Peninsula, *R. pilosa* has been found exclusively in galls on yellow toadflax. This strong preference for yellow toadflax has been confirmed at three stands of *Linaria genistifolia* and yellow toadflax occurring in the same location in Serbia in which *R. pilosa* was recorded exclusively on yellow toadflax. In total, 20 *Linaria* species and subspecies from 290 populations have been surveyed for *R. pilosa* galls. *Rhinusa pilosa* has been reared only from stem galls on yellow toadflax in Serbia (12 populations), Hungary (1 population), and in Romania (1 population).

c. Host Specificity Testing

Host specificity tests are tests to determine how many plant species *R. pilosa* attacks, and whether nontarget species may be at risk.

(1) Site of Quarantine Studies

Laboratory tests were conducted at the quarantine facilities located at Montana State University, Bozeman, Montana.

(2) Test Plant List

Test plant lists are usually developed on the basis of phylogenetic relationships between the target weed and other plant species (Wapshere 1974). It is generally assumed that plant species more closely related to the target weed species are at greater risk of attack than more distantly related species. The test plant list for *Linaria* spp. (Appendix 1) applied the phylogenetic approach, emphasizing: 1) species from the same genus (including synonyms) as the target weed; 2) species from the same tribe (Antirrhineae) as the target weed; and 3) plant species in different tribes but the same family (Plantaginaceae) as the target weed. The inclusion of closely related plant species identified as economically (=crop or ornamental) or environmentally important (especially threatened, endangered, or species of concern) was prioritized.

The *Linaria* test plant list also considered the biology, phenology, architecture, habitat, geographical distribution, and availability of plant species, along with the known host plants (or related species) of weevil species closely related to the potential candidate agent or of other toadflax biological control agents.

(3) Discussion of Host Specificity Testing

In host specificity testing (De Clerck-Floate et al., 2012), the percentage of eggs (as indicated by the number of oviposition marks) that develop to the adult stage is a good indicator of host plant suitability. The shape and size of galls induced on non-target plants, and tissue structure changes within the plant in response to the insect, are also good indicators of the plant species' suitability for larval development. Galls on the native range field host of *Rhinusa pilosa*, yellow toadflax, are more-or-less rounded in shape, and contain specialized tissues to nourish developing larvae. In contrast, galls induced on several other *Linaria* species and on native North American species in tribe Antirrhineae are irregular in shape; are not more than elongated, slight radial swellings of the stem; and usually do not allow normal larval development to the adult stage. These rudimentary galls display few cellular changes relative to what occurs with normal gall development on yellow toadflax, and importantly, may lack the nutritive tissue required to sustain larval development. In yet other plant species (e.g., some *Linaria* spp.), the tissue response can take the form of a defensive reaction whereby the plant quickly rejects the insect and its gall induction stimulus. See appendix 2 for a synopsis of no-choice gall induction tests.

Under no-choice conditions, all 11 non-target *Linaria* species exposed to *R. pilosa* were acceptable for oviposition. From those, nine species were suitable to varying degrees for gall and larval development. In Europe, field host records for *R. pilosa* reported in the literature need to be confirmed, because in most cases, no distinction was made between *R. pilosa* and the similar species *R. brondelii*. Of the 20 *Linaria* species and subspecies that have been field surveyed in Europe, the only demonstrated host of *R. pilosa* is yellow toadflax, and as previously mentioned, some *Linaria* species are unsuitable hosts for gall and larval development due to defensive reactions by the attacked plants. The restricted host range also is reflected in the reduced amount of adult feeding by *R. pilosa* on non-target *Linaria* species.

Oviposition was limited to four native North American species in no-choice tests of the 63 native North American species screened. Oviposition and gall development on native North American species was further reduced in choice experiments, with only three larvae developing to the adult stage on only one species, *Sairocarpus virga*, out of a large number of test replicates. The three native North American species that were suitable for oviposition or that supported gall production but no adult development under no-choice conditions (*Epixiphium wislizeni*, *Nuttallanthus canadensis*, and *Sairocarpus nuttallianus*) are thought to be at negligible risk from *R. pilosa* impact. The same is true for *S. virga*, which supported only minor adult development and was not negatively affected by *R. pilosa* galling during potted plant experiments, even when higher than normal attack rates were imposed on this native species. In addition, adult feeding and survival was limited or rare on native North American species in the tribe Antirrhineae. There are no North American threatened, endangered, and sensitive species in the tribe Antirrhineae.

Gall-inducing insects are highly host- and organ-specific organisms; they induce galls on only one plant species or on a closely related group of species (Dreger-Jauffret and Shorthouse, 1992). Host-range studies demonstrated that yellow toadflax is the most suitable host plant for *R. pilosa*. Choice tests with yellow toadflax and a few non- target species confirmed that *R. pilosa* clearly prefers to oviposit on yellow toadflax, which was also the most suitable host for successful gall and larval development. To date, *R. pilosa* has been associated in the field exclusively with yellow toadflax, and demonstrates the potential to exert significant impact on host weed populations if introduced where it will be free from regulation by native range natural enemies. See appendix 3 for host specificity testing methods.

2. Impact of *R. pilosa* on Yellow Toadflax

In studies determining the impact of *R. pilosa* on yellow toadflax, galled plants were 55 percent shorter than control plants, had 75 percent less

below-ground biomass, and produced fewer flowering stems in comparison to controls. Although there was no significant difference between the galled and control plants in the amount of total dried above-ground biomass produced, gall biomass constituted 40 percent of this total in galled plants, representing a significant loss to normal yellow toadflax growth productivity. No significant differences were detected in the number of new stems and lateral shoots produced during the experiment, suggesting that the galled plants were unable to compensate for their losses.

All yellow toadflax populations tested were susceptible to *R. pilosa*, and successfully produced fully developed galls and live adult weevils. The average number of galls produced per weevil-exposed plant in this low treatment density experiment was 4.5 ± 0.4 ($n = 45$), and there was no significant difference in susceptibility to galling among geographically-delineated host plant populations. However, not all populations of yellow toadflax tested responded the same in terms of correlates of fitness to low density treatments with *R. pilosa*. Plants from Tie Lake, British Columbia were more negatively affected by galling, with treated plants producing 16 percent less above-ground biomass, 41 percent less below-ground biomass, 39 percent fewer flowering stems, and 58 percent fewer new stems during the experiment, compared to control plants.

3. Uncertainties Regarding the Environmental Release of *R. pilosa*

Once a biological control agent such as *R. pilosa* is released into the environment and becomes established, there is a slight possibility that it could move from the target plants (yellow toadflax) to attack nontarget plants. Host shifts by introduced weed biological control agents to unrelated plants are rare (Pemberton, 2000). Native species that are closely related to the target species are the most likely to be attacked (Louda et al., 2003). If other plant species were to be attacked by *R. pilosa*, the resulting effects could be environmental impacts that may not be easily reversed. Biological control agents such as *R. pilosa* generally spread without intervention by man. In principle, therefore, release of this biological control agent at even one site must be considered equivalent to release over the entire area in which potential hosts occur, and in which the climate is suitable for reproduction and survival. However, significant non-target impacts on plant populations from previous releases of weed biological control agents are unusual (Suckling and Sforza, 2014).

In addition, this agent may not be successful in reducing yellow toadflax populations in the contiguous United States. Worldwide, biological weed control programs have had an overall success rate of 33 percent; success rates have been considerably higher for programs in individual countries (Culliney, 2005). Actual impacts on yellow toadflax by *R. pilosa* will not be known until after release occurs and post-release monitoring has been conducted. However, it is expected that *R. pilosa* will reduce the

vigor/competitiveness, population density, and spread of yellow toadflax, either alone or in concert with other compatible biological control agents or methods of control.

Managing yellow toadflax with classical biological control agents such as *R. pilosa* allows for a more gradual reduction in weed infestation in natural areas. If sufficient native plant propagules are available in the area, the gradual reduction in number and weakening of yellow toadflax plants through biological control will reduce the competitive edge of this invasive plant, and allow for the re-establishment of desirable native vegetation. However, replacement of yellow toadflax with another opportunistic and highly invasive weedy species such as cheatgrass or knapweed also is a possibility, thus vigilance in monitoring release sites for plant community changes post release will be important, and may necessitate the implementation of a reclamation plan.

4. Human Health

Rhinusa pilosa is a plant-feeding insect and poses no risk to humans. *Rhinusa pilosa* does not produce any defensive secretions, nor does it sequester host plant phytochemicals that may be hazardous to humans through direct contact. Reduction of yellow toadflax may result in a reduction in pollen that would be beneficial to those allergic to it.

5. Animal Health

Rhinusa pilosa is a plant-feeding insect and poses no risk to animal species. Reduction of yellow toadflax may be beneficial to cattle because they will not eat it unless there is nothing else to eat, and it contains toxic compounds.

6. Beneficial Uses

Rhinusa pilosa would reduce (but not eliminate) the presence of yellow toadflax in the environment. It may cause damage to ornamental plantings of yellow toadflax.

7. Cumulative Impacts

"Cumulative impacts are defined as the impact on the environment which results from the incremental impact of the action when added to other past, present and reasonably foreseeable future actions regardless of what agencies or person undertakes such other actions" (40 CFR 1508.7).

Other private and public concerns work to control yellow toadflax in invaded areas using available chemical, mechanical, cultural, and biological control methods. Release of *R. pilosa* is not expected to have any negative cumulative impacts in the contiguous United States because of its host specificity to yellow toadflax. Effective biological control of yellow toadflax will have beneficial effects for Federal, State, local, and private weed management programs, and may result in a long-term, non-damaging method to assist in the control of yellow toadflax.

8. Endangered Species Act

Section 7 of the Endangered Species Act (ESA) and ESA's implementing regulations require Federal agencies to ensure that their actions are not likely to jeopardize the continued existence of federally listed threatened and endangered species or result in the destruction or adverse modification of critical habitat.

There are three plants that are federally-listed or proposed for listing in the contiguous United States in the family Plantaginaceae, the same family as the target weed, yellow toadflax: parachute beardtongue (*Penstemon debilis*) (threatened) with critical habitat, blowout penstemon (*Penstemon haydenii*) (endangered), and Penland beardtongue (*Penstemon penlandii*) (endangered). Based on host specificity of *R. pilosa* reported in testing, field observations, and in the scientific literature, APHIS has determined that environmental release of *R. pilosa* may affect, but is not likely to adversely affect these plant species or their critical habitat. In addition, yellow toadflax may oocur in the habitat of the Karner blue butterfly (*Lycaeides melissa samuelis*), but is not reported as a nectar plant for it. Therefore, APHIS has determined that release of *R. pilosa* may affect, but is not likely to adversely affect the Karner blue butterfly. APHIS prepapred a biological assessment and requested concurrence from the U.S. Fish and Wildlife Service on these determinations, and received a concurrence letter dated December 16, 2016.

V. Other Issues

Consistent with Executive Order (EO) 12898, "Federal Actions to Address Environmental Justice in Minority Populations and Low-income Populations," APHIS considered the potential for disproportionately high and adverse human health or environmental effects on any minority populations and low-income populations. There are no adverse environmental or human health effects from the field release of *R. pilosa* and will not have disproportionate adverse effects to any minority or low-income populations.

Consistent with EO 13045, "Protection of Children from Environmental Health Risks and Safety Risks," APHIS considered the potential for disproportionately high and adverse environmental health and safety risks to children. No circumstances that would trigger the need for special environmental reviews are involved in implementing the preferred alternative. Therefore, it is expected that no disproportionate effects on children are anticipated as a consequence of the field release of *R. pilosa*.

EO 13175, "Consultation and Coordination with Indian Tribal Governments," was issued to ensure that there would be "meaningful consultation and collaboration with tribal officials in the development of Federal policies that have tribal implications…."

APHIS is consulting and collaborating with Indian tribal officials to ensure that they are well-informed and represented in policy and program decisions that may impact their agricultural interests in accordance with EO 13175.

VI. Agencies, Organizations, and Individuals Consulted

The Technical Advisory Group for the Biological Control Agents of Weeds (TAG) recommended the release of *R. pilosa* on August 30, 2013. TAG members that reviewed the release petition (De Clerck-Floate et al., 2012) included USDA representatives from National Institute of Food and Agriculture, Agricultural Research Service, Animal and Plant Health Inspection Service, and U.S. Forest Service; U.S. Department of Interior's Bureau of Reclamation; U.S. Army Corps of Engineers; and representatives from Mexico (SAGARPA-SENASICA-DGSV) and Agriculture and Agri-Food Canada.

This EA was prepared by personnel at APHIS, USDA-Forest Service, and Agriculture and Agri-Food Canada. The addresses of participating APHIS units, cooperators, and consultants follow.

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APPENDIX 1: Summary list of test plant species for determining host ranges of candidate *Linaria* biological control agents (De Clercke-Floate et al., 2012)

Genetic types of the target weed species

Scientific Name	Common Name	U.S. - State Distribution Canada - Province Distribution	Growth Habit	Duration	Nativity	Status
Linaria vulgaris USA, CANADA, EU	yellow or common toadflax		forb/herb	perennial	introduced	target weed
Linaria dalmatica USA, CANADA, EU	Dalmatian toadflax		forb/herb	perennial	introduced	target weed
Linaria genistifolia USA, CANADA,EU	broomleaved toadflax		forb/herb	perennial	introduced	target weed
Linaria vulgaris x L. dalmatica hybrids USA	hybrid toadflax		forb/herb	perennial	introduced	target weed

Species from the same genus (*Linaria*) as the target weed species

Scientific Name	Common Name	U.S. - State Distribution Canada - Province Distribution	Growth Habit	Duration	Nativity	Status
Linaria alpina (L.) Mill	Alpine toadflax		forb/herb	perennial		ornamental; previously attacked
L. angustissima (Loisel.) Borbás	Italian toadflax	MA	forb / herb	perennial	introduced	ornamental; previously attacked
L. maroccana Hook. f.	Moroccan toadflax	AZ, CA, CT, MA, ME, NH, NY, OR, VA, WV	forb/herb	annual	introduced	ornamental; previously attacked
L. repens (L.) Mill.	striped toadflax	CT, MA, ME, NJ, PA NB, NF, NS	forb / herb	annual	introduced	ornamental; previously attacked
L. saxatilis (L.) Chaz.			forb/herb	annual/ biennial		
L. supina (L.) Chaz.	lesser butter and eggs	CA, MA, NJ, OR, PA	forb / herb	annual	introduced	ornamental; previously attacked
L. purpurea (L.) Mill.	purple toadflax	CA, WA BC	forb/herb	perennial	introduced	ornamental

Species from other genera within the same family (Plantaginaceae) as the target weed species						
Species from the same tribe (Antirrhineae) as the target weed species						
Scientific Name	**Common Name**	**U.S. - State Distribution Canada - Province Distribution**	**Growth Habit**	**Duration**	**Nativity**	**Status**
Antirrhinum majus L.	garden snapdragon		forb/herb	annual/ perennial	introduced	ornamental; folk medicine; previously attacked; recorded host genus for *Mecinini* spp.
Chaenorhinum minus (L.) Lange	dwarf snapdragon	AL, AR, CO, CT, DC, DE, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MT, NC, ND, NE, NH, NJ, NY, OH, OK, OR, PA, RI, TN, TX, VA, VT, WA, WI, WV AB, BC, MB, NB, NL, NS, ON, PE, PQ, SK	forb/herb	annual	introduced	weed; previously attacked; recorded host genus for *Mecinini* spp.
Cymbalaria muralis Gaertn., Mey. & Scherb.	Kenilworth ivy		forb/herb	annual	introduced	ornamental; previously attacked
Epixiphium wislizeni (Engel. ex A. Gray) Munz	balloonbush	AZ, NM, TX	forb/herb/ vine	perennial	native	previously attacked
Galvezia juncea (Benth.) A. Gray	Baja bush snapdragon		shrub	perennial	native	
Gambelia speciosa Nutt	showy greenbright	CA	subshrub / shrub	perennial	native	
Howelliella ovata (Eastw.) Rothm.	ovateleaf snapdragon	CA	forb/herb	annual	native	
Kickxia elatine (L.) Dumort.	sharpleaf cancerwort	AL, AR, CA, CT,DC, DE, GA, IL,IN, KY, KS, LA, MA, MD, MI, MO, NC, NJ, NY, OH, OK, OR, PA,RI, SC, TN, TX, VA, WA, WI, WV BC, ON	forb/herb	annual	introduced	weed; previously attacked

Mabrya acerifolia (Pennell) Elisens	brittlestem	AZ	forb/herb/ vine	perennial	native	
Maurandella antirrhiniflora (Humb. & Bonpl. ex Willd.) Rothm.	roving sailor	AZ, CA, FL, MD, NM, NV,TX, UT	forb/herb/ vine	perennial	native	ornamental; folk medicine; previously attacked
Misopates orontium (L.) Raf.	linearleaf snapdragon	AK, CA, CT, FL, ID, IL, KY, ME, MI, NJ, NY, OH, OR, PA, UT, VA, WA NB, ON, PQ	forb/herb	annual	introduced	weed; previously attacked; recorded host genus for *Mecinini* spp.
Mohavea confertiflora (A. DC.) A. Heller	ghost flower	AZ, CA, NV	forb/herb	annual	native	
Neogaerrhinum strictum (Hook. & Arn.) Rothm.	Kellogg snapdragon	CA	forb/herb/ vine	annual	native	previously attacked
Nuttallanthus canadensis (L.) D.A. Sutton	Canada toadflax	AL, AR, CA, CT, DC, DE, FL, GA, IA, IL, IN, KS, LA, MA, MD, ME, MI, MN, MO, MS, NC, ND, NH, NJ, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, VA, VT, WA, WI, WV NB, NS, ON, PQ,	forb/hereb	annual/ biennial	native	TES (OH: endangered); previously attacked
Nuttallanthus texanus (Scheele) D.A. Sutton	Texas toadflax	AL, AR, AZ, CA, CO, FL, GA, IL, KS, KY, LA, MN, MO, MS, MT, NC, ND, NE, NM, OK, OR, SC, SD, TN, TX, UT, VA, WA, WY AB, BC, SK	forb/herb	annual/ biennial	native	previously attacked
Pseudorontium cyathiferum (Benth.) Rothm.	dog's-mouth	AZ, CA	forb/herb	annual	native	
Sairocarpus nuttallianus (Benth. ex A. DC.) D.A. Sutton	violet snapdragon	AZ, CA	forb/herb	annual/ biennial	native	
Sairocarpus virga (A. Gray) D.A. Sutton	tall snapdragon	CA	forb/herb	perennial	native	previously attacked
Sairocarpus multiflorus (Pennell) D.A. Sutton	Sierra snapdragon	CA	forb/herb	annual	native	

Species from tribes other than that of the target weed species						
Scientific Name	**Common Name**	**U.S. - State Distribution** **Canada - Province Distribution**	**Growth Habit**	**Duration**	**Nativity**	**Status**
Bacopa rotundifolia (Michx.) Wettst.	disk waterhyssop	AL, AR, AZ, CA,CO, IA, ID, IL, IN, KS, KY, LA, MD, MN, MO, MS, MT, NC, ND, NE, NM, OK, SD, TN, TX, VA, WI, WY AB	forb/herb	perennial	native	TES (IN: endangered)
Besseya wyomingensis (A. Nelson) Rydb.	Wyoming besseya	CO, ID, MT, NE, SD, UT, WY AB, BC, SK	forb/herb	perennial	native	
Chelone obliqua L.	red turtlehead	AL, AR, FL, GA, IA, IL, IN, KY, MA, MD, MI, MN, MO, MS, NC, SC, TN, VA	forb/herb	perennial	native	TES (MI: endangered; MD: threatened)
Collinsia parviflora Lindl.	maiden blue eyed Mary	AK, AZ, CA, CO, ID, MA, MI, MT, ND, NE, NM, NV, OR, PA, SD, UT, VT, WA, WY AB, BC, ON, MB, NS, PE, SK, YT	forb/herb	annual	native	TES (MI: threatened)
Digitalis purpurea L.	purple foxglove	AK, CA, CO, CT, ID, MA, MD, ME, MT, NH, NJ, NY, OH, OR, PA, UT, VT, WA, WI, WV, WY BC, NB, NL, NS, ON, PQ	forb/herb	perennial	introduced	economic
Gratiola neglecta Torr.	clammy hedgehyssop	AL, AR, AZ, CA, CO, CT, DC, DE, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NS, ON, QC, SK	forb/herb	annual	native	surrogate for *G. Aurea* Pursh TES (PA: endangered; MI: threatened)
Keckiella antirrhinoides (Benth.) Straw	snapdragon penstemon	AZ, CA, NV	shrub/ subshrub	perennial	native	

Keckiella breviflora (Lindl.) Straw	bush beardtongue	CA, NV	shrub/ subshrub	perennial	native	
Keckiella ternata (Torr. ex A. Gray) Straw	scarlet keckiella	CA	shrub/ subshrub	perennial	native	
Keckiella cordifolia (Benth.) Straw	heartleaf keckiella	CA	shrub/ subshrub	perennial	native	
Lindernia dubia (L.) Pennell	yellowseed false pimpernel	AL, AR, AZ, CA, CO, CT, DC, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, VA, VT, WA, WI, WV BC, NB, NS, ON, QC	forb/herb	annual/ biennial	native	
Penstemon centranthifolius (Benth.) Benth	scarlet bugler	CA	forb/herb/ subshrub	perennial	native	
Penstemon confertus Douglas ex Lindl.	yellow pemstemon	ID, MT, OR, WA AB, BC, SK	forb/herb/ subshrub	perennial	native	
Penstemon digitalis Nutt. ex. Sims	talus slope penstemon	AL, AR, CT, DC, DE, GA, IA, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, NC, ND, NE, NH, NJ, NY, OH, OK, PA, RI, SC, TN, TX, VA, VT, WI, WV NB, NS, ON, PE, PQ	forb/herb	perennial	native	TES (RI: special concern)
Penstemon grinnellii Eastw	Grinnell's beardtongue	CA	forb/herb/ subshrub	perennial	native	
Penstemon heterophyllus Lindl.	bunchleaf penstemon	CA	forb/herb/ subshrub	perennial	native	
Penstemon nitidus Douglas ex Benth.	waxleaf penstemon	CO, ID, MT, ND, SD, WY AB, BC, MB, SK	forb/herb/ subshrub	perennial	native	
Penstemon ovatus Douglas ex Hook.	eggleaf beardtongue	OR, WA BC	forb/herb/ subshrub	perennial	native	
Penstemon procerus Douglas ex Graham	littleflower penstemon	AK, CA, CO, ID, MT, ND, NV, OR, UT, WA, WY AB, BC, MB, SK, YT	forb/herb/ subshrub	perennial	native	
Penstemon spectabilis Thurb. ex A. Gray	showy penstemon	CA	forb/herb/ subshrub	perennial	native	

Plantago eriopoda Torr.	redwool plantain	AK, AZ, CA, CO, IA, ID, MN, MT, ND, NE, NM, NV, NY, OR, SD, UT, WA, WY AB, BC, MB, NT, PQ, SK, YT	forb/herb	perennial	native	TES (NY: threatened); recorded host genus for *Mecinini* spp.
Plantago lanceolata	narrowleaf plantain	AK, AL, AR, AZ, CA, CO, CT, DC, DE, FL, GA, HI, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NF, NS, ON, PE, QC, SK	forb/ herb	annual/ biennal/ perennial	introduced	Surrogate for *P. eriopoda* Torr.
Plantago major	common plantain	AK, AL, AR, AZ, CA, CO, CT, DC, DE, FL, GA, HI, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, LB, MB, NB, NF, NS, NT, ON, PE, QC, SK, YT	forb/ herb	perennial	introduced	Surrogate for *P. eriopoda* Torr.
Scoparia dulcis L.	licorice weed	AL, FL, GA, LA, MS, SC, TX	forb/herb/ subshrub	annual/ perennial	native	
Synthyris pinnatifida S. Watson	featherleaf kittentails	ID, MT, UT, WA, WY	forb/herb	perennial	native	TES (WA: sensitive)
Tonella floribunda A. Gray	manyflower tonella	ID, OR, WA	forb/herb	annual	native	Surrogate for *T. tenella* (Benth.) A. Heller TES (Canada : endangered)

Veronica americana Schwein. ex Benth.	American speedwell	AK, AR, AZ, CA, CO, CT, DE, IA, ID, IL, IN, KS, KY, MA, MD, ME, MI, MN, MO, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NL, NS, NT, ON, PE, PQ, SK, YT, SPM	forb/herb	perennial	native	recorded host genus for *Mecinini* spp.
Veronica chamaedrys L.	germander speedwell	AK, CA, CT, DC, ID, IL, IN, MA, MD, ME, MI, MO, MT, NC, NH, NJ, NY, OH, OR, PA, RI, VA, VT, WA, WI, WV AB, BC, NB, NF, NS, ON, PE, QC	forb/ herb	perennial	introduced	
Veronica hederifolia L.	ivyleaf speedwell	AL, AR, CA, CT, DC, DE, FL, GA, IL, IN, KS, KY, LA, MD, MO, NC, NE, NJ, NY, OH, OK, OR, PA, SC, SD, TN, UT, VA, WA, WV BC, ON	forb/ herb	annual	introduced	
Veronica officinalis L.	common gypsyweed	CA, CT, DC, DE, GA, IA, ID, IL, IN, KS, KY, MA, MD, ME, MI, MN, MT, NC, ND, NH, NJ, NY, OH, OR, PA, RI, SC, SD, TN, VA, VT, WA, WI, WV, WY BC, NB, NF, NS, ON, PE, QC	forb/ herb	perennial	introduced	
Veronica spicata L.	spiked speedwell	NY ON, PQ	forb/herb	perennial	introduced	economic; previously attacked; recorded host genus for *Mecinini* spp.
Veronicastrum virginicum (L.) Farw.	Culver's root	AL, AR, CT, DC, DE, FL, GA, IA, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, NC, ND, NE, NJ, NY, OH, OK, PA, SC, SD, TN, TX, VA, VT, WI, WV MB, NS, ON, PE	forb/herb	perennial	native	TES (VT: endangered; MA, NY: threatened)
Veronica wormskjoldii Roem. & Schult.	Alpine Speedwell	AK, AZ, CA, CO, ID, ME, MT, NH, NM, NV, OR, UT, WA, WY AB, BC, LB, NF, NT, NU, ON, QC, YT	forb/ herb	perennial	native	

Species from other families in the Lamiales order						
Scientific Name **FAMILY**	**Common Name**	**U.S. - State Distribution** **Canada - Province Distribution**	**Growth Habit**	**Duration**	**Nativity**	**Status**
Castilleja applegatei Fernald	wavyleaf Indian paintbrush	CA, ID, NV, OR, UT, WY	forb/herb/ subshrub	perennial	native	
Castilleja lineariifolia Benth. OROBANCHACEAE	Wyoming Indian paintbrush	AZ, CA, CO, ID, MT, NM, NV, OR, UT, WY	forb/herb/ subshrub	perennial	native	biochemical similarity (iridoid glycosides)
Castilleja minor (A. Gray) A. Gray	lesser Indian paintbrush	AZ, CA, CO, ID, MT, NM, NV, OR, UT, WA, WY BC	forb/ herb	annual / perennial	native	
Castilleja occidentalis Torr.	western Indian paintbrush	CO, ID, MT, NM, UT AB, BC	forb/herb/ subshrub	perennial	native	
Fraxinus pennsylvanica Marsh. OLEACEAE	green ash	AL, AR, CO, CT, DC, DE, FL, GA, IA, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NY, OH, OK, PA, RI, SC, SD, TN, TX, UT, VA, VT, WI, WV, WY AB, MB, NB, NS, ON, PE, QC, SK	tree	perennial	native	recorded host genus for *Mecinini* spp.; biochemical similarity (iridoid glycosides)
Mimulus guttatus DC. PHYRAMCEAE	seep monkeyflower	AK, AZ, CA, CO, CT, ID, MD, MI, MT, ND, NM, NV, NY, OR, PA, SD, UT, WA, WY AB, BC, NB, NT, SK, YT	forb/herb	perennial	native	biochemical similarity (iridoid glycosides)
Ruellia caroliniensis (J.F. Gmel.) Steud. ACANTHACEAE FAMILY	Carolina wild petunia	AL, DC, DE, FL, GA, IL, IN, KY, LA, MD, MS, NC, NJ, OH, OK, PA, SC, TN, TX, VA, WV	forb/herb	perennial	native	biochemical similarity (iridoid glycosides)
Ruellia strepens L.	limestone wild petunia	AL, AR, DC, DE, FL, GA, IA, IL, IN, KS, KY, LA, MD, MI, MO, MS, NC, NE, NJ, OH, OK, PA, SC, TN, TX, VA, WV	forb / herb	perennial	native	
Salvia azurea Michx. ex Lam. LAMIACEAE	azure blue sage	AL, AR, CO, CT, FL, GA, IA, IL, IN, KS, KY, LA, MI, MN, MO, MS, NC, NE, NM, NY, OH, OK, TN, TX, UT, WI	forb/herb	perennial	native	recorded host genus

Scrophularia californica Cham. & Schltdl	California figwort	CA	forb / herb	perennial	native	
Scrophularia lanceolata Pursh SCROPHULARIACEAE	lanceleaf figwort	CA, CO, CT, DE, IA, ID, IL, IN, KS, NC, MA, MD, ME, MI, MN, MO, MT, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SD, UT, VA, VT, WA, WI, WV, WY AB, BC, ON, NB, NS, PE, PQ, SK	forb/herb	perennial	native	recorded host genus for *Mecinini* spp.; biochemical similarity (iridoid glycosides)
Teucrium canadense L. LAMIACEAE	Canada germander	AL, AR, AZ, CA, CO, CT, DC, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY BC, MB, NB, NS, ON, PE, PQ, SK	forb/herb	perennial	native	recorded host genus for *Eteobalea beata* species group
Trichostema lanatum Benth. LAMIACEAE	wooly bluecurls	CA	subshrub/ shrub	perennial	native	biochemical similarity (iridoid glycosides)
Verbascum nigrum L.	black mullein	IL, MA, MN, NH, PA, WI AB, ON	forb/ herb	perennial	introduced	
Verbascum thapsus L.	common mullein	AK, AL, AR, AZ, CA, CO, CT, DC, DE, FL, GA, HI, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NF, NS, ON, PE, QC, SK	forb/ herb	biennal	introduced	

Verbascum virgatum Stokes SCROPHULARIACEAE	wand mullein	AL, AZ, CA, FL, GA, HI, ID, IL, IN, LA, NC, NM, NV, NY, OH, PA, SC, TX, UT NS, ON,PE, PQ	forb/herb	biennial	introduced	ornamental; recorded host genus for *Mecinini* spp.; biochemical similarity (iridoid glycosides)
Verbena lasiostachys Link (syn. *V. prostrata*) VERBENACEAE	western vervain	CA, NY, OR	forb/herb	perennial	native	biochemical similarity (iridoid glycosides)
Species from other orders with shared morphological or biochemical characteristics to the target weed species or on which a biocontrol agent or its close relatives have been recorded to feed and/or reproduce						

Scientific Name **FAMILY**	**Common Name**	**U.S. - State Distribution** **Canada - Province Distribution**	**Growth Habit**	**Duration**	**Nativity**	**Status**
Campanula rotundifolia L CAMPANULACEAE	bluebell bellflower	AK, AZ, CA, CO, CT, DE, IA, ID, IL, IN, MA, MD, ME, MI, MN, MO, MT, NC, ND, NE, NH, NJ, NM, NY, OH, OR, PA, RI, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NL, NS, NT, NU, ON, PE, PQ, SK, YT, SPM *also* GREENLAND	forb/herb	perennial	native	recorded host genus for *Mecinini* spp.
Comandra umbellata (L.) Nutt. SANTALACEAE	bastard toadflax	AK, AL, AR, AZ, CA, CO, CT, DC, DE, GA, IA, ID, IL, IN, KS, KY, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY AB, BC, MB, NB, NL, NS, NT, ON, PE, PQ, SK, YT, SPM	forb/herb/ subshrub	perennial	native	morphological similarity
Gentiana affinis Griseb. GENTIANACEAE	pleated gentian	AZ, CA, CO, ID, MN, MT, ND, NM, NV, OR, SD, TX, UT, WA, WY AB, BC, MB, NT, SK	forb/herb	perennial	native	biochemical similarity (iridoid glycosides);

APPENDIX 2: Synopsis of no-choice gall induction tests with *Rhinusa pilosa* in 2006–11 (De Clercke-Floate et al., 2012)

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No. of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
Genetic types of the target weed species										
Linaria vulgaris (L.) Mill. EU (control)	26	152	1223	319	2.1±1.6 (0-11)	922	6.1±5.1 (0-34)	2.9±2.1 (0-12)	85.3	75.4
L. vulgaris (NA pop.s; pooled)	42	231	2341	597	2.6±1.7 (0-11)	1701	7.4±5.6 (0-40)	2.8±2.0 (0-12)	89.6	72.7
L. vulgaris (NA pop. No. 1)	5	34	289	66	1.9±1.4 (0-6)	225	6.6±5.6 (0-22)	3.4±2.7 (0-12)	86.4	77.9
L. vulgaris (NA pop. No. 2)	5	31	268	91	2.9±1.5 (1-7)	199	6.4±3.4 (1-15)	2.2±1.5 (0-8)	89.0	74.3
L. vulgaris (NA pop. No. 3)	5	32	268	72	2.2±1.5 (1-7)	209	6.5±3.4 (2-19)	2.9±1.7 (0-7)	93.1	78.0
L. vulgaris (NA pop. No. 7)	5	30	260	70	2.3±1.5 (0-6)	200	6.7±4.7 (0-15)	2.9±2.1 (0-12)	87.1	76.9
L. vulgaris (NA pop. No. 8)	5	35	303	89	2.5±1.4 (0-5)	212	6.2±3.3 (0-13)	2.4±1.6 (0-7)	86.5	70.0
Species from the same genus (*Linaria*) as the target weed species										
L. alpina (L.) Mill.	10	45	552	149	3.3±3.1 (0-11)	113	2.5±3.8 (0-19)	0.8±1.2 (0-6)	39.6	20.5
L. angustissima (Loisel.) Borbás	3	9	65	20	2.2±1.3 (1-5)	19	2.1±1.9 (0-5)	1±1.3 (0-5)	50	29.2
Linaria dalmatica (L.) Mill. (NA pop.s; pooled)	20	99	604 (238)	147	1.5±1.8 (0-6)	184	1.9±2.9 (0-13)	1.2±1.3 (0-7)	63.2	30.5
L. dalmatica (NA pop. No. 1)	1	4	24 (19 HR)	3	0.8±1.5 (0-3)	1	0.3±0.5 (0-1)	0.3±0.6 (0-1)	33.3	4.2
L. dalmatica (NA pop. No. 2)	1	4	32 (16 HR)	5	1.2±2.5 (0-5)	12	3±6 (0-12)	2.4±2.6 (1-7)	100	37.5
L. dalmatica (NA pop. No. 7)	4	20	119 (80 HR)	12	0.6±1.0 (0-4)	26	1.3±2.8 (0-10)	2.2±1.8 (0-6)	66.7	21.8
L. dalmatica (NA pop. No. 9)	1	7	46 (40 HR)	1	0.1±0.4 (0-1)	5	0.7±1.9 (0-5)	5	100	10.9
L. dalmatica (NA pop. No. 11)	1	7	37 (23 HR)	8	1.1±1.6 (1-4)	7	1±1.4 (0-3)	0.9±0.8 (0-2)	62.5	18.9

Plant species[a]	**No of females tested**	**No. of replicates**	**No of oviposition marks**[b]	**No. of galls induced**	**Mean no. of galls ±SD /replicate (range)**	**No of adults emerged**	**Mean no. of adults ±SD /replicate (range)**	**Mean no. of adults ±SD /gall (range)**	**% of galls supporting development to adult**	**% of oviposition marks resulting in adult production**
L. dalmatica (L.) Mill. ssp. *dalmatica* EU	3	14	74 (45 HR)	2	0.1±0.4 (0-1)	6	0.4±1.1 (0-3)	3	100	8.1
L. dalmatica (L.) Mill. ssp. *macedonica* EU	5	20	262 (19 HR)	74	3.7±4.5 (0-13)	41	2.1±4.5 (0-17)	0.6±1 (0-4)	29.7	15.6
L. genistifolia (L.) Mill. EU	18	93	594 (476 HR)	20	0.2±0.7 (0-5)	37	0.4±1.4 (0-7)	1.8±2.0 (0-6)	65.0	6.2
L. genistifolia (L.) Mill. ssp. *confertiflora* EU	5	18	64	17	0.9±1.2 (0-3)	0	-	-	-	-
L. genistifolia (L.) Mill. ssp. *artvinensis* EU	3	9	110	38	4.2±3.2 (2-11)	66	7.3±7.7 (0-22)	1.7±1.2 (0-6)	84.2	60
L. kurdica Boiss. & Hohen.[c]	2	8	65 (62 HR)	22	2.8±1.2 (1-5)	0	-	-	-	-
L. maroccana Hook. f.	9	49	126	38	0.8±1.2 (0-4)	0	-	-	0	0
L. pancicii Janka ex Nyman[c]	5	19	159 (24 HR)	48	2.5±2.3 (0-8)	96	5.0±5.1 (0-16)	2.0±1.3 (0-4)	89.6	60.4
L. purpurea (L.) Mill.	12	42	265 (23 HR)	66	1.6±2.2 (0-7)	68	1.6±3.4 (0-17)	1.0±1.2 (0-5)	53	25.7
L. repens (L.) Mill.	9	30	21	7	0.2±0.7 (0-3)	1	0.03±0.2 (0-1)	0.1±0.4 (0-1)	14.3	4.8
L. rubioides Vis. & Pančić[c] (*macedonian* phenotype)	3	9	98	35	3.9±0.9 (3-5)	64	7.1±2.7 (4-13)	1.8±1.0 (0-4)	88.6	65.3
L. rubioides Vis. & Pančić ssp. *nissana*[c]	10	30	538	140	4.7±4.0 (0-16)	104	3.5±5.4 (0-19)	0.7±1.4 (0-13)	41.4	19.3
L. saxatilis (L.) Chaz.	8	27	188	65	2.4±2.6 (0-9)	24	0.9±3.3 (0-17)	0.4±0.8 (0-3)	23.1	12.8
L. supina (L.) Chaz.	9	33	484	134	4.1±3.9 (0-14)	79	2.4±3.6 (0-15)	0.6±0.8 (0-5)	43.3	16.3
Species from the same tribe (Antirrhineae) as the target weed species										
Species from the same clade (Antirrhinum) as the target weed species										
Antirrhinum majus L.	9	19	0	0	-	-	-	-	-	-
Chaenorhinum minus (L.) Lange	10	16	0	0	-	-	-	-	-	-

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
Misopates orontium (L.) Raf.	9	25	0	0	-	-	-	-	-	-
Mohavea confertiflora (A. DC.) A. Heller *	5	5	0	0	-	-	-	-	-	-
Species from other clades than that of the target weed species										
Cymbalaria muralis G. Gaertn., B. Mey. & Scherb.	10	16	0	0	-	-	-	-	-	-
Epixiphium wislizeni (Engelm. ex A. Gray) Munz *	10	35	99	0	-	-	-	-	-	0
Galvezia juncea (Benth.) A. Gray. *	5	10	0	0	-	-	-	-	-	-
Gambelia speciosa Nutt. *	5	10	0	0	-	-	-	-	-	-
Kickxia elatine (L.) Dumort.	5	5	0	0	-	-	-	-	-	-
Mabrya acerifolia (Pennell) Elisens *	5	5	0	0	-	-	-	-	-	-
Maurandella antirrhiniflora (Humb. & Bonpl. ex Willd.) Rothm. *	10	31	0	0	-	-	-	-	-	-
Neogaerrhinum strictum (Hook. & Arn.) Rothm. *	5	13	0	0	-	-	-	-	-	-
Nuttallanthus canadensis (L.) D.A. Sutton *	24	97	17	13	0.1±0.5 (0-3)	0	-	-	0	0
Sairocarpus multiflorus (Pennell) D.A. Sutton *	15	38	0	0	-	0	-	-	-	0
S. nuttallianus (Benth. ex A. DC.) D.A. Sutton *	29	131	1240	148	1.1±1.1 (0-6)	0	-	-	0	0
S. virga (A. Gray) D.A. Sutton *	53	242	2120	248	1.2±1.4 (0-8)	3	0.01±0.1 (0-1)	0.01±0.1 (0-1)	1.2	0.1
Species from tribes other than that of the target weed species										
Besseya wyomingensis (A. Nelson) Rydb. *	5	5	0	0	-	-	-	-	-	-
Chelone obliqua L.*	5	5	0	0	-	-	-	-	-	-
Collinsia bicolor Benth. (syn. *C. heterophylla*) *	9	27	0	0	-	-	-	-	-	-

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
C. parviflora Lindl. *[c]	6	6	0	0	-	-	-	-	-	-
Digitalis purpurea L.	5	5	0	0	-	-	-	-	-	-
Keckiella antirrhinoides (Benth.) Straw *	7	18	0	0	-	-	-	-	-	-
K. breviflora (Lindl.) Straw *	5	14	0	0	-	-	-	-	-	-
K. cordifolia (Benth.) Straw *	5	12	0	0	-	-	-	-	-	-
K. ternata (Torr. ex A. Gray) Straw *	5	11	0	0	-	-	-	-	-	-
Penstemon centranthifolius (Benth.) Benth. *	5	8	0	0	-	-	-	-	-	-
P. confertus Douglas ex Lindl. *	5	5	0	0	-	-	-	-	-	-
P. digitalis Nutt. ex Sims *	12	31	0	0	-	-	-	-	-	-
P. eriantherus Pursh *[c]	2	4	0	0	-	-	-	-	-	-
P. glaber Pursh *[c]	3	7	0	0	-	-	-	-	-	-
P. grinnellii Eastw.*	5	9	0	0	-	-	-	-	-	-
P. laricifolius Hook. & Arn. *[c]	3	9	0	0	-	-	-	-	-	-
P. nitidus Douglas ex Benth. *	5	5	0	0	-	-	-	-	-	-
P. ovatus Douglas ex Hook. *	13	27	0	0	-	-	-	-	-	-
P. paysoniorum D.D. Keck *[c]	2	10	0	0	-	-	-	-	-	-
P. procerus Douglas ex Graham *	14	32	0	0	-	-	-	-	-	-
P. spectabilis Thurb. ex A. Gray *	5	7	0	0	-	-	-	-	-	-
Plantago eriopoda Torr.*	5	5	0	0	-	-	-	-	-	-

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
P. lanceolata L.	5	5	0	0	-	-	-	-	-	-
P. major L.	5	5	0	0	-	-	-	-	-	-
Scoparia dulcis L.*	5	5	0	0	-	-	-	-	-	-
Synthyris pinnatifida S. Watson *	10	10	0	0	-	-	-	-	-	-
Veronica americana Schwein. ex Benth. *	5	5	0	0	-	-	-	-	-	-
V. chamaedrys L.	5	5	0	0	-	-	-	-	-	-
V. hederifolia L.	5	5	0	0	-	-	-	-	-	-
V. officinalis L.	5	5	0	0	-	-	-	-	-	-
V. spicata L.	5	5	0	0	-	-	-	-	-	-
V. wormskjoldii Roem. & Schult.*	4	20	0	0	-	-	-	-	-	-
Veronicastrum virginicum (L.) Farw. *	5	5	0	0	-	-	-	-	-	-
Species from other families in the Lamiales order										
Ajuga chamaepitys (L.) Schreb. [c] LAMIACEAE	5	5	0	0	-	-	-	-	-	-
Castilleja applegatei Fernald * OROBANCHACEAE	4	4	0	0	-	-	-	-	-	-
C. linearifolia (Benth.) T.I. Chuang & Heckard *	5	5	0	0	-	-	-	-	-	-
C. minor (A. Gray) A. Gray *	5	5	0	0	-	-	-	-	-	-
C. occidentalis Torr *	4	4	0	0	-	-	-	-	-	-
C. sp. (*miniata* ?) Douglas ex Hook. ssp. miniata *[c]	5	5	0	0	-	-	-	-	-	-
Fraxinus pennsylvanica Marsh. * OLEACEAE	5	5	0	0	-	-	-	-	-	-

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
Mimulus aurantiacus W. Curtis *[c] PHYRAMCEAE	3	5	0	0	-	-	-	-	-	-
M. brevipes Benth *[c]	2	3	0	0	-	-	-	-	-	-
M. guttatus DC. *	6	18	0	0	-	-	-	-	-	-
M. lewisii Pursh *[c]	3	15	0	0	-	-	-	-	-	-
M. longiflorus Nutt. *[c]	3	5	0	0	-	-	-	-	-	-
M. puniceusi Nutt. *[c]	3	4	0	0	-	-	-	-	-	-
Ruellia caroliniensis (J.F. Gmel.) Steud. * ACANTHACEAE	5	5	0	0	-	-	-	-	-	-
R. strepens L.*	5	5	0	0	-	-	-	-	-	-
Salvia azurea Michx. ex Lam. * LAMIACEAE	5	5	0	0	-	-	-	-	-	-
Scrophularia californica Cham. & Schltdl. * SCROPHULARIACEAE	5	5	0	0	-	-	-	-	-	-
S. lanceolata Pursh *	5	5	0	0	-	-	-	-	-	-
Teucrium canadense L. * LAMIACEAE	5	5	0	0	-	-	-	-	-	-
Trichostema lanatum Benth. *	5	5	0	0	-	-	-	-	-	-
Verbascum nigrum L. SCROPHULARIACEAE	5	5	0	0	-	-	-	-	-	-
V. thapsus L.	5	5	0	0	-	-	-	-	-	-
V. virgatum Stokes	5	5	0	0	-	-	-	-	-	-
Verbena lasiostachys Link * VERBENACEAE	5	5	0	0	-	-	-	-	-	-
Species from other orders with shared morphological or biochemical characteristics to the target weed species or on which a biocontrol agent or its close relatives have been recorded to feed and/or reproduce										
Campanula rotundifolia L. * CAMPALUNACEAE	5	5	0	0	-	-	-	-	-	-

Plant species[a]	No of females tested	No. of replicates	No of oviposition marks[b]	No. of galls induced	Mean no. of galls ±SD /replicate (range)	No of adults emerged	Mean no. of adults ±SD /replicate (range)	Mean no. of adults ±SD /gall (range)	% of galls supporting development to adult	% of oviposition marks resulting in adult production
Gentiana cruciata L. [c] GENTIANACEAE	5	5	0	0	-	-	-	-	-	-
Symphoricarpos albus (L.) S.F. Blake * CAPRIFOLIACEAE	5	5	0	0	-	-	-	-	-	-
Valeriana edulis Nutt. ex Torr. & A. Gray * VALERIANACEAE	5	5	0	0	-	-	-	-	-	-
Viburnum opulus L. var. *opulus* CAPRIFOLIACEAE	5	5	0	0	-	-	-	-	-	-
Other families and crops										
Centaurea jacea L. [c] ASTERACEAE	5	5	0	0	-	-	-	-	-	-
Cirsium arvense (L.) Scop. [c]	5	5	0	0	-	-	-	-	-	-
Lactuca sativa L.[c]	5	5	0	0	-	-	-	-	-	-
Trifolium repens L. [c] FABACEAE	5	5	0	0	-	-	-	-	-	-
Triticum sativum L. [c] POACEAE	5	5	0	0	-	-	-	-	-	-
Zea mays L. [c]	5	5	0	0	-	-	-	-	-	-

* = species native to North America

[a] EU = European population
vulgaris No. 1 = Nisku, Alberta, Canada
vulgaris No. 2 = Komloops, British Columbia (BC), Canada
vulgaris No. 3 = Sweet Grass Co., Montana, USA *vulgaris* No. 7 = Boulder, Colorado, USA
vulgaris No. 8 = Edmonton, Alberta, Canada
dalmatica No. 1 = Ft. Macleod area, Alberta, Canada
dalmatica No. 2 = Komloops, BC, Canada
dalmatica No. 7 = Grand Forks, BC, Canada
dalmatica No. 9 = East Helena, Clarck Co., Montana, USA
dalmatica No. 11 = Ft. Macleod, Alberta, Canada

[b] HSR: hypersensitive stem reaction [c] Species not in the test plant list

Appendix 3. Host-specificity testing methods (De Clerck-Floate et al. 2012)

No-choice Adult Feeding and Survival Tests

Feeding by adult *R. pilosa* on non-target plants was evaluated in 2003-2005 under no-choice conditions. Feeding was assessed in early summer with naïve adults extracted from galls obtained in the gall induction tests ("ex-gall adults") and in early spring with reproductive adults that had overwintered in field cages ("post-hibernated adults"), in the garden at IZBIS, Zemun (Belgrade), Serbia.

No-choice adult feeding with ex-gall adults:
Methods. In 2003, cut stems of 24 different test plant species were each exposed to four adult weevils in unreplicated tests until feeding ceased. Feeding was assessed visually using a semi-quantitative method that ranked the amount of feeding relative to what was observed on the control plant (European *Linaria vulgaris*) within the test (i.e., maximum, moderate, slight, nibbling or none). Adult survival was recorded at the end of the test period as a percentage of the four weevils on each test plant.

No-choice adult feeding with post-hibernated adults:
Methods. In 2004 and 2005, cut stems of 12 different test plant species were each exposed to four post-hibernated (reproductive) adult weevils in unreplicated tests until feeding ceased. As in 2003, feeding was assessed visually and ranked according to deviation from injury observed on the control plant (European *Linaria vulgaris*). Adult survival was recorded at the end of the test period as a percentage of the four weevils on each test plant.

No-choice Oviposition, Gall Induction and Larval Development Tests

Methods. Potted plant material, not cut stems, was used for all oviposition and larval development no-choice tests to ensure that test conditions were as optimal as possible for a stem gall-forming insect, which requires *in situ*, growing plant tissues for normal gall induction to occur. Individually potted test plants were caged using a capped, ventilated plastic cylinder (10 x 35 cm). The soil surface was covered with coarse sand to prevent buildup of excessive moisture inside cages and to make the weevils more visible.

No-choice gall and larval development tests were conducted between 2006 and 2011 at IZBIS in Zemun (Belgrade), Serbia, and in the quarantine facility at AAFC, Lethbridge, Alberta, Canada. Individual potted plants were exposed for 1-4 days, depending on plant size, to one newly emerged post-hibernated and mated female. Only egg-laying females were used in this study, which was confirmed by observing each female weevil ovipositing on a *Linaria vulgaris* plant prior to being added to caged test plants. All study plants were retained under optimal indoor growing conditions favourable to normal gall development. Each potted plant was covered with a gauze bag at the appropriate time, as dictated by weevil phenology, to ensure retention of emerging F1 adults.

All plants with galls were examined under a stereo microscope and the number of oviposition marks visible on the galls was recorded. All galls from which no adults had emerged were dissected and gall contents inventoried. Plants that had deteriorated in quality for reasons other than exposure to *Rhinusa pilosa*, or those that failed to form typical galls were also dissected, and

the number of eggs and/or larval instars, if present, was recorded. Each plant with its separate female *R. pilosa* was considered a replicate within each test, and replicate number per plant species per test was 5-10. A total of 113 plant species or populations were included in the no-choice gall induction and larval development tests; 63 species, across 30 plant genera, were native to North America.

Sequential No-choice Tests

Sequential no-choice test, Sairocarpus virga *to* Linaria vulgaris:
Methods. Ten female-male *Rhinusa pilosa* pairs were individually retained in a cylinder cage (10 x 35 cm) on a potted *Sairocarpus virga* plant for 24 hours (ca. 10 replicate plants sequentially presented to each weevil pair). The same 10 pairs of *R. pilosa* were then exposed to *Linaria vulgaris* for the same period of time (ca. 10 replicate plants sequentially presented to each weevil pair). Each individual plant was exposed only once to any *R. pilosa* pair. The test was set up on April 20, 2006 and continued until all females had died. The number of oviposition marks visible on induced galls was recorded in early June.

Single-choice Oviposition, Gall Induction and Larval Development Tests

Few plant species were accepted for oviposition in these single-choice tests, so gall induction and larval development were negligible. Regardless, the results are presented below for single-choice tests conducted in 2005-2007.

Single-choice field cage test, *Linaria vulgaris* vs. *Linaria genistifolia* (2005):
Methods. Five *Rhinusa pilosa* male: female pairs were released into a field cage (210 x 210 x 190 cm) set up with 40 potted plants each of *Linaria vulgaris* and *L. genistifolia*. The plants within the cage were subsequently regularly inspected and any instances of gall development were recorded. All potted galled plants were removed from cages approximately 2.5 months after set-up and transferred into a greenhouse to follow gall and larval development. Each pot was covered with a gauze bag so that adult emergence could be recorded separately.

Single-choice field cage test *Linaria vulgaris* vs. *Linaria genistifolia* (2006):
Methods. Six *Rhinusa pilosa* pairs were released into a field cage (210 x 210 x 190 cm) set up with eight potted plants each of *Linaria vulgaris* and *L. genistifolia*. The plants within the cages were subsequently inspected on a weekly basis and gall development recorded. All potted galled plants were removed from the cage approximately 2 months after set up and transferred to a greenhouse to follow gall and larval development. Each pot was covered with a gauze bag so that adult emergence could be recorded separately.

Single-choice field cage test, *Linaria vulgaris* vs. *Sairocarpus virga* (2005):
Methods. Five *Rhinusa pilosa* pairs were released into a field cage (210 x 210 x 190 cm) set up with 12 potted plants each of *Linaria vulgaris* and *Sairocarpus virga*, and left for approximately 2.5 months. Gall development and adult emergence was recorded as described previously.

Single-choice test *Linaria vulgaris* vs. *Sairocarpus virga* (2006):
Methods. One *Rhinusa pilosa* female-male pair was exposed to two *Linaria vulgaris* and two

Sairocarpus virga plants growing together in a large 15 cm diameter pot covered with a gauze sleeve. Weevil pairs were transferred to new plants every 2-3 days until all the females died. The weevils were always placed on the soil surface to allow them to freely orient toward any plant within the cage. The number of galls, oviposition marks and emerging adults was recorded as described previously.

Single-choice field cage test, *Linaria vulgaris* vs. *Sairocarpus nuttallianus* (2005):
Methods. Five *Rhinusa pilosa* pairs were released into a field cage (210 x 210 x 190 cm) set up with eight potted plants each of *L. vulgaris* and *S. nuttallianus*, and left for approximately 2.5 months. Gall development and adult emergence were recorded as described previously.

Multiple-choice Oviposition, Gall Induction and Larval Development Tests

Multiple-choice field cage test, *Linaria vulgaris* vs. *Sairocarpus virga* vs. *S. nuttallianus* (2007):

Methods. Six pairs of *Rhinusa pilosa* were released into each of two field cages (210 × 210 × 190 cm), both set up with six each of potted *Sairocarpus virga* and *S. nuttallianus*, and 24 potted *L. vulgaris*, and were left for approximately 2 months. The design of the experiment was meant to simulate field conditions in which target hosts are typically more abundant than nontarget hosts, thus allowing the females to express their preference for ovipositing on shoots without galls. Gall development and adult emergence were recorded as described previously.

Impact of Gall Development on *Sairocarpus virga*

Two impact experiments were established with the sole North American species that allowed larval development to the adult stage, *S. virga.*

Impact of gall development on biomass and growth of Sairocarpus virga:
Methods. Paired plants of the same size were selected, with one randomly assigned to receive one mated and ovipositing *Rhinusa pilosa* female while the other was used as a control. In total, 30 *Sairocarpus virga* plants were exposed for 24 hours to oviposition using a total of 12 mated females. All plants were assessed 3 months later.

Impact of gall development on stem growth and flowering of Sairocarpus virga:
Methods. Twelve individually potted, well-developed single stem *Sairocarpus virga* plants of the same height (20 cm), each covered with a plastic cylinder (10 × 35 cm), were exposed to one mated *Rhinusa pilosa* female until oviposition was observed. Another 12 plants of similar size, and from the same cohort as the treatment plants, were used as controls. Treatment and control plants were randomly assigned at experiment set-up. All plants were retained under the same environmental conditions then assessed 4 months later.

Appendix 4. Release Protocol and Post-Release Monitoring Plan for *Rhinusa pilosa* (De Clerck-Floate et al., 2012).

Release Protocol

Methods to ensure pure cultures and correct identification of agent to be released:
The source population for potential releases of *Rhinusa pilosa* will come from northern Serbia via CABI Europe-Switzerland. The weevil's identity and colony purity will be ensured by:

-Identification by Italian weevil systematist, Roberto Caldara, who together with Ivo Toševski (CABI-Europe Serbia), André Gassmann (CABI-Europe Switzerland) and other colleagues described the species through characterization of its morphology, biology and genetics (i.e., molecular analysis) (Caldara et al., 2008).

-Taking adults for shipment to North America from the same colonies established and used by Ivo Toševski in northern Serbia at IZBIS, Zemun (Belgrade), Serbia for host specificity testing.

-Depositing voucher specimens of *R. pilosa* from the Serbian source population in the Canadian National Collection (AAFC, Ottawa, Ontario) ahead of importations of living weevils, thereby preparing for taxonomist verification of each *R. pilosa* shipment sent to the United States.

General release protocol to ensure absence of natural enemies and cryptic or sibling species:

The source Serbian colony will be of known identity and will be expected to be free of natural enemies. However as an extra precaution, the shipped weevils will be reared through at least one generation on its known host plant species (*Linaria vulgaris*) in regulator-inspected and certified quarantine facilities in Canada (AAFC, Lethbridge Research Centre, Insect Microbial Containment Facility, Lethbridge, Alberta) and the United States (Montana State University Quarantine, Bozeman, Montana). During rearing in respective quarantine facilities, insects will be securely caged and inspected frequently for emergence of parasitoids, inquilines, or other unwanted organisms, and for disease symptoms that may indicate the presence of entomopathogens. Unwanted arthropods or infected *R. pilosa* will be immediately removed and destroyed through submersion in ethyl alcohol or autoclaving.

Voucher specimens of *Rhinusa pilosa* will also be available in containment at Bozeman, MT for on site verifications of identity by entomologists familiar with the species. Any cryptic or sibling species are expected to be host-associated (Caldera et al., 2008); thus, their likelihood of accidental release would be reduced through rearing the weevils on its known host in containment, and through closely watching for any abnormal insect behaviour or anomalies in gall development (e.g., atypical position of galls on host stems, or non-spherical and asymmetrical gall morphology, both which are accurately diagnositic of gall induction on a novel host). Any suspicious insects can be collected, preserved and genetically characterized with the help of project collaborators ahead of release.

Specific location of rearing or culturing facilities:

The source insects will come from a field plot and greenhouse reared colony in northern Serbia at IZBIS, Zemun (Belgrade). Shipped *R. pilosa* will be received and reared in quarantine facilities at the Montana State University Quarantine, Bozeman, Montana.

Intended sites for initial release, timing of release, and methods to be used:

Initial releases are planned for an outdoor nursery (caged plots of yellow toadflax) within a large chain-link fenced garden study area at the USDA-Forest Service, Rocky Mountain Research Station, Forestry Sciences Laboratory on the campus of Montana State University in Bozeman, Montana, and in secure sites on Forest Service and privately-owned lands in west central Montana, also where establishment can be closely monitored.

Releases will be made of spring-emerged adults, timed to coincide with the growth phenology of *Linaria vulgaris* shoots at the chosen sites. *Rhinusa pilosa* females prefer actively-growing shoots that have emerged approximately 10 cm above ground in the early spring (April-May). Both female and male weevils will be released at the same time in either caged or open situations. The number of adults released per site will depend on the availability of lab-reared weevils, but if possible, initial releases will be made in densities of approximately 100 adults/site. Experiments during the initial release stage are being planned to determine optimum release strategies for increasing establishment success.

Post-Release Monitoring

Anticipated time of initial releases:

The anticipated time of initial releases will be early spring (April-May) and will be dependent on the phenological development of *Linaria vulgaris* at chosen release sites. Young, actively-growing shoots should be approximately 10 centimeters above ground for optimum acceptance by female weevils and gall induction.

Groups performing monitoring:

Field monitoring will be initially conducted, and monitoring methods for subsequent use by state collaborators (e.g., Colorado Department of Agriculture) will be designed by classical weed biological control researchers (permittee) involved with the release and study of *Rhinusa pilosa* in the United States. Monitoring data will be assessed by the researchers to determine establishment, dispersal and impact of *R. pilosa*, and to develop optimum strategies for the successful use of the agent by collaborators.

Monitoring techniques:

Initial monitoring will be for establishment, local spread, and impact of *Rhinusa pilosa*:

Establishment: Monitoring will take place yearly in mid-late summer (June-August) for galls and in spring (April-May) for emerged adults, for up to 3-4 years after initial releases to ascertain establishment. To generally determine presence of the insect, release patches of toadflax will be carefully inspected during a slow walk-through (e.g., minimum of 30 minutes to 1 hour depending on patch size) by two or more people familiar with the insect and its galls. Gall and/or adult density will be determined by randomly selecting 30-100 *Linaria vulgaris* stems within a known area (e.g., within a caged plot) or systematically along transects laid through a release patch (e.g., every 1 meter), and the number of galls or encountered adults recorded. Other data, such as position of the galls within shoots, or gall size may be recorded at this time. A subsample of galls will be collected and dissected under a microscope in the laboratory to determine the stage of weevil development, the incidence of any mortality and gall growth abnormalities, and/or if any parasitoids or other arthropod gall inhabitants are present. A sample of late summer galls also may be collected for laboratory rearing of inhabitants to evaluate level of parasitism or attack by inquilines.

Local spread: Once establishment has been confirmed, monitoring of *Rhinusa pilosa* spread at the patch level will commence. The point or restricted area of initial release within a yellow toadflax patch will be designated as the center of two crossing, perpendicular transects, each 30-100 meters long depending on the size of the patch. At regular intervals along each transect, permanent quadrats (e.g., 50 x 20 centimeters) will be established and monitored yearly for presence and number of galls or adult weevils during the seasonal periods previously mentioned. Location of each quadrat relative to the center and transect identifier also will be recorded for determining the rate and direction of *R. pilosa* spread within the patch.

Impact: The monitoring protocols have yet to be developed, but the same permanent quadrats established to monitor spread could be used to determine the impact of *Rhinusa pilosa* on host plants and any responses of the local plant community post release. In addition to conducting a yearly gall count within each quadrat, stems could be randomly selected and measured for height or incidence of flowering, and percent cover of *Linaria vulgaris*, other invasive plants, and native plant species recorded. Although non-target attack is unanticipated, any plants within the Plantaginaceae also will be inspected for galls during the impact assessments. Additional field experiments may later be designed and conducted, using replicated, treatment (with gall weevils) and control (without) plots, to determine impact.

Appendix 5. Response to comments on EA.

APHIS received comments from the U.S. Fish and Wildlife Service (Service). These comments are addressed below.

Issue 1. The Service issued a concurrence letter on December 16, 2016 in response to APHIS' February 9, 2016, email initiating informal consultation for the release of the stem gall weevil (*Rhinusa pilosa*) for biological control of yellow toadflax. In that letter, the Service concurred with APHIS' determination that the environmental release of stem gall weevil to control yellow toadflax may affect, but is not likely to adversely affect parachute beardtongue (*Penstemon debilis*) (threatened) with critical habitat, blowout penstemon (*Penstemon haydenii*) (endangered), and Penland beardtongue (*Penstemon penlandii*) (endangered) pursuant to section 7 of the Endangered Species Act. The Service concurred based on several factors in the biological assessment, including the commitment to monitor for non-target impacts at initial release sites. If APHIS opts to issue permits to release stem gall weevils, without monitoring for non-target impacts at initial release sites, this would represent a change to the proposed action, as concurred to in our December 16, 2016 letter. In that case, consistent with the regulations addressing interagency consultation (50 CFR 402.16), APHIS should reinitiate formal consultation, prior to issuing permits to release stem gall weevils.

Response: In the informal consultation for *Rhinusa pilosa*, APHIS indicated that the permittee would be conducting the monitoring of non-target impacts at initial release sites, and was very clear in the consultation that APHIS personnel would not be conducting such monitoring. APHIS would include conditions in the release permit requiring that the permittee conduct monitoring. There will be no deviation from the permittee's commitment to monitor for non-target impacts at initial release sites as referenced in the December 16, 2016 letter of concurrence from the Service. Particular attention will be focused on finding and monitoring other members of the Plantaginaceae, including *Penstemon* species.

Issue 2: The Service is also concerned with the lack of information on efficacy and interactions among biocontrol agents and the environment. The Service seeks additional information to clarify the expected efficacy or cumulative benefit of releasing the stem gall weevil. The EA lacks specific details that would suggest that the stem gall weevil is any more suitable for controlling yellow toadflax than the other eight permitted biocontrol agents previously released. The literature indicates that other control agents did just as well under test conditions but their efficacy decreased considerably under field conditions. The Service understands the potential benefit of using several options to control yellow toadflax. The EA should include information from empirical studies that address efficacy and cumulative benefit toward controlling yellow toadflax with multiple biocontrol agents. The Service seeks an explanation regarding the results of the interactions among existing biocontrol agents already in use. For example, the EA mentioned that the larvae of certain flower feeding control agents within toadflax flowers might consume the eggs of some seed feeding weevils, without explaining the significance of this interaction or how the interaction may reduce the overall effectiveness of the stem gall weevil as a control agent.

Response:
Efficacy of the agent: McClay and Balciunas (2005) suggest that the efficacy of weed biocontrol agents could be predictively assessed by adapting a conceptual model, impact = range x abundance x per-capita effect, used for evaluating the ecological impact of invading species (Parker et al., 1999). The authors assert that range and abundance as functions of agent life-history characteristics and responses to physical and biotic attributes of the release environment can be predicted pre-release, with the caveat that the influence of natural enemies on abundance is difficult to anticipate a priori. Per-capita effect can be measured, through quarantine greenhouse or laboratory experiments, by exposing the target weed to known densities or levels of attack by the candidate biological control agent, then measuring the agent's effect on specific correlates of host fitness (e.g., propagule production, biomass).

The primary goal of pre-release efficacy assessments (PREAs) is to evaluate the impact, at high densities, of candidate biological control agents on their target weed species. The rationale for exposing host plants to high densities of the candidate agent is to approximate the impact of agents when they attain so-called outbreak densities, which occurs when host availability is the main limitation to agent abundance. Agents at outbreak densities that fail to control the target weed are conventionally considered ineffective agents, and may also present a higher risk of nontarget effects. PREAs using low densities of the candidate agent can also be useful for indicating if and what type of effects on the target weed occur below outbreak agent densities.

Two complimentary PREAs have been conducted to assess potential biocontrol efficacy of the yellow toadflax stem galling weevil *R. pilosa*; the results of these assessments were not available to be reported in the petition submitted in 2012 for this agent. One assessment was conducted in the weed and candidate agent's native range, as a garden based study, at the Institute for Plant Protection and Environment (IPPE), Zemun (Belgrade), Serbia (hereafter referred to as the Serbian study) (Gassmann et al., 2014). For that study, 60 field-collected, single shoot *L. vulgaris* plants were transplanted into plastic pots with the root system of each plant cut to 3 centimeters (cm) in length to standardize for pre-treatment growth. Half of the plants were randomly selected to be individually caged and exposed to one mated female for 24 hours. All plants were harvested two months later and plant height, number of shoots, and dry biomass were recorded.

The second assessment was conducted in the Insect Microbial Containment Facility at the Lethbridge Research Centre, Agriculture and Agri-Food Canada in Lethbridge, Alberta, Canada (hereafter referred to as the Canadian study). For that study, mated and ovipositing *R. pilosa* females were caged on overwintered, previously galled *L. vulgaris* plants to assess the impact of high agent densities on the target weed (Barnewall, 2011). Previously galled plants were intentionally used in the test to simulate post-release field conditions. Because yellow toadflax is a short-lived perennial species, individual plants would likely be exposed to established biological control agents over multiple years, especially for agent species whose populations typically build up post-release. Plants used in the Canadian study were established from material field collected in late June in Mountain View, Alberta, Canada and potted in 15 cm clay pots held at 22°C under ambient light for 1-2 months in a greenhouse, then subjected to a reduced temperature and light vernalization period through mid-March. Plants were removed from vernalization to a quarantine rearing room and initially held at 10˚C for 7 days, then retained at 22˚C day/ 18˚C night and exposed to a range of

adult *R. pilosa* densities for 72–75 days. A 12:12-hour light-dark photoperiod was maintained in the rearing room with incandescent and florescent plant grow lights. Plants were retained in the greenhouse for two months after all galls had been removed, then again subjected to vernalization until February. Plants removed from vernalization were grown in a quarantine rearing room under the same light and temperature conditions as in the previous year. Five mated and ovipositing females and three males were caged on each of five test plants for two weeks in mesh cages that were 20 cm in diameter by 70 cm tall. Five control plants also were caged, but did not receive any insects. Eleven weeks after the female weevils were added to treatment plants, plant growth and reproductive output were measured for comparison between treatment and control plants. Oviposition occurred on 77 percent of the stems available on treatment plants. Galls were observed on all treatment plants, with 29 ± 11.8 galls produced per plant and a total of 142 across all five treatment plants.

The efficacy of the candidate agent *R. pilosa* is indicated by reductions in highly relevant correlates of host plant fitness, through a comparison of galled versus control plants (*Linaria vulgaris*). The most significant fitness reducing effect of galling, detected by both PREAs, was the reduction in below-ground biomass in the galled plants. Below-ground biomass was reduced 75 percent in galled plants ($F_{1,7} = 8.16$, $P = 0.025$; Canadian study), with mean dry below-ground biomass of galled plants 0.4 ± 0.06 grams (g) versus 1.2 ± 0.1 g for control plants ($F_{1, 58} = 32.9$, $P = 0.001$; Serbian study). This finding is particularly relevant because it indicates that galling might not only compromise the spread of *L. vulgaris* by limiting rhizomatous, clonal stem growth (Nadeau et al., 1992; Lehnhoff et al., 2008), but probably also reduces multi-year persistence and overwintering survival of the weed, which in *Linaria* depends on adequate storage of carbohydrates in the root (Bakshi and Coupland, 1960; Robocker et al., 1972).

Galled plants were 55 percent shorter than control plants ($F_{1,7} = 22.37$, $P = 0.002$; Canadian study), with the height of galled plants 37.2 ± 3.1 cm versus 58.4 ± 3.2 cm for control plants ($F_{1, 58} = 22.1$, $P = 0.001$; Serbian study). The number of shoots produced by galled plants was significantly lower, 3.5 ± 0.6 versus 13.6 ± 1.3 for control plants ($F_{1, 58} = 10.7$, $P = 0.0018$; Serbian study). Gall tissue represented 40 percent of above-ground biomass of treated plants (Canadian study). Reductions of mean dry above-ground biomass for treated plants, galls excluded, were 4.6 ± 0.7 g versus 12.6 ± 1.4 g for control plants ($F_{1, 58} = 26.3$, $P = 0.001$; Serbian study). Reductions in above-ground biomass probably have less lasting negative impacts than the loss of below-ground biomass, but timing ultimately dictates how well yellow toadflax can tolerate the loss of top growth due to grazing or burn down from herbicide application (Krick, 2011; Sing et al., 2016). Galling also significantly reduced the potential for sexual propagation. The proportion of flowering stems was much lower in treatment versus control plants ($F_{1, 7} = 13.37$, $P = 0.008$), and flowering was either fully suppressed (4 of 5 treatment plants vs. 0 of 5 control plants) or delayed in galled plants (all results from Canadian study).

Increased suitability of the candidate agent: Control efficacy reported for the majority of specialist herbivores and legitimate biocontrol agents associated with *Linaria vulgaris* in North America has been minimal (Sing et al., 2005; DeClerck-Floate and McClay, 2013; Gassmann et al., 2014; Sing et al., 2016). The beetle specialist herbivores adventively introduced with yellow toadflax from their shared native range before the mid-1900s, *Rhinusa antirrhini*, *Rhinusa neta*, and

Brachypterolus pulicarius, are now ubiquitous and generally abundant wherever their host plant occurs in North America. Unfortunately all remain insignificantly effective for biological control, primarily because their shared sites of attack, flowers and seeds, have minimal impact on the target weed at a population level. Conversely, many of the biocontrol agents approved for release against this target weed since the 1900s infrequently build to high densities, for a variety of reasons. The toadflax defoliating moth *Calophasia lunula* is thought to be subject to high levels of insect and bird predation, and may be vulnerable to pathogenic attack when moth populations build to high densities in under certain environmental conditions. Root attacking agents such as the weevil *Rhinusa linariae* and the moth *Eteobalea serratella* have successfully established only in a few (former) or no (latter) North American locations; additionally, their presence is confirmed and monitored through destructive sampling, which further impedes population buildup.

To date, only the yellow toadflax stem mining weevil *Mecinus janthinus*, and then only in limited locations, has shown some efficacy against yellow toadflax. Researchers specializing in toadflax biocontrol in Europe, Canada and the United States conclude that *M. janthinus*' inability to consistently establish and impact the target weed at the majority of North American release sites is due primarily to two issues: the agent was released on the wrong target weed, or the agent has been unable to build up population densities to significantly injurious levels. *Mecinus janthinus* has become established on *L. dalmatica* and *L. vulgaris* in both Canada and the United States, although it has shown greater success in controlling *L. dalmatica*. The species now recognized as *M. janthinus sensu stricto* is associated with yellow toadflax, and its sister species *M. janthiniformis* is associated with Dalmatian toadflax. The two species are genetically, ecologically and to a lesser degree, morphologically distinct (Toševski et al., 2011; 2013).

Molecular diagnostics were performed on 632 specimens collected during a comprehensive western North American survey of known sites where *Mecinus janthinus sensu lato* had been released since the 1990s. The results of this study confirmed the presence of both *Mecinus* species in North America, and revealed with a few exceptions that they had an extremely high and consistent level of host fidelity throughout both the adopted and native ranges. The results of this study (Toševski et al., in review) and another evaluating host fidelity of the toadflax flower feeding beetle *B. pulicarius* (MacKinnon et al., 2005; 2007) suggest that host specificity, even among specialist toadflax feeders, can be specific for a single *Linaria* species. Thus, the commenter's assertion about the availability of eight other toadflax natural enemies is not relevant to the petition to release *Rhinusa pilosa*: in the majority of cases, adventive agents have established and are widespread but exert no appreciable control on target weed populations, and approved agents either failed to establish, or have yet to have a predictable impact at most locations where yellow toadflax is present.

The frequent failure of *Mecinus janthinus sensu strcito* to attain breakout densities on yellow toadflax has been attributed to high levels of mortality in pre-emerged adults, during summer when ambient conditions are extremely hot and dry, as a result of wild temperature fluctuations that often occur in spring and fall, or if adequately insulating snow depth is not attained and maintained throughout winter. The life history characteristics of the candidate agent *Rhinusa pilosa* significantly enhance its suitability for release for biological control of *Linaria vulgaris*. Gall development is complete approximately 8–10 days after oviposition under laboratory conditions, corresponding with emergence of the first larval instar (Barnewall, 2011). *Rhinusa pilosa* has a total of three larval

instars that feed and continue development on host tissues within the developed galls. Pupation is also completed within the gall. Eclosed adults remain within the natal gall for 10–15 days while they continue to feed on remnant host tissues before escaping via holes chewed through the gall's outer surface. Larval/pupal development lasts about 45–50 days; the estimated time for complete development from egg to ex-gall adult emergence is 55–65 days. After emergence, the adults feed externally on host stems for about 10 days. Thereafter, the adults repose in litter or cracks in the soil during the day. Summer aestivation is interrupted by occasional feeding, mainly in the evening and at night. In late autumn, adults feed shortly before going into diapause within soil or leaf litter. Because this species avoids most environmental challenges by sheltering under temperature and humidity stable conditions in galls or soil/leaf litter, it will suffer fewer impediments to successful establishment, population increase and impact.

Interaction of multiple biological control agents: The commenter refers specifically to the negative interaction of seed and flower feeding beetles whose offspring compete for the same host resources. This interaction is not germane to the release of *Rhinusa pilosa* for a few reasons. The species in question, *Brachypterolus pulicarius*, *Rhinusa antirrhini*, and *R. neta* were adventively introduced to North America and not therefore released following the established approval process. Redistribution of these species is actively discouraged due to their lack of biocontrol efficacy and undefined legal status (Sing et al., 2016). Because none of the life stages of *Mecinus janthinus* or the candidate agent *R. pilosa* are critically reliant on yellow toadflax flowers or seed capsules, no adverse interaction or negative impact is anticipated from the presence of the three adventively introduced toadflax feeding species. Similarly, no adverse interactions are anticipated from the questionably established but approved yellow toadflax root attacking agents *Rhinusa linariae* or *Eteobalea serratella*.

Adults of the candidate agent *R. pilosa* become active extremely early in the spring, as soon as shoot growth is initiated from sub-soil root buds. Newly emerged adults feed and mate for 3–5 days before the onset of oviposition at approximately 10 days post emergence (Gassmann et al., 2014). *Mecinus janthinus* also emerges early in spring, but because this species goes through a significantly longer pre-oviposition period spent feeding on the apical meristems of yellow toadflax, significant interaction of *R. pilosa* and *M. janthinus* adults is not anticipated. Interspecific interference during oviposition is similarly unlikely, simply based on species specific spatial partitioning of host stem resources. All of *R. pilosa* development occurs within the natal gall, which is positioned between the middle and tip of the host stem. Oviposition and subsequent larval mining by *M. janthinus* occur in the lower part of the host stem. Further, Barnewall (2011) alludes to a potential self-regulating feedback that may occur between *R. pilosa* and yellow toadflax plants at the time that they are being exploited for reproduction, which could govern (and explain) the observed frequency and spacing of oviposition so as not to exceed the host plant's carrying capacity for gall maintenance.

Issue 3: The Service seeks additional information to clarify the expected results of interactions between the stem gall weevil and the non-native parasitoid wasp. The EA provides information on a nonnative parasitic wasp that parasitizes the larval stages of the toadflax flower-feeding beetle (*Brachypterolus pulicarius*) and another closely related biocontrol agent (*Rhinusa antirrhini*) of the stem gall weevil. The wasp has been a factor in reduced effectiveness of these two control agents

under field conditions. However, the EA failed to address whether this wasp will have a similar effect on stem gall weevil and its effectiveness at controlling yellow toadflax.

Response: Initially, it was unclear to APHIS what non-native parasitoid wasp the commenter was referring to because none were mentioned in the EA. However, APHIS contacted the commenter and determined that the wasp they were referring to was *Pteromalus microps*.

The oviposition period of the candidate agent *Rhinusa pilosa* is April–May (Gassmann et al., 2014), ending well before that of *Rhinusa antirrhini* and *Brachypterolus pulicarius*, which is early June through early August (Wilson et al., 2005). The significant temporal difference in the availability of acceptable or suitable immature stages of *R. pilosa* late in the growing season likely reduces the threat of parasitism to *R. pilosa* by the same parasitoid species that attack *B. pulicarius* and *R. antirrhini*. Additionally, *R. pilosa* deposits eggs in the host stem, which triggers gall initiation; development of all subsequent life stages of *R. pilosa* takes place within the same large, protective gall. Oviposition and larval development of *R. antirrhini* and *B. pulicarius* take place in the flowers of yellow toadflax, making the eggs and larvae of these two species more easily detected and vulnerable to exploitation by a generalist parasitoid such as *Pteromalus microps* (Volenberg and Krauth, 1996). Given these fundamental differences in life history characteristics between *R. pilosa* and *R. antirrhini* and *B. pulicarius*, it seems unlikely that *R. pilosa* or its effectiveness at controlling yellow toadflax will be will impacted by this non-native parasitoid wasp.

Issue 4. The EA does not describe the overarching plan for yellow toadflax control. The Service is aware that State Departments of Agriculture may or may not develop and implement integrated weed management strategies. This approach aligns with the concept of Integrated Pest Management (IPM). The EA mentions IPM, but there is a lack of details on how this biocontrol effort and past biocontrol efforts are coordinated and integrated among other control approaches for yellow toadflax. The APHIS, Pest Permitting Branch (PPB) could support an IPM approach for yellow toadflax with the development and implementation of a national control plan that includes a broad communication plan beyond the issuance of permits, sets goals and objectives, monitors actions and tactics, and outlines a reasonable process for reporting on the current releases, efficacy of the biocontrol agents, and national accomplishments toward the control of yellow toadflax. There are examples of national invasive species control plans and teams that implement those plans. The Service is willing to work with PPB to meet our common objectives.

Response: APHIS recognizes the importance of an IPM approach to control yellow toadflax. However, such an IPM program is beyond the scope of this EA which is for the proposed action of issuing a permit for the release of *R. pilosa* into the environment. The PPB does not have the authority or ability to implement or support IPM programs or develop national control plans for weeds.

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Decision and Finding of No Significant Impact
for
Field release of the stem gall weevil *Rhinusa pilosa* (Coleoptera: Curculionidae) for classical biological control of yellow toadflax (*Linaria vulgaris*) (Plantaginaceae) in the contiguous United States.
December 2017

The U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) is proposing to issue permits for release of a stem gall weevil, *Rhinusa pilosa* (Gyllenhal) (Coleoptera: Curculionidae). The agent would be used for the biological control of yellow toadflax (*Linaria vulgaris* Mill.) (Plantaginaceae) in the contiguous United States. Before permits are issued for release of *R. pilosa*, APHIS must analyze the potential impacts of its release into the contiguous United States in accordance with USDA, APHIS National Environmental Policy Act implementing regulations (7 Code of Federal Regulations Part 372). APHIS has prepared an environmental assessment (EA) that analyzes the potential environmental consequences of this action. The EA is available from:

U.S. Department of Agriculture
Animal and Plant Health Inspection Service
Plant Protection and Quarantine
Pests, Pathogens, and Biocontrol Permits
4700 River Road, Unit 133
Riverdale, MD 20737
http://www.aphis.usda.gov/plant_health/ea/index.shtml

The EA analyzed the following two alternatives in response to a request for a permit authorizing environmental release of *R. pilosa*: (1) no action, and (2) issue permits for the release of *Rhinusa pilosa* for biological control of yellow toadflax. A third alternative, to issue permits with special provisions or requirements concerning release procedures or mitigating measures, was considered. However, this alternative was dismissed because no issues were raised that indicated that special provisions or requirements were necessary. The No Action alternative, as described in the EA, would likely result in the continued use at the current level of chemical, mechanical, cultural, and biological controls for the management of yellow toadflax. These control methods described are not alternatives for decisions to be made by APHIS, but are presently being used to control yellow toadflax in the United States and may continue regardless of permit issuance for field release of *R. pilosa.* Notice of this EA was made available in the Federal Register on October 2, 2017 for a 30-day public comment period. The comment period was extended for an additional 15 days to November 16, 2017. One comment was received on the EA by the close of the extended comment period. This comment is addressed in Appendix 5 of the EA.

I have decided to authorize APHIS to issue permits for the environmental release of *Rhinusa pilosa*. The reasons for my decision are:

- *Rhinusa pilosa* is sufficiently host specific and poses little, if any, threat to the biological resources, including non-target plant species, of the contiguous United States.

- *Rhinusa pilosa* is not likely to adversely affect federally listed threatened and endangered species or their critical habitats in the contiguous United States.

- *Rhinusa pilosa* poses no threat to the health of humans or animals.

- No negative cumulative impacts are expected from release of *R. pilosa.*

- There are no disproportionate adverse effects to minorities, low-income populations, or children in accordance with Executive Order 12898 "Federal Actions to Address Environmental Justice in Minority Populations and Low-income Populations" and Executive Order 13045, "Protection of Children from Environmental Health Risks and Safety Risks."

- While there is not total assurance that the release of *R. pilosa* into the environment will be reversible, there is no evidence that this organism will cause any adverse environmental effects.

I have determined that there would be no significant impact to the human environment from the implementation of the action alternative and, therefore, no Environmental Impact Statement needs to be prepared.

Steven Crook, Director
Permitting and Coordination Compliance
U.S. Department of Agriculture
Animal and Plant Health Inspection Service
Plant Protection and Quarantine

12/7/17
Date