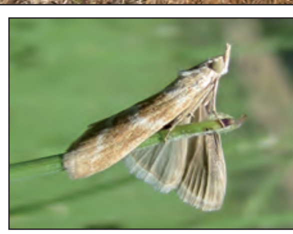
United States Department of Agriculture

Biology and Biological Control of Rush Skeletonweed





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Top: Rush skeletonweed plant (Rachel Winston, MIA Consulting).
Bottom, left to right: *Aceria chondrillae* (Eric Erbe, USDA ARS; bugwood.org); *Bradyrrhoa gilveolella* (Joseph Milan, BLM); *Cystiphora schmidti* (Charles Turner, USDA ARS; bugwood.org); *Puccinia chondrillina* (Joseph Milan, BLM).

BIOLOGY AND BIOLOGICAL CONTROL OF RUSH SKELETONWEED

Joseph Milan, Carol Bell Randall,
Jennifer E. Andreas, and Rachel L. Winston

For additional copies of this publication, contact:

Carol Bell Randall
U.S. Forest Service
Forest Health Protection
2502 E. Sherman Avenue
Coeur d'Alene, ID 83814
(208) 769-3051
crandall@fs.fed.us

Richard Reardon
U.S. Forest Service
Forest Health Technology Enterprise Team
180 Canfield Street
Morgantown, WV 26505
(304) 285-1566
rreardon@fs.fed.us

This publication is available online at
http://www.fs.fed.us/foresthealth/technology/

Authors

Joseph Milan, Biological Control Specialist, Bureau of Land Management, Boise District, Boise, ID; jmilan@blm.gov

Carol Bell Randall, Entomologist, U.S. Forest Service, Forest Health Protection, Coeur d'Alene, ID; crandall@fs.fed.us

Jennifer E. Andreas, Integrated Weed Control Project, Washington State University Extension, Puyallup, WA; jandreas@wsu.edu; www.invasives.wsu.edu

Rachel L. Winston, Environmental Consultant, MIA Consulting, LLC, Sandpoint, ID; rachel@getmia.net

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Chapter 1: Introduction

Overview

Rush skeletonweed (*Chondrilla juncea* L., Figure 1-1) is a perennial plant that can grow up to 4 feet (1.2 m) tall. In its native range, rush skeletonweed occurs from Western Europe to Central Asia, and from southern Russia to Northern Africa.

Rush skeletonweed was first introduced to the northeastern United States in the 1870s. Though present in 10 states and one province in eastern North America (Figure 1-2), it is sparsely distributed in fields and roadsides and is not considered an agricultural problem in the East. Western USA infestations are much more severe and are believed to have begun via contaminated orchard and vineyard rootstocks. Rush skeletonweed was first reported in Washington State in 1938, in Idaho in 1960, in California in 1965, in Oregon in 1971, and in Montana in 1991. The weed has most recently spread to the states of Wyoming, Colorado, Utah, and Arizona, and the province of British Columbia. Rush skeletonweed now occupies more than 6.15 acres (2.5 million ha) in northwestern North America. In Idaho alone, the infested area increased from 50 acres (20 ha) in the 1960s to 3.5 million acres (1.4 million ha) in the 1980s.

Figure 1-1. Rush skeletonweed plant. (Rachel Winston, MIA Consulting)

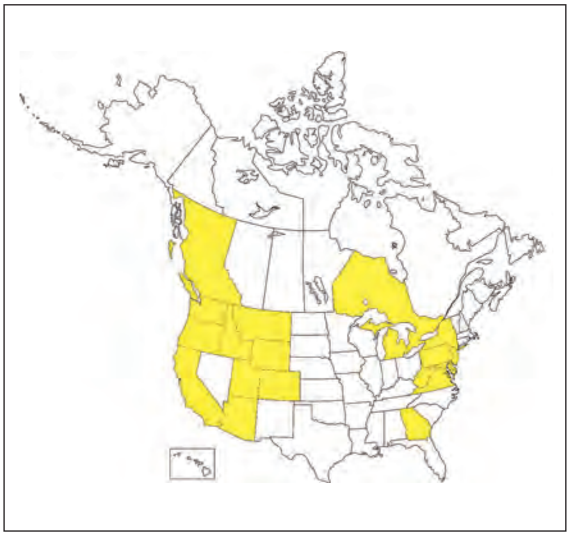


Figure 1-2. Rush skeletonweed North American distribution. Some states and provinces are more heavily infested than others. (USDA PLANTS Database, EDDMapS)

Throughout its native and introduced ranges, rush skeletonweed is found in highly disturbed sites, including roadsides, river banks, dry river beds, degraded coastal dunes, overgrazed rangeland, and in fallow and abandoned fields. It is most commonly found in Mediterranean and steppe climates characterized by cool winters and hot, dry summers, and in coarse-textured, well-drained soils.

Rush skeletonweed is one of the most problematic exotic plant species currently threatening rangeland, forests, agriculture, and conservation areas in the Intermountain West of the United States. Although young rosettes are nutritious and often eaten by livestock and wildlife, cattle still prefer grasses to young rush skeletonweed; older flowering stems of rush skeletonweed are not palatable to most domestic cattle and sheep. Consequently, grazing of infested pastures or rangeland often increases the amount of rush skeletonweed and decreases livestock production. Because of its propensity to compete aggressively for light, water, and nutrients, rush skeletonweed is also a major concern for agricultural crops and for displacing native and/or more desirable species in natural areas.

Responding to the Threat of Rush Skeletonweed

Rush skeletonweed is an invasive species not native to North America whose introduction causes or is likely to cause economic or environmental harm. A general management response to the threat of invasive species is based on four key elements or intermediate outcomes: prevention and preparedness, eradication, containment, and asset-based protection. In order to ensure a timely and appropriate management response, land managers must continually monitor, evaluate, and report, new rush skeletonweed infestations and evaluate how rush skeletonweed responded to each control effort. Research and development informed by the observations and needs of land managers will play a critical role in the eventual success or failure of rush skeletonweed prevention and management activities in its invaded range.

Prevention and Preparedness

Preventing high-risk invasive species from establishing is the most cost-effective approach to managing the threat they pose. Considerable resources and planning are required to maintain prevention of a large number of species. Preparedness encompasses all the activities and resources necessary to successfully manage new invasions.

Eradication

Eradication is generally only possible in the early stages of establishment when distribution and abundance of the invasive species are low. This approach can be almost as cost-effective as prevention.

Containment

Where an invasive species cannot be eradicated, there can be substantial net benefit gained from preventing its further spread. Containment involves measures to eradicate outlying (satellite) infestations and prevent spread beyond the boundaries of core infestations (those that are too large and well established to eradicate). Obtaining a high degree of community support is a prerequisite for any long-term containment program.

Asset-Based Protection

An asset-based approach to managing an invasive species is appropriate once it has become so widespread that it would be inefficient to control the species everywhere it occurs and containment would provide a low return on investment. The asset-based approach is to manage the species only where reducing its adverse effects provides the greatest benefits by achieving protection and restoration outcomes for specific highly valued assets.

Monitoring, Evaluation, and Reporting

For science-based programs, such as invasive species management, monitoring, evaluation, and reporting are elements of adaptive management, whereby programs are continually reviewed and analyzed to ensure that their approaches are consistent with and supportive of any changes in environmental response, community expectation, or scientific knowledge.

Research and Development

The knowledge that comes from research and development is critical to implement evidence-based management approaches. In many cases, substantial advances in invasive species management will require development of new techniques and acquisition of greater knowledge. The investment in research needs to be sufficient to ensure future management is not seriously constrained by insufficient research and development support.

The Invasion Curve

The invasion curve (Figure 1-3) shows that eradication of an invasive species such as rush skeletonweed becomes less likely and control costs increase as an invasive species spreads over time. Prevention is the most cost-effective solution, followed by eradication. If a species is not detected and removed early, intense and long-term control efforts will be unavoidable.

While rush skeletonweed infests large acreages, there are areas, even entire states and provinces, where rush skeletonweed is absent or is present at very low population levels. The diversity of rush skeletonweed populations, from absent to widespread and abundant, throughout its potential range requires land managers to coordinate their management response to rush skeletonweed across larger landscapes to prevent current infestations from spreading into uninfested areas.

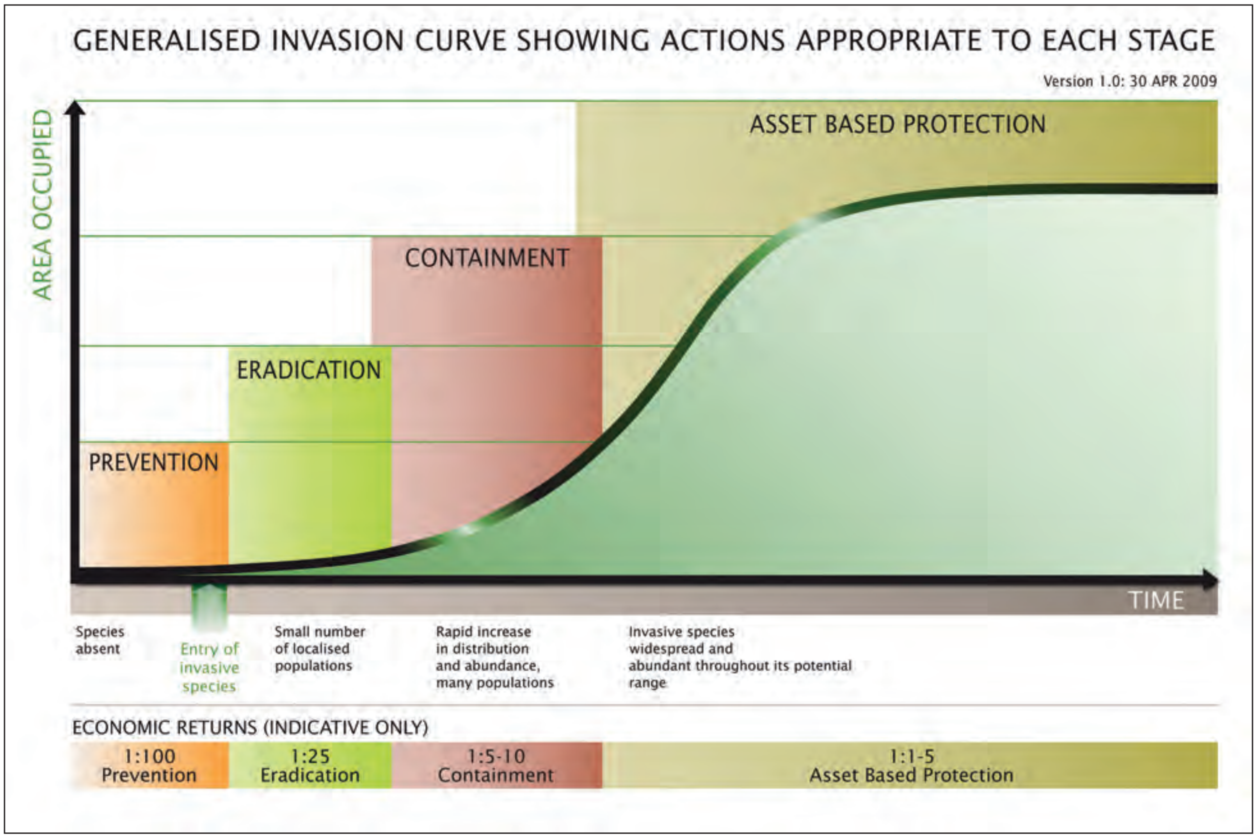


Figure 1-3. Generalized invasion curve showing actions appropriate to each stage. (© State of Victoria, Department of Economic Development, Jobs, Transport and Resources. Reproduced with permission.)

Identifying where rush skeletonweed is on the invasion curve in a particular area is the first step to taking management action. Inventorying and mapping current rush skeletonweed populations coupled with research efforts to predict where rush skeletonweed is most likely to move enables land managers to concentrate resources in areas where rush skeletonweed is likely to invade, and then to treat individual plants and small populations of rush skeletonweed before it is too late to remove them.

Biological control is one of many control methods available to land managers, but biological control is not appropriate for areas on the left side of the invasion curve (species absent [prevention] – small number of localized populations [eradication]). Biological control as a control method is best suited to rush skeletonweed populations in the later phases of the invasion curve (rapid increase in distribution and abundance [containment] – widespread and abundant throughout its potential range [asset based protection]).

Management of Rush Skeletonweed Infestations

Successful management of rush skeletonweed populations is an intensive process which requires land managers to continuously inventory, map, and assess the extent and severity of rush skeletonweed infestations. Land managers must also understand the benefits and shortcomings of each weed control method, alone and in combination, when applied to rush skeletonweed. Chemical control (herbicides) may be used to successfully control small rush skeletonweed infestations where land managers are committed to annual monitoring and, when necessary, re-treatment; however, chemical control can be impractical, prohibitively expensive, and damaging to desired vegetation when treating large rush skeletonweed infestations. Hand pulling small, individual rush skeletonweed plants may be feasible; however, pulling large numbers of small plants or large plants is difficult and may increase the number of rush skeletonweed plants post treatment if viable root fragments are left behind to generate new shoots. Severed rush skeletonweed roots buried up to 4 feet underground (1.2 m) can still send shoots that reach the surface. Repeated mowing can reduce rush skeletonweed vigor and seed production, but may exacerbate the problem by triggering re-growth and spreading seeds. Burning is largely ineffective as a rush skeletonweed control method as it typically encourages the re-growth of rush skeletonweed and may have severe negative, long-term consequences for desired plants. Grazing rush skeletonweed can be an effective control method in certain circumstances, but grazing can be difficult and/or time-consuming, and may have severe negative, long-term consequences for plant communities. Since chemical, physical, and cultural control methods were not universally effective in managing rush skeletonweed throughout its invaded range, a biological control program was initiated in 1936 in Australia, though the first approved biocontrol agent was not released in North America until 1975. This manual discusses the biological control of rush skeletonweed in North America, within the larger context of an integrated rush skeletonweed management strategy.

The most effective weed management strategies are based on regular inventory and monitoring of target weed populations, application of one or many weed control methods, evaluation of treatment efficacy, additional inventory and mapping, and adjustment of control methods as needed to meet management objectives in response to changing weed populations through time.

Integrated Pest Management (IPM) incorporates additional activities that enable land managers to address the threat of rush skeletonweed invasions in infested as well as uninfested areas across a landscape. Integrated pest management activities include education and outreach, inventory and mapping, prevention methods, and control methods (physical control [hand pulling or mowing], cultural control [grazing or fire], chemical control [herbicides], and biological control). IPM relies on the development of realistic pest management objectives, accurate pest identification and mapping, appropriate prevention and control methods, and post-treatment monitoring to ensure current pest-management activities are meeting rush skeletonweed-management goals.

Land managers choose control methods that enable them to achieve their rush skeletonweed management goals or objectives in the most cost-effective manner. No single control method will enable managers to meet their rush skeletonweed management goals in all environments or instances. Control method(s) employed will depend on the size and location of the infested area and specific management goals (e.g., eradication vs. weed density reduction). Small patches of rush skeletonweed may be eliminated through a persistent herbicide program, but large infestations will often require the use of additional control methods. A combination of control methods consistently applied, evaluated, and adjusted through time is usually necessary to attain and maintain weed management goals for rush skeletonweed.

Classical Biological Control of Weeds

Most invasive plants (weeds) in the United States are not native to North America; they arrived with immigrants, through commerce, or by accident from different parts of the world. These non-native plants are generally introduced without their natural enemies, the complex of organisms that feed on or attack the plant in its native range. The enemy release hypothesis suggests that a lack of natural enemies is thought to be one reason plant species become invasive weeds when introduced to areas outside of their native range.

Biological control (also called "biocontrol") of weeds is the deliberate use of living organisms to limit the abundance of a target weed. In this manual, biological control refers to "classical biological control," which reunites host-specific natural enemies from the target weed's native range with the target weed in its introduced range. Natural enemies used in classical biological control of weeds include different organisms such as insects, mites, nematodes, and pathogens. In North America, most weed biological control agents are plant-feeding insects, of which beetles, flies, and moths are among the most commonly used.

Biological control agents may attack a weed's flowers, seeds, roots, foliage, and/or stems. Effective biological control agents seldom kill weeds outright, but work with other stressors such as moisture or nutrient shortages to reduce vigor and reproductive capability, or facilitate secondary infection from pathogens—all of which compromise the weed's ability to compete with other plant species. Once established, root- and crown-feeding biocontrol agents are usually more effective on perennial plants that primarily spread by root buds. Flower- and seed-feeding biocontrol agents are typically more effective on annual or biennial plants that spread only by seed. Regardless of the plant part attacked by biocontrol agents, the aim is always to reduce populations and vigor of the target weed.

There are advantages and disadvantages to biological control of weeds as a management tool. These are listed in Table 1.

Table 1. Advantages/disadvantages of classical biological control as a weed management tool

Advantages	Disadvantages
Target specificity	Will not work on every weed in every setting
Continuous action	Permanent; cannot be undone
Long-term cost-effective; can provide sustained control at the landscape scale	Funding and testing candidate biocontrol agents is expensive; measurable impact may take years or even decades to materialize
Integrates well with other control methods	Approved biocontrol agents are not available for all exotic weeds
Generally environmentally benign	Like all weed control methods, “nontarget” effects are possible, but pre-release testing reduces the risks
Self-dispersing, even into rough or difficult to access terrain	Unpredictable level of control; generally does not eliminate weed

To be approved for release in North America, weed biocontrol agents must be host-specific, meaning they must feed and develop only on the target weed, or in limited cases, on a few closely related plant species. They must never feed on any crop or protected plant species; attack on ornamental plants may be minimally tolerated and is evaluated on a case-by-case basis. Rigorous testing is required to confirm that biocontrol agents are host specific and effective. Potential biocontrol agents often undergo five or more years of testing to ensure that rigid host specificity requirements are met, and results are vetted at a number of stages in the approval process.

The United States Department of Agriculture’s Animal and Plant Health Inspection Service - Plant Protection and Quarantine (USDA-APHIS-PPQ) is the federal regulatory agency responsible for providing testing guidelines and authorizing the importation of biocontrol agents into the United States. The Canadian Food Inspection Agency (CFIA) serves the same regulatory role in Canada. Federal laws and regulations are in place to identify and avoid potential risks to native and economically valuable plants and animals that could result from exotic organisms introduced to manage weeds. The Technical Advisory Group (TAG) for Biological Control Agents of Weeds is an expert committee with representatives from USA federal regulatory, resource management, and environmental protection agencies, and regulatory counterparts from Canada and Mexico. TAG members review all petitions to import new biocontrol agents into the USA, and make recommendations to USDA-APHIS-PPQ regarding the safety and potential impact of prospective biocontrol agents. Weed biocontrol researchers work closely with USDA-APHIS-PPQ and TAG to accurately assess the environmental safety of potential weed biocontrol agents and programs. In addition, some states in the USA have their own approval process to permit field release of weed biocontrol agents. In Canada, the Biological Control Review Committee (BCRC) draws upon the expertise and perspectives of Canadian-based researchers (e.g. entomologists, botanists, ecologists, weed biological control scientists) from academic, government, and private sectors for scientific review of petitions submitted to the CFIA. The BCRC reviews submissions for compliance with the North American Plant Protection Organization's (NAPPO) Regional Standards for Phytosanitary Measures (RSMP) No. 7. The BCRC also reviews submissions to APHIS. The BCRC conclusions factor into the final TAG recommendation to APHIS on whether to support the release of the proposed biocontrol agent in the USA. When release of a biocontrol agent is proposed for both the USA and Canada, APHIS and the CFIA attempt to coordinate decisions based on the assessed safety of each country's plant resources.

Code of Best Practices for Classical Biological Control of Weeds

Biological control practitioners have adopted the International Code of Best Practices for Biological Control of Weeds. The Code was developed in 1999 by delegates and participants in the Tenth International Symposium for Biological Control of Weeds to both improve the efficacy of, and reduce potential negative impacts from, weed biological control. In following the Code, practitioners reduce the potential for causing environmental damage through the use of weed biological control by voluntarily restricting biocontrol activities to those most likely to result in success and least likely to cause harm.

International Code of Best Practices for Classical Biological Control of Weeds[1]

1. Ensure that the target weed's potential impact justifies release of non-endemic biocontrol agents
2. Obtain multi-agency approval for target
3. Select biocontrol agents with potential to control target
4. Release safe and approved biocontrol agents
5. Ensure that only the intended biocontrol agent is released
6. Use appropriate protocols for release and documentation
7. Monitor impact on the target
8. Stop releases of ineffective biocontrol agents or when control is achieved
9. Monitor impacts on potential nontargets
10. Encourage assessment of changes in plant and animal communities
11. Monitor interaction among biocontrol agents
12. Communicate results to public

[1]Ratified July 9, 1999, by the delegates to the X International Symposium on Biological Control of Weeds, Bozeman, MT

Although weed biological control is an effective and important weed management tool, it does not work in all cases and should not be expected to eradicate the target weed. Even in the most successful cases, biocontrol often requires multiple years before impacts become noticeable. When classical biological control alone does not result in an acceptable level of weed control, other weed control methods (e.g., physical, cultural, or chemical control methods) may be incorporated to achieve desired results. For a more in-depth description of weed control methods in the context of rush skeletonweed management, please refer to Chapter 5.

Biological Control of Rush Skeletonweed

In 1975 the rush skeletonweed gall midge, *Cystiphora schmidti* (Rübsaamen), became the first biocontrol agent approved and released in North America on rush skeletonweed (Figure 1-4). By 2002, three additional species had been approved and released in the United States, including a rust fungus, a mite, and a moth. The moth was intentionally redistributed to Canada starting in 2007.

Is Biological Control of Rush Skeletonweed Right For You?

When biological control is successful, biocontrol agents increase in abundance until they suppress (or contribute to the suppression of) the target weed. As local target weed populations are reduced, their biological control agent populations also decline due to starvation and/or dispersal to other target weed infestations. In many biocontrol systems, there are fluctuations over time with the target weed becoming more abundant, followed by increases of its biocontrol agent, until the target weed/biocontrol agent populations stabilize at a much lower abundance.

As stated in Table 1, biological control is not effective in every weed system or at every infestation. We recommend that you develop an integrated weed management program in which biological control is one of several control methods considered. Here are some questions you should ask before you begin a biological control program:

Figure 1-4. Adult *Cystiphora schmidti*, the rush skeletonweed gall midge. (Charles Turner, USDA ARS, bugwood.org)

Is my management goal to eradicate the weed or reduce its abundance?
Biological control does not eradicate target weeds, so it is not a good fit with an eradication goal; however, depending on the target weed, which biological control agent is used, and land use, biological control can be effective at reducing the abundance and vigor of a target weed to an acceptable level.

How soon do I need results: this season, one to two seasons, or within five to ten years?
Biological control requires time and patience to work. Generally, it can take one to three years after release to confirm that biological control agents are established at a site, and even longer for biocontrol agents to cause significant impacts to the target weed. For some weed infestations, 5-30 years may be needed for biological control to reach its weed management potential.

What resources can I devote to my weed problem?
If you have only a small rush skeletonweed problem (few infested acres or much smaller), weed control methods such as hand pulling and/or herbicides, followed by regular monitoring for re-growth and re-treatment when necessary, may be most effective. These intensive control methods may allow you to achieve rapid control and prevent the weed from infesting more area, especially when infestations occur in high-priority treatment areas such as travel corridors where the weed is more likely to readily disperse. If the target weed is well established over a large area (>1 acre or 0.4 ha), and resources are limited, biological control may be the most economical weed control option.

Is the weed the problem, or a symptom of the problem?
Invasive plant infestations often occur where there is or has been a disturbance in a desirable plant community. Without restoration of a desirable, resilient plant community, and especially if disturbance continues, biological control is unlikely to solve your weed problems.

The ideal biological control program:

1. Is based upon an understanding of the target weed, its habitat, land use and condition, and management objectives
2. Is part of a broader integrated weed management program
3. Has considered all weed control methods and determined that biological control is the best option based on available resources and weed management objectives
4. Has realistic weed management goals and timetables
5. Includes resources to ensure adequate monitoring of the target weed, the vegetation community, and populations of biological control agents

About This Manual

This manual provides information on the biology and ecology of rush skeletonweed and each of its biological control agents. It also presents guidelines to establish and manage biological control agents as part of an integrated rush skeletonweed management program.

Chapter 1: Introduction provides introductory information on rush skeletonweed (including its distribution, habitat, and economic impact) and classical biological control.

Chapter 2: Getting to Know Rush Skeletonweed provides detailed descriptions of the taxonomy, growth characteristics and features, invaded habitats, and occurrence of rush skeletonweed in North America. It also describes how to differentiate rush skeletonweed from look-alike species.

Chapter 3: Biology of Rush Skeletonweed Biological Control Agents describes biological control agents of rush skeletonweed, including details on each biocontrol agent's native range, original source of releases in North America, parts of rush skeletonweed plants attacked, life cycle, description, host specificity, known nontarget effects, habitat preferences, and availability. This chapter is particularly useful for identifying biological control agents in the field.

Chapter 4: Elements of a Rush Skeletonweed Biological Control Program includes detailed information and guidelines on how to plan, implement, monitor, and evaluate an effective rush skeletonweed biological control program. Included are guidelines and methods for:

- Selecting and preparing biological control agent release sites
- Collecting, handling, transporting, shipping, and releasing biological control agents
- Monitoring biological control agents and vegetation

Chapter 5: An Integrated Rush Skeletonweed Management Program discusses the role of biological control in the context of an integrated rush skeletonweed management program.

The **Glossary** defines technical terms frequently used by those involved in rush skeletonweed biological control and found throughout this manual.

References lists selected publications and resources utilized to compile this manual.

Appendices:

I. Troubleshooting Guide: When Things Go Wrong
II. Sample Biological Control Agent Release Form
III. Rush Skeletonweed Standardized Impact Monitoring Protocol
IV. General Biological Control Agent Monitoring Form
V. Rush Skeletonweed Quantitative Monitoring Form – Associated Vegetation

Chapter 2: Getting to Know Rush Skeletonweed

Taxonomy and Related Species

Rush skeletonweed belongs to the sunflower family (Asteraceae) and the tribe Cichorieae. Members of the sunflower family produce flower heads, or capitula, that are an aggregation of many individual flowers (Figure 2-1a). These flowers, called florets, are clustered together and attached to a receptacle. There are two types of florets: disc and ray (Figure 2-1b, 2-1c). Some species produce both types of florets, while others (like rush skeletonweed) produce only one. The receptacle and florets are enclosed by modified leaves called involucral bracts. Each floret produces one seed (achene) from early to late summer. Seeds often have a tuft of whitish hairs (pappus) on one end, similar to those on dandelion seeds (Figure 2-1d).

Members of the Cichorieae tribe are most easily identified by the milky sap exuded upon damage to their foliage (Figure 2-2). The tribe Cichorieae includes *Chondrilla*, *Lactuca*, *Taraxacum*, and ~97 other genera worldwide. In North America, the tribe is represented by 49 genera and 229 species that range from annual forbs to perennial shrubs. Within *Chondrilla*, there are approximately 25 species, out of which only rush skeletonweed occurs in North America.

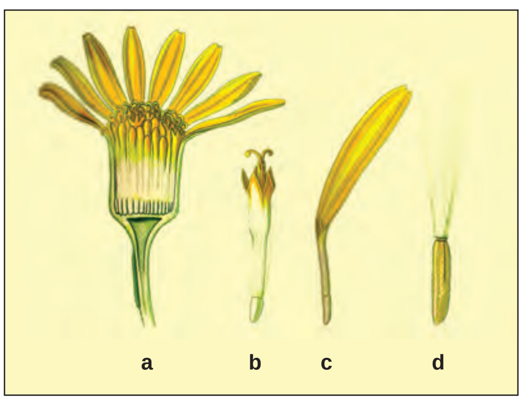


Figure 2-1. Sunflower family: a. capitulum; b. disc floret; c. ray floret; d. seed with pappus. (a-d: Prof. Dr. Otto Wilhelm Thomé Flora von Deutschland, Österreich und der Schweiz 1885, Gera, Germany, www.biolib.de; © expired)

Figure 2-2. Milky latex in rush skeletonweed stem. (Rachel Winston, MIA Consulting)

Rush Skeletonweed

Scientific Name

Chondrilla juncea L.

Common Names

Rush skeletonweed, skeletonweed, hogbite, nakedweed, gum succory, rushlike gum-succory, devil's-grass

Classification

KINGDOM	Plantae	Plants
SUBKINGDOM	Tracheobionta	Vascular plants
SUPERDIVISION	Spermatophyta	Seed plants
DIVISION	Magnoliophyta	Flowering plants
CLASS	Magnoliopsida	Dicotyledons
SUBCLASS	Asteridae	
ORDER	Asterales	
FAMILY	Asteraceae	Sunflower family
GENUS	*Chondrilla*	*Chondrilla*
SPECIES	*Chondrilla juncea* L.	Rush skeletonweed

History

Rush skeletonweed, a native of Eurasia and northern Africa, was inadvertently introduced to northeastern North America in the 1870s. It was first recorded in western North America in 1938.

Description

At a Glance

Rush skeletonweed is a herbaceous perennial typically growing 1-4 feet (0.3-1.2 m) tall from a deep and sometimes rhizomatous root system. Rosettes have deeply lobed, hairless leaves up to 5 inches (13 cm) long. Plants produce multiple wiry stems (Figure 2-3a). Bottom portions of stems are covered with stiff, golden-reddish and downward pointing hairs (trichomes). Stem leaves are alternate, small, narrow, and up to 4 inches (10 cm) long. As flowering stems mature, stem leaves often wither; the remaining bare stems give the plant an overall skeleton appearance (Figure 2-3b). Flower heads are 0.5 inches (1 cm) across and consist of 9-12 yellow ray florets that produce seeds without fertilization. Flower heads are produced along and at tips of branches in late summer. Seeds are small, brown, and topped by tufts of pappus. All parts of the plant exude a milky latex when damaged.

Figure 2-3. Rush skeletonweed: a. flowering plant (Eric Coombs, Oregon Department of Agriculture, bugwood.org); b. plant with stem leaves withered back, giving a skeletal appearance (Joseph Milan, BLM).

Roots

Rush skeletonweed develops taproots that are slender and deep, growing up to 6.5 feet (2 m) long. Roots have short, lateral branches along their length and fork repeatedly in their lower half (Figure 2-4a). Most lateral roots are short-lived and less than 3 inches (8 cm) long, but some lateral roots near the surface can become rhizomatous, often in very sandy, gravelly, or waterlogged soils. Upper portions of the primary taproot and larger lateral roots form buds that produce daughter rosettes in undisturbed plants. All parts of the root are brittle and easily broken. Root pieces as small as 1 inch (2.5 cm) long and 0.5 inches (1.25 cm) in diameter can develop into new plants. Severed roots buried up to 4 feet underground (1.2 m) can still send shoots that reach the surface.

Stems

Plants typically grow 1-4 feet (0.3-1.2 m) tall and have one or more flowering stems with multiple spreading and ascending branches. Stems are often wiry and rigid and may lack leaves. When present, stem leaves often wither back with maturity; remaining stems give the plant an overall skeleton appearance. Upper portions of stems are not hairy, but at the base of flowering stems are many stiff, golden-reddish, and downward pointing hairs (trichomes, Figure 2-4b).

Leaves

Rosettes consist of numerous hairless leaves. Each is 2-5 inches (4-13 cm) long and 0.6-1.8 inches (1.5-4.5 cm) wide, though they are wider at the tip than the base (Figure 2-4c). Rosette leaves have lobed margins. The lobes are

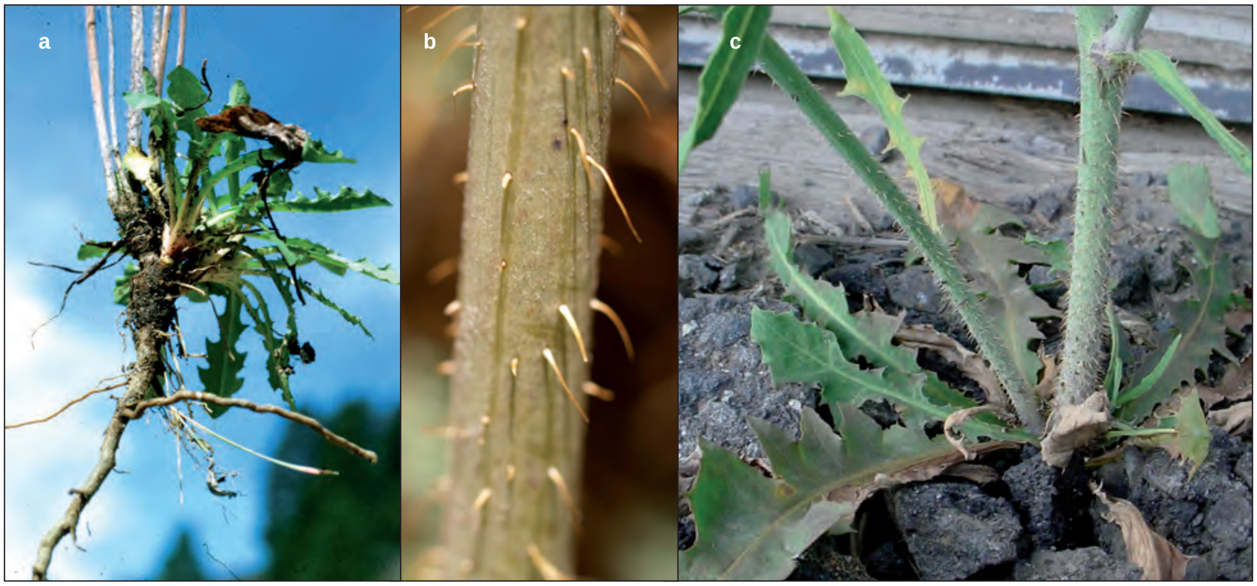


Figure 2-4. Rush skeletonweed: a. root system (Steve Dewey, Utah State University); b. distinctive stiff, golden-reddish, downward pointing basal stem hairs (Rachel Winston, MIA Consulting); c. rosette leaves (Richard Old, XID Services, Inc, www.xidservices.com); (a,c: bugwood.org).

irregular, opposite each other, and point backwards. Leaves are often tinged with purple or reddish-brown, especially along margins and near leaf tips. When present, stem leaves are small, linear, 0.8-4 inches (2-10 cm) long and 0.04 to 0.3 inches (1-8 mm) wide. As flowering stems bolt and mature, basal and stem leaves often wither; upper leaves are at times no more than scale-like bracts.

Flowers

Flower heads are produced along and at tips of branches, either solitary or in clumps of 2-5. Each flower head consists of 9-12 bright yellow ray florets (Figure 2-5a). Florets themselves (each resembling one single petal) consist of 5 fused petals, their individual tips separate at the ends of flowers. The involucre (base of the flower head) is small, less than 0.5 inches (13 mm) tall, and attached to branches via a short and sometimes nonexistent stem (Figure 2-5b). Bracts are cylindrical as a unit and occur in two unequal rows at the base of the involucre, the outer row being much smaller than the inner. Flowering occurs from May to October, depending upon location.

Fruits and Seeds

Fruits are achenes (hereafter referred to as seeds). They are oblong, tapered at both ends, pale to dark brown, and 0.1 inches (3-4 mm) long. Each seed has many ribs running lengthwise, and is topped by a large amount of pappus consisting of numerous, fine white bristles (Figure 2-5c). First-year plants typically produce 50 to 150 flower heads annually, which equates to 500 to 1,500 seeds per plant. Longer-lived individuals are capable of producing much more, ~20,000 seeds per plant on average per year.

Figure 2-5. Rush skeletonweed: a. flower head; b. flower head and involucre (a,b: Rachel Winston, MIA Consulting); c. seed with pappus (D. Walters and C. Southwick, USDA, bugwood.org).

See Figure 2-6 for an artist's rendition of rush skeletonweed traits.

Biology and Ecology

Rush skeletonweed spreads by seeds as well as rhizomes and root fragments. The plant reproduces mostly through apomixis which means its seeds are typically produced without fertilization; however, limited amounts of fertilization have been observed in its native range. Apomixis is often beneficial to an invasive species growing where pollinators, environmental factors, or other rush skeletonweed plants may be limiting. Seeds are readily carried by wind, water, humans, and other animals and are dispersed fall through winter. While the vast majority of seeds germinate within one year, the soil seed bank can at times yield rush skeletonweed seedlings several years after seed drop. The highest rates of germination have been recorded for seeds buried shallowly in loamy or sandy soil. Germination is lowest for seeds on the soil surface or those in clay soil where water is difficult to access.

Autumn rains stimulate seedling germination, and seedlings or rosettes overwinter. Seedlings require a continuous supply of water for 3-6 weeks following germination. Consequently, seedlings that germinate in the summer following a single rain event often die of desiccation shortly thereafter, while seedlings that germinate in the fall or spring often receive water from subsequent rainfalls and survive. Seedlings are also sensitive to shading from other plants and survive better in areas with little competition for light. Increasing day length in spring induces flowering stems to bolt and branch. During this stage, rosette and stem leaves wither back, and photosynthesis takes place in the green stems. The mature size of the plant will depend, in part, on soil type, water level, the genetic potential of the plant, and

Figure 2-6. Artist's rendition of rush skeletonweed key traits. (Christiaan Sepp, Flora Batava of Afbeelding en Beschrijving van Nederlandsche Gewassen, X Deel, Amsterdam, Netherlands, 1849; © expired)

plant density. Flowering occurs from spring to fall. Plants less than one year old are capable of producing seeds. Plants re-sprout each spring from adventitious buds in their roots.

In undisturbed plants, buds near the top of the taproot and on major lateral roots (rhizomes) can produce several new rosettes sharing a common root system (Figure 2-7a). When the original lateral root connection with the parent plant breaks down, these rosettes may form their own roots to become satellite plants. The majority of rush skeletonweed roots are brittle and easily fragmented. Root pieces as small as 1 inch (2.5 cm) long and 0.5 inches (1.25 cm) in diameter can develop into new plants, provided the fragments are from older plants and there is sufficient soil moisture. Severed roots buried up to 4 feet underground (1.2 m) can still send shoots that reach the surface.

Habitat

Soil disturbance is a very important contributor to rush skeletonweed seedling establishment. The weed can often be found creating dense monocultures along railroads, roadsides, riverbanks, fallow fields, abandoned lots, and overgrazed rangeland (Figure 2-7b,c). A variety of habitat types and plant communities can be invaded by rush skeletonweed following heavy grazing, trampling, cultivation, logging, and burning. The weed does best in semiarid conditions with cool, moist winters and warm summers without extensive drought. Rush skeletonweed also performs best in well-drained soils and in areas without significant competing vegetation.

Distribution

As of 2015, rush skeletonweed is considered established in 18 states and two Canadian provinces (Figure 1-2, repeated here in Figure 2-8a). It has been declared noxious in seven of the western states and the one western province in which it is currently established, as well as two additional states where it is not yet present (Figure 2-8b).

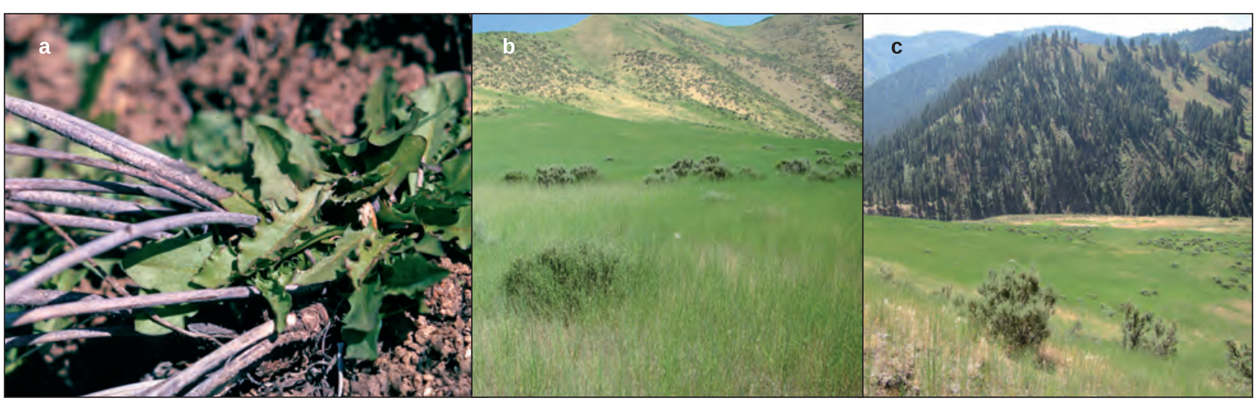


Figure 2-7. Rush skeletonweed: a. sprouting from root buds amid previous year's dead stems (Utah State University Archive, bugwood.org); b. infestation on rangeland; c. infestation in a valley (b,c: Joseph Milan, BLM).

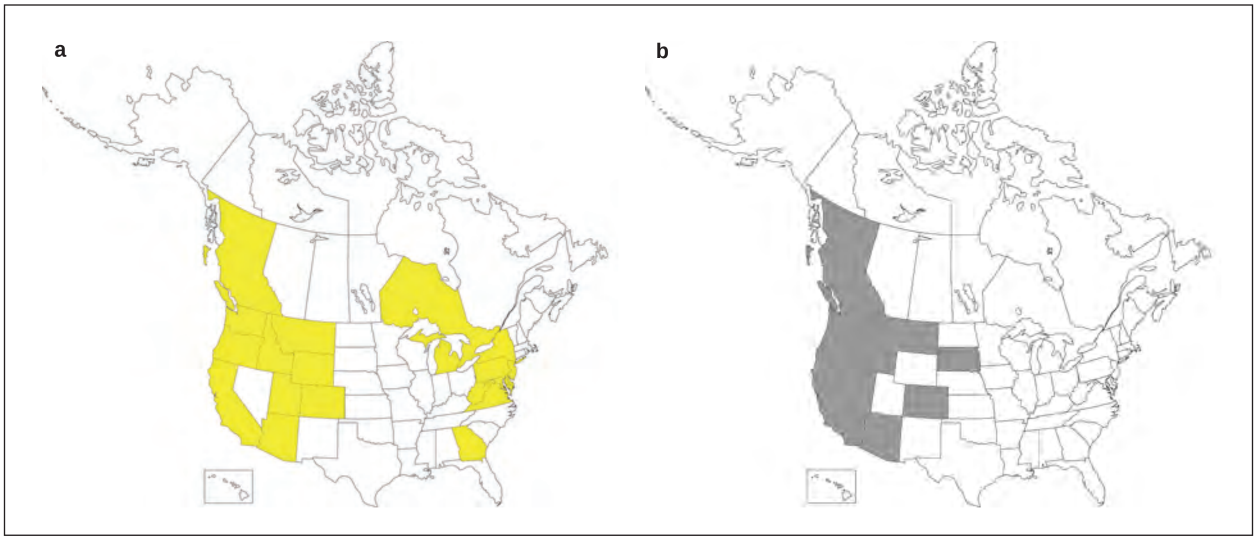


Figure 2-8. States and provinces where rush skeletonweed is: a. established (USDA PLANTS Database, EDDMapS); b. listed as noxious. Note that some states and provinces are more heavily infested than others.

Comments

Seven genotypes of rush skeletonweed are currently recognized in North America (Figure 2-9a,b), and it is believed the genotypes respond differently to environmental conditions and control methods. Some biocontrol agents behave differently depending on the genotype of the attacked plant, as is discussed further in Chapter 3.

Some sources claim there are distinct morphological differences between the most common genotypes (1, 2, 3), in that genotype 2 grows bushier and more branched and flowers in early summer, compared to the less bushy and later-flowering genotypes 1 and 3. The same sources indicate genotype 1 grows shorter than genotypes 2 and 3, but the majority of land managers have observed a wide variety of morphological traits between and among the three genotypes, depending on habitat and climatic conditions.

Commonly Confused Species

Numerous species present in North America have an appearance similar to rush skeletonweed, especially those in the same family and tribe. The species most closely resembling rush skeletonweed are listed in Table 2, along with key characteristics that can be used to differentiate the look-alikes.

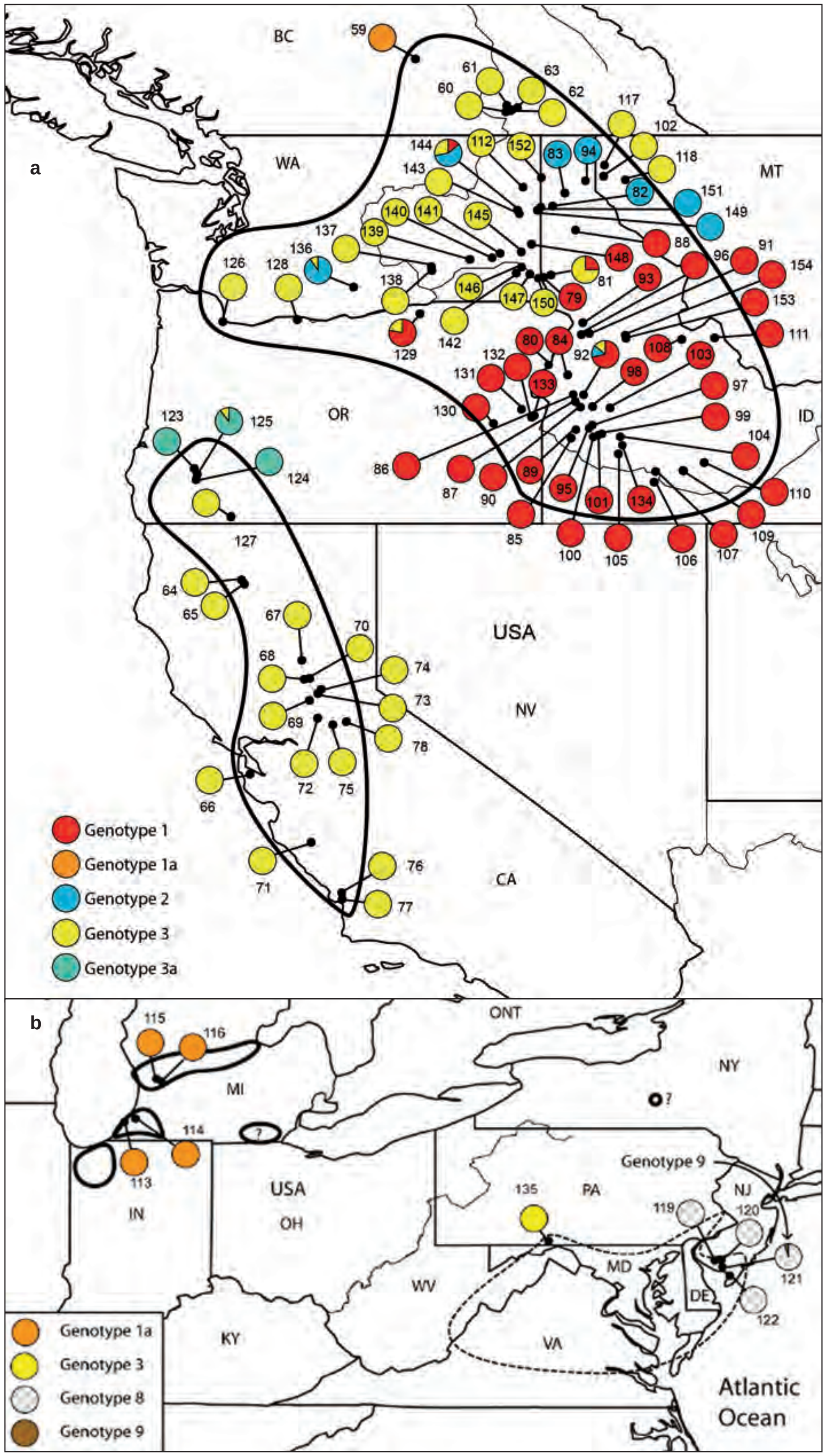


Figure 2-9. Distribution of the seven genotypes of rush skeletonweed identified in: a. western North America; b. eastern North America. (Reprinted with permission from Gaskin, J.F., M. Schwarzländer, C.L. Kinter, J.F. Smith, and S.J. Novak. 2013. Propagule pressure, genetic structure, and geographic origins of *Chondrilla juncea* [Asteraceae]: an apomictic invader on three continents. American Journal of Botany 100[9]: 1871-1882.)

Table 2. North American species in the same family (Asteraceae) and similar in appearance to rush skeletonweed (RSW), along with key traits for differentiation.

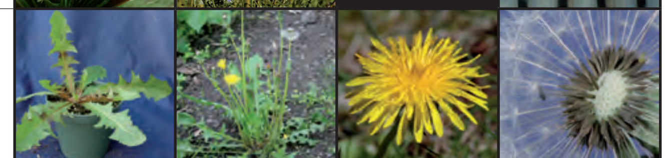
Species	Similarities to RSW	Differences from RSW	Look-alike Appearance
Rush skeletonweed *Chondrilla juncea* Exotic perennial forb			
Chicory *Cichorium intybus* Exotic biennial or perennial forb	Rosette leaf lobes often point outwards or backwards. Plants appear skeleton-like later in the season. All parts of the plant exude a milky latex when torn. There are coarse hairs on some stems and leaves.	Rosette leaf lobes sometimes point forward and are not always opposite. Hairs on stems (when present) and leaves are white rather than reddish, and don't always point downward. Flower heads are blue.	
Dandelion *Taraxacum officinale* Exotic and native perennial forb	Rosette leaves are hairless, leaf lobes point backwards and are opposite each other. Flower heads are yellow. All parts of the plant exude a milky latex when torn.	Stems are hollow, unbranched, leafless, and fleshy. Flower heads are much larger.	
Prickly lettuce *Lactuca serriola* Exotic annual or biennial forb	Rosette leaves are lobed. Stem bases have stiff hairs. Flower heads are yellow and of similar size. All parts of the plant exude a milky latex when torn.	Rosette leaf lobes are jagged along margins. Stiff hairs extend all along the stem and leaf undersides. There is one main flowering stem that is branched in its upper half.	
Rush skeletonplant *Lygodesmia juncea* Native perennial forb	Plants appear skeleton-like. All parts of the plant exude a milky latex when torn.	Lacks an overwintering rosette. Plants only grow 4-16 inches (10-40 cm) tall. Flower heads are pink.	
Yellow starthistle *Centaurea solstitialis* Exotic annual forb	Rosette leaves are lobed. Flower heads are yellow.	Rosette leaf lobes point outwards and are not always opposite. Plants are gray-green, covered in fine hair, and have winged stems. Flower heads have very long, spiny bracts.	

Photo Credits: Rush skeletonweed: 1: Richard Old, XID Services, Inc., www.xidservices.com; 2: Joseph Milan, BLM; 3: Rachel Winston, MIA Consulting; 4: Steve Dewey, Utah State University; 1,4: bugwood.org. **Chicory:** 1,4: Bruce Ackley, Ohio State University, bugwood.org; 2: R.A. Howard, Smithsonian Institution, USDA NRCS PLANTS Database; 3: Joaquim Alves Gaspar. **Dandelion:** 1: Bruce Ackley, Ohio State University; 2: Robert Vidéki, Doronicum Kft; 3: Chris Evans, Illinois Wildlife Action Plan; 4: Joseph Berger; 1-4: bugwood.org. **Prickly lettuce:** 1: Ohio State University Weed Lab Archive; 2,3: Mary Ellen (Mel) Harte; 4: Steve Dewey, Utah State University; 1-4 bugwood.org. **Rush skeletonplant:** 1: Rachel Winston, MIA Consulting; 2: ©Al Schneider, www.swcoloradowildflowers.com; 3: Matt Lavin, Bozeman, MT; 4: "Lostinfog", www.flickr.com/photos/lostinfog/. **Yellow starthistle:** 1: Rachel Winston, MIA Consulting; 2: Steve Dewey, Utah State University; 3: Peggy Greb, USDA ARS; 4: Cindy Roche; 1-4: bugwood.org.

Chapter 3: Biology of Rush Skeletonweed Biological Control Agents

Classical biocontrol agents may be found in a number of taxonomic groups. The majority of approved biocontrol agents are invertebrates in the kingdom animal and the phylum Arthropoda (insects and mites); however, there are approved biocontrol agents which are in the kingdom fungi. Rush skeletonweed biocontrol agents currently approved for use in North America include two species of insects (a moth and a fly [arthropods in the class Insecta]), a mite (arthropod in the class Arachnida), and a rust fungus (basidiomycete in the class Pucciniomycetes). Their taxonomic groups are described in greater detail in the following sections.

Insects

Insects are the largest and most diverse class of animals. Basic knowledge of insect anatomy and lifecycle will help in understanding insects, and recognizing them in the field.

Most insects used in weed biocontrol have complete metamorphosis, which means they exhibit a life cycle with four distinct stages: egg, larva, pupa, and adult (Figure 3-1a). Insects have an exoskeleton (a hard external skeleton) and a segmented body divided into three regions (head, thorax, and abdomen, Figure 3-1b). Adult insects have three pairs of segmented legs attached to the thorax, and a head with one pair each of compound eyes and antennae (Figure 3-1c).

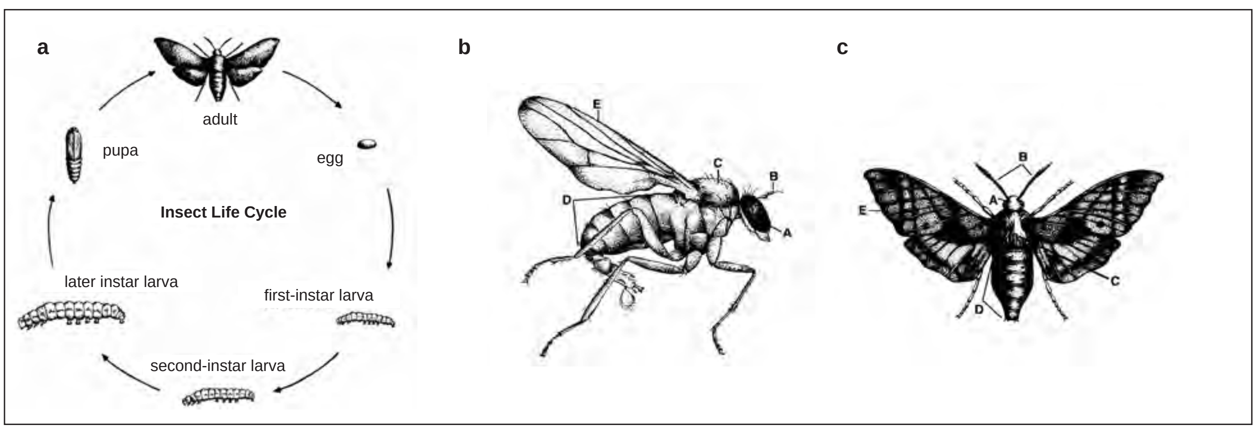


Figure 3-1. Line drawings of: a. fly lifecycle showing complete metamorphosis (Susan Kedzie-Webb); b. fly and c. moth anatomy: A. head, B. antenna, C. thorax, D. abdomen, E. wing (a-c: adapted from Biological Control of Weeds in the West, Rees et al. 1996).

Because insects have an external skeleton, they must shed their skeleton in order to grow. This process of shedding the exoskeleton is called molting. Larval stages between molts are called “instars.” Larvae generally complete three to five instars before they molt into pupae. During the pupal stage, insects change from larvae to adults. Insects do not feed or molt during the pupal stage. Adult insects emerge from the pupal stage and do not grow or molt.

Butterflies and Moths (Order Lepidoptera)

Adult Lepidoptera have two pairs of membranous wings that are covered with powder-like scales. Adult butterflies/moths have prominent antennae and coiled mouthparts that are adapted to siphoning sap and nectar from plant flowers. They can be bright- or dull-colored, and males and females of the same species do not always have the same coloration. Adult butterflies/moths feed very little, if at all. Lepidoptera larvae (known as caterpillars) have a toughened head capsule, chewing mouthparts, and a soft body; they are active feeders. The pupal stage of butterflies/moths is known as a chrysalis and is often enclosed in a cocoon.

Flies (Order Diptera)

Many insects have the word “fly” in their name, though they may not be true flies. In the common names of true flies, “fly” is written as a separate word (e.g., house fly) to distinguish them from other orders of insects that use “fly” in their name (e.g., butterfly in the order Lepidoptera and mayfly in the order Ephemeroptera). Adult true flies are easily distinguished from other orders of insects by their single pair of membranous wings and typically soft bodies. Larvae of most true flies, called maggots, are legless and worm-like.

Mites

Like insects, mites are in the phylum Arthropoda; however, they belong to a different class, Arachnida, whose adult members are characterized by having 8 legs (compared to the 6 legs of insects). Mites have gradual metamorphosis. The first immature stage in mites is called larva; mites in this stage have only 6 legs. The second immature stage is called nymph and has 8 legs. Nymphs are usually very similar in appearance to adults (Figure 3-2). Larvae, nymphs, and adults all feed by piercing and sucking cell contents.

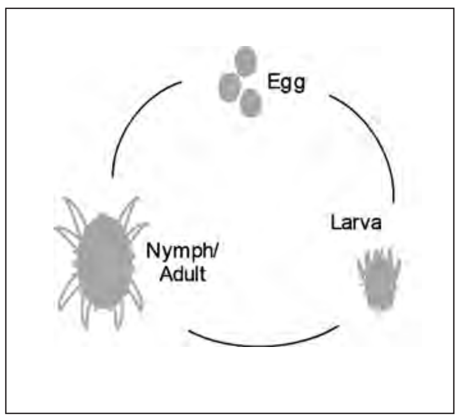


Figure 3-2. Mite life cycle.
(Rachel Winston, MIA Consulting)

Fungi

The fungus biocontrol agent for rush skeletonweed is a rust in the phylum Basidiomycota, class Pucciniomycetes. Rusts are obligate parasites; they require a living host to obtain nutrients and complete their life cycle. Rusts typically attack leaves and stems of the host plant. Rust infections usually appear as numerous rusty, orange, yellow, or even white colored spots (pustules) that rupture the leaf surface and release spores that resemble colored powder (typically yellow, orange, or brown). Most rust infections are local spots but some may spread internally through the plant. Rusts spread from plant to plant mostly by windblown spores, although insects, rain, and animals may aide in the transmission and infection process.

The life cycle of rust fungi can be very complicated. Rust fungi can produce up to five distinctive spore types which have different functions from infesting a new host plant, re-infecting the same host plant, and producing pustules on infected plant leaves and stems.

Rush Skeletonweed Biological Control Agents

The four species used for North American rush skeletonweed biocontrol attack different parts of the plant (Figure 3-3). The moth attacks rush skeletonweed roots, the gall midge attacks leaves and stems, and the mite and rust fungus both attack all aboveground growth. Each species is described in the following sections.

Aceria chondrillae (Canestrini)

Rush skeletonweed gall mite
Synonyms: *Eriophyes chondrillae* (Canestrini)

Kingdom	Animalia
Phylum	Arthropoda
Class	Arachnida
Subclass	Acari
Family	Eriophyidae
Native Distribution	Eurasia, Mediterranean
Original Source	USA: Italy CAN: Italy via USA
First Release	USA: 1977 CAN: 1993
Nontarget Effects	None reported

Description

All stages are tiny and best viewed with a microscope. Nymphs are pale yellow and 0.10 mm long in the first stage. Second stage nymphs are humpbacked, orange, have four legs, and reach 0.17 mm. Adults are worm-like, yellow-orange, and have two pairs of legs (Figure 3-4a). Males are up to 0.18 mm long while females are 0.26 mm.

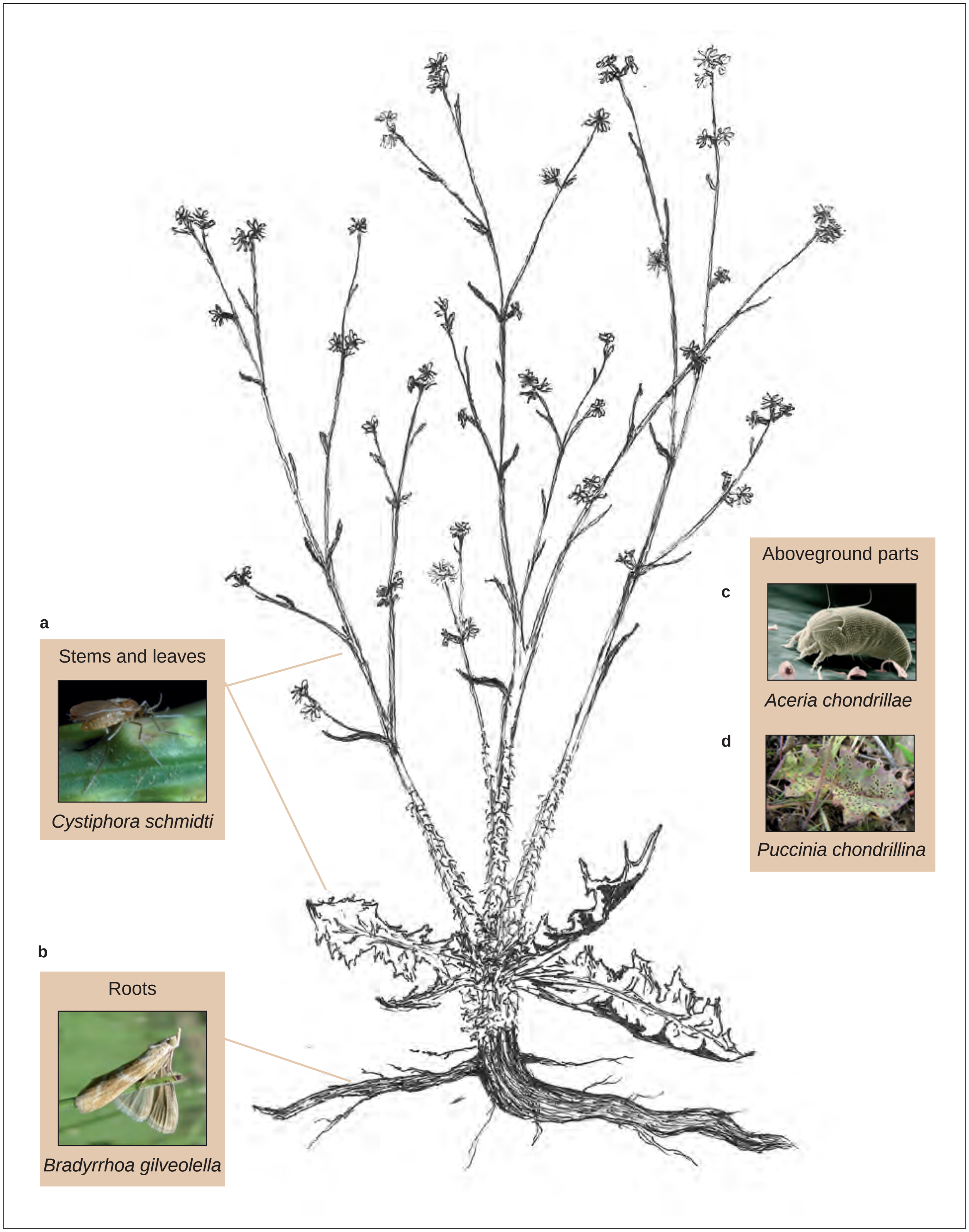


Figure 3-3. General location of greatest attack by rush skeletonweed biological control agents (Plant: Rachel Winston, MIA Consulting): a. *Cystiphora schmidti* (Charles Turner, USDA ARS); b. *Bradyrrhoa gilveolella* (Joseph Milan, BLM); c. *Aceria chondrillae* (Eric Erbe, USDA ARS); d. *Puccinia chondrillina* (Joseph Milan, BLM); (a,c: bugwood.org).

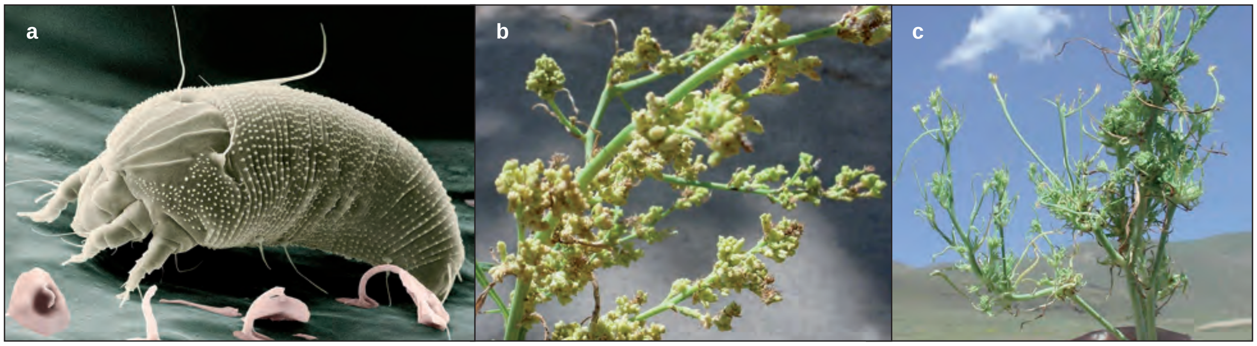


Figure 3-4. *Aceria chondrillae*: a. magnified adult (Eric Erbe, USDA ARS); b., c. damage (b: Richard Old, XID Services, INC; c: Biotechnology and Biological Control Agency); (a,b: bugwood.org).

Life Cycle

There are multiple generations per year. Overwintering adults attack shoot buds when rush skeletonweed bolts in spring. Feeding leads to the formation of contorted galls; each gall may contain several hundred mites. Females lay 60-100 eggs within the gall they occupy. One generation can be completed in 10-12 days. Mites spread with wind-dispersed seeds throughout the growing season via silk strands that act as parachutes. Mite populations and galls increase until skeletonweed dies back in the winter (Figure 3-5).

Habitat Preference

The mite is well adapted to a variety of environmental conditions. It rapidly colonizes plants growing in undisturbed, well-drained soils on south- or west-facing slopes. Mite populations do not persist in sites subjected to repetitive soil disturbance, such as cropland. High overwintering mortality occurs in areas with harsh winters and without winter rosettes of rush skeletonweed.

Damage

Galls induced by mite feeding create a characteristic deformed appearance (Figure 3-4b,c). The galls stunt shoot growth, reduce rosette and seed production, reduce root carbohydrate reserves, and often result in seedling death. Though this typically does not kill older plants, mite galling can help reduce the rate of spread.

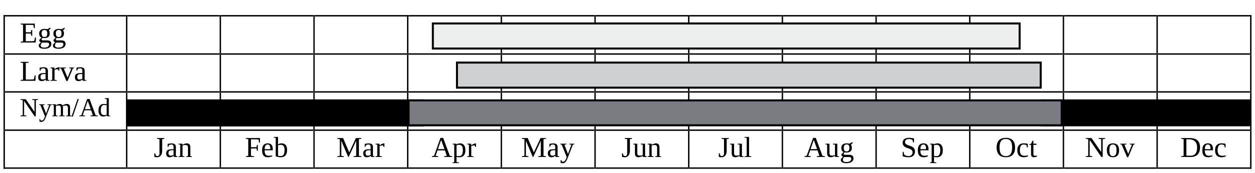


Figure 3-5. Life cycle of *Aceria chondrillae*. Bars indicate the approximate length of activity for each life stage; dates will vary depending on local conditions. Black bars represent the inactive overwintering period. Note: there are multiple generations annually.

Current Status and Availability

In the United States, the abundance and impact of the mite are variable (Figure 3-6). It is widespread in Idaho, Oregon, and Washington where it reduces flowering and seed production by 50-90 percent, depending on environmental conditions and plant size and genotype. The mite can reportedly develop on the three main genotypes of rush skeletonweed found in western North America, though it is believed to be more effective on genotypes 1 and 3. Efficacy is limited in California due to predation.

Figure 3-6. North American establishment of *Aceria chondrillae*.

After spreading to Canada naturally from the United States, the mite was intentionally redistributed for a few years within British Columbia. Though it is established at multiple locations, weed populations are persisting. Mite abundance is low and overall abundance is limited in Canada.

Bradyrrhoa gilveolella (Treitschke)

Rush skeletonweed root moth

Kingdom	Animalia
Phylum	Arthropoda
Class	Insecta
Order	Lepidoptera
Family	Pyralidae
Native Distribution	Europe, Mediterranean
Original Source	USA: Greece CAN: Greece via USA
First Release	USA: 2002 CAN: 2007
Nontarget Effects	None reported

Description

Eggs are tiny, flattened, and initially white but turn reddish and darken with age. Newly hatched larvae are pink with brown heads. Late-instar larvae are 20-25 mm long, off-white, and have brown head capsules (Figure 3-7a). Pupae are tan and up to 25 mm long (Figure 3-7b). Adults are 13 mm long, cream-colored, and have three brown, horizontal bands on their front wings (Figure 3-7c). They have wingspans up to 28 mm.

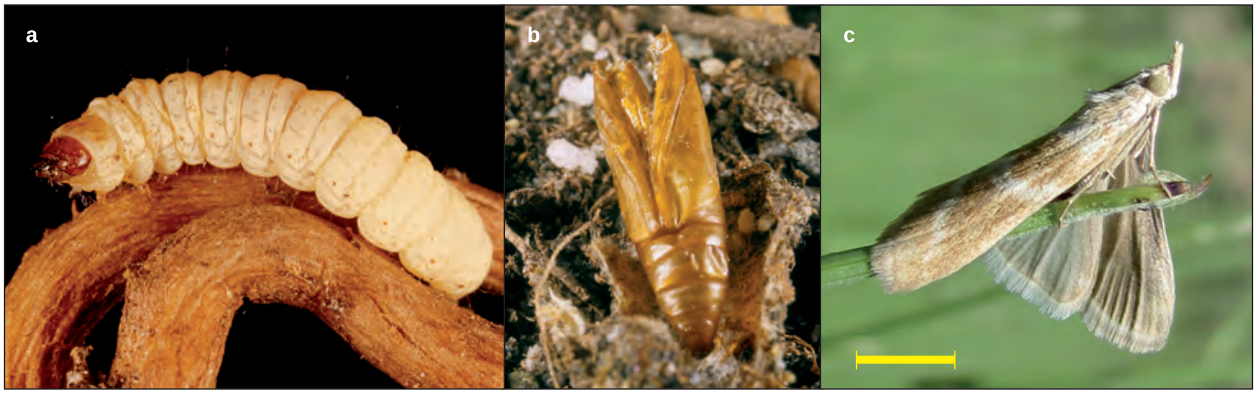


Figure 3-7. *Bradyrrhoa gilveolella*: a. larva; b. pupa (a,b: Laura Parsons and Mark Schwarzländer, University of Idaho); c. adult; yellow scale bar depicts adult body length (Joseph Milan, BLM).

Life Cycle

Two generations per year have been reported in Europe, but in North America, it appears most Bradyrrhoa gilveolella have one generation per year. In North America, adults emerge from late spring through early summer as rush skeletonweed bolts and flowers. Females lay eggs (up to 250 each) on stems or soil near plants. Once in contact with the plant, larvae feed into the stem base, and move downward to attach themselves to the root. Several larvae may feed on the same root simultaneously. Larvae develop through five instars, feeding on root cortex and spinning feeding tubes made of silk, sand, and frass as they travel. Tubes are 2.8 inches (7.1 cm) long on average and extend to the soil surface and are used by the moths to reach the soil surface. Many refer to these tubes as exit chimneys (Figure 3-8a,b). Larvae overwinter in feeding tubes, and pupation occurs in the tubes throughout spring and summer (Figure 3-9).

Figure 3-8. *Bradyrrhoa gilveolella* feeding tubes/exit chimneys: a. close up among roots (Laura Parsons & Mark Schwarzländer, University of Idaho); b. indicated by red arrow (Joseph Milan, BLM).

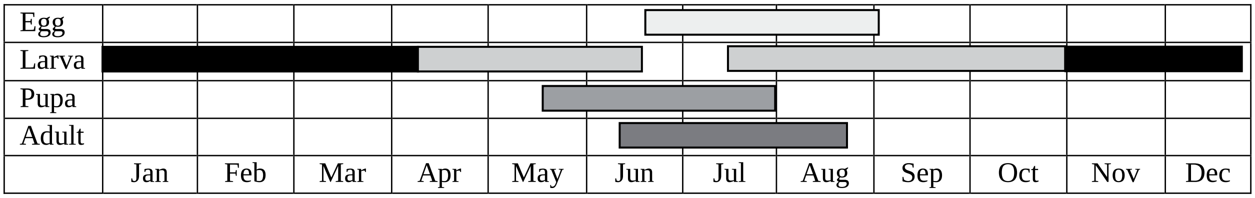


Figure 3-9. Life cycle of *Bradyrrhoa gilveolella*. Bars indicate the approximate length of activity for each life stage; dates will vary depending on local conditions. Black bars represent the inactive overwintering period.

Habitat Preference

The rush skeletonweed root moth does best on plants growing in sandy, granitic, or loose-textured soils on south-facing slopes.

Damage

Adults do not do any appreciable damage. Heavy larval feeding results in diminished root reserves and decreased plant vigor.

Current Status and Availability

In the United States, the first several years of releases failed to establish. Releases made in recent years have been successful, and populations are becoming locally abundant at some of those sites. It is too soon following establishment to determine the overall impact to rush skeletonweed populations. Releases are continuing. The moth can reportedly develop on the three main genotypes of rush skeletonweed found in western North America (Figure 3-10).

Figure 3-10. North American establishment of *Bradyrrhoa gilveolella*.

Although early results in Canada were promising, it is believed that all *Bradyrrhoa gilveolella* releases have thus far failed to establish.

Cystiphora schmidti (Rübsaamen)

Rush skeletonweed gall midge

Kingdom	Animalia
Phylum	Arthropoda
Class	Insecta
Order	Diptera
Family	Cecidomyiidae
Native Distribution	Eurasia, Mediterranean
Original Source	USA: Germany via Australia
First Release	USA: 1975
Nontarget Effects	None reported

Description

Eggs are tiny and oval. Larvae are flattened, 1-2.5 mm long, and are pink or orange (Figure 3-11a, b). Adults are light brown and very small, usually 1 to 1.5 mm long. Legs are long and delicate (Figure 3-11c). Female abdomens end in a bulbous enlargement.

Life Cycle

Adults emerge in spring, and females deposit 60-180 eggs in leaves of rush skeletonweed rosettes. Larvae feed on stem and leaf tissue, inducing the formation of purplish galls. Leaf galls are circular, 3 mm in diameter, and slightly raised, whereas stem galls are elongated and usually more elevated (Figure 3-12a, b). Pupation occurs within galls with each larva spinning a silky cocoon around itself prior to pupation. Adults emerge from cocoons and galls using pupal head spines, destroying plant tissue in the process. New eggs are laid in stems and stem leaves. There are four or five generations per year (Figure 3-13). Complete development from egg to adult can take just under four weeks. Larvae or pupae overwinter in galls or soil.

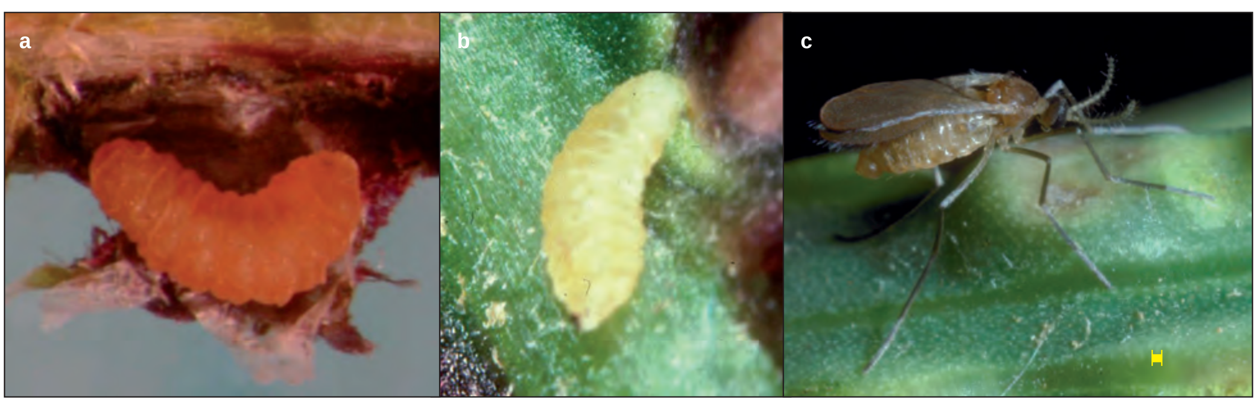


Figure 3-11. *Cystiphora schmidti*: a. larva (Eric Coombs, Oregon Department of Agriculture); b. larva (Gary Piper, Washington State University); c. adult (Charles Turner, USDA ARS); (a-c: bugwood.org).

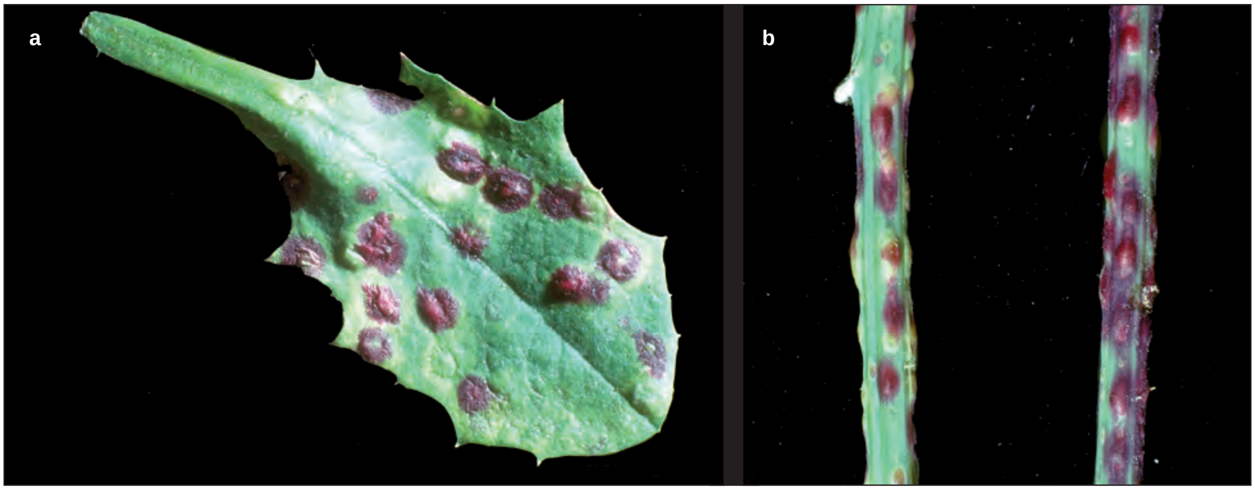


Figure 3-12. *Cystiphora schmidti*: a., b. damage (a,b: Gary Piper, Washington State University, bugwood.org).

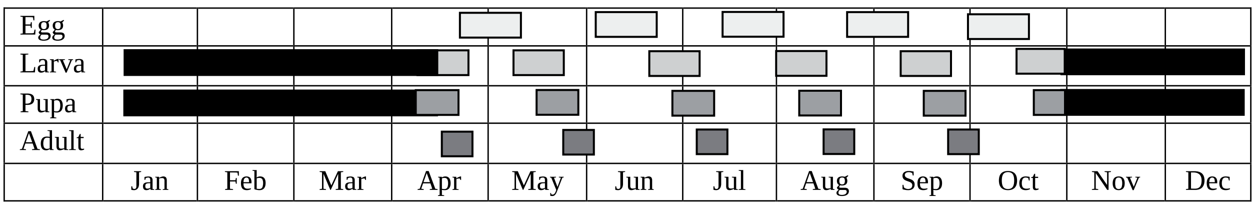


Figure 3-13. Life cycle of *Cystiphora schmidti*. Bars indicate the approximate length of activity for each life stage; dates will vary depending on local conditions. Black bars represent the inactive overwintering period.

Habitat Preference

The rush skeletonweed gall midge does best in warm, dry areas and on plants growing in open locations in well-drained soil.

Damage

Attacked tissue is injured or destroyed, leading to fewer branches and flower heads and less viable seeds. This does not kill existing plants, but can help reduce the rate of spread.

Current Status and Availability

All genotypes of rush skeletonweed present in North America are believed to be susceptible to attack from this biological control agent. In the United States, infested plants are stunted and have decreased seed production. Midge populations are generally small, however, as a result of high rates of parasitism and predation. Consequently, the overall abundance and impacts of the midge are limited.

Figure 3-14. North American establishment of *Cystiphora schmidti*.

The rush skeletonweed gall midge is not present in Canada, nor is it approved for release in that country.

Puccinia chondrillina (Bubák & Syd.)

Rush skeletonweed rust fungus

Kingdom	Fungi
Phylum	Basidiomycota
Class	Pucciniomycetes
Order	Pucciniales
Native Distribution	Eurasia, Mediterranean
Original Source	USA: Italy CAN: Italy via USA
First Release	USA: 1976 CAN: 1992
Nontarget Effects	None reported

Description and Life Cycle

The fungus produces up to five spore stages throughout the growing season. In the spring, overwintering spores germinate and infest rush skeletonweed rosette leaves, forming yellowish chlorotic lesions with raised centers. These turn into orangish-brown pustules that produce large amounts of dry, powdery, round, spores that are rusty or dark brown in coloration (Figure 3-15). These spread rapidly from plant to plant; they are easily dispersed by both wind and rain. Multiple cycles may be produced throughout the year.

Habitat Preference

The rush skeletonweed rust fungus does best in mesic climates with regular dew periods. The rust is hindered by overly harsh winters that kill infected hosts, or overly dry spells that lack a consistent dew period.

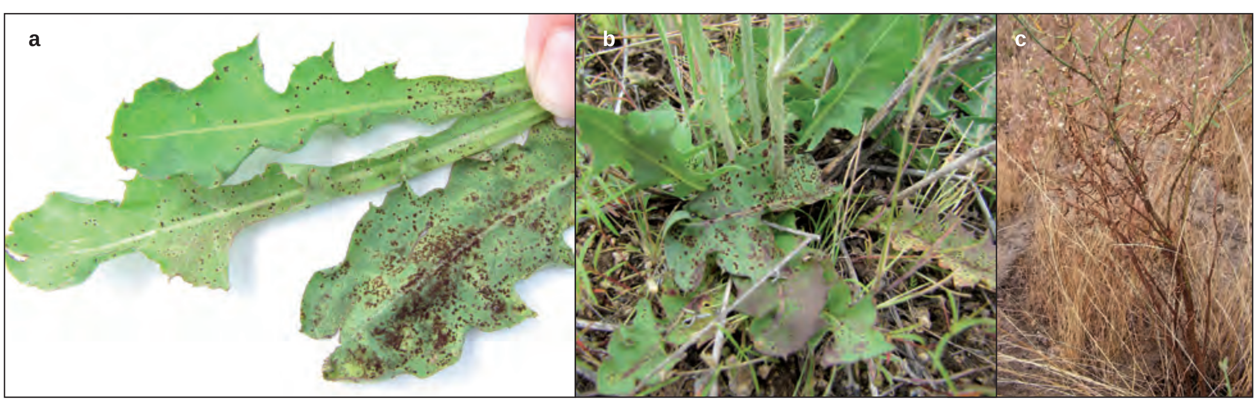


Figure 3-15. *Puccinia chondrillina*: a. spores and pustules on infected leaves (Jennifer Andreas, Washington State University Extension); b. infected rosette (Joseph Milan, BLM); c. infected stems (Eric Coombs, Oregon Department of Agriculture, bugwood.org).

Damage

Pustules reduce rush skeletonweed photosynthetic capabilities and deplete root nutrient storage, leading to plant weakening and even death. Small rosettes and seedlings are often destroyed by heavy rust infestations. If larger plants are infected sufficiently early in the season, flowering stems are stunted and deformed and produce few viable seeds.

Current Status and Availability

Two strains of this rust fungus were first introduced from Italy and released in California, Idaho, Oregon, and Washington beginning in 1976. The rust was also introduced accidentally in the eastern United States (Maryland) in an unknown year. One intentionally introduced strain spread naturally from the United States to British Columbia by 1976.

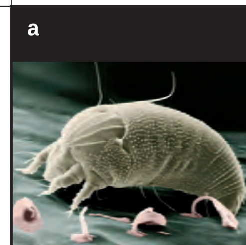
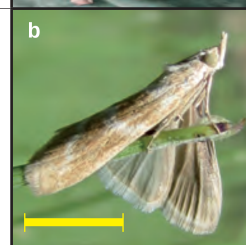
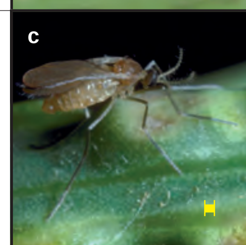
In the United States, efficacy varies by rust strain, weed genotype, and site conditions. Of the three most prevalent genotypes of rush skeletonweed in North America, genotype 2 is resistant to both rust strains, genotype 1 is resistant to one strain but not the other, and genotype 3 is susceptible to both strains. The rust is considered the most effective biological control agent in Washington and California where it decreases plant size and reproductive output; it is less effective in Idaho, Montana, and Oregon. The rust fares poorly on hot and dry sites, and one strain is parasitized.

In Canada, the rust is widespread and has been observed stunting and reducing the density of young rush skeletonweed plants. It is most effective in high moisture areas and in regions where infected overwintering rosettes are not killed by harsh temperatures. Despite being abundant in British Columbia, its overall impact is considered limited as rush skeletonweed populations are still persisting (Figure 3-16).

Figure 3-16. North American establishment of *Puccinia chondrillina*.

See Tables 3 and 4 for comparisons of traits and activity of rush skeletonweed biological control agents.

Table 3. Traits of biological control agents introduced for the control of rush skeletonweed in North America.

Biological Control Agent	Generations per Year	Overwintering Stage and Location	Appearance			
Aceria chondrillae Rush skeletonweed gall mite	Multiple	Adults in galls		EGG: Microscopic	NYMPHS: microscopic; first stage pale yellow and 0.10 mm long; second stage humpbacked, orange, have four legs, and reach 0.17 mm	
Bradyrrhoa gilveolella Rush skeletonweed root moth	Two	Larvae in feeding tubes in roots		EGG: Tiny; cylindrical; pale orange; laid stems or soil near skeletonweed plants	LARVA: Early instars pink with brown heads; late instars off-white with brown head capsules, 20-25 mm long; 5 instars	PUPA: Tan; up to 25 mm long; in larval feeding tubes made of silk, sand and frass
Cystiphora schmidti Rush skeletonweed gall midge	Four or five	Larvae or pupae in galls or soil		EGG: Tiny; oval-shaped; first generation laid in rosette leaves; later generations laid in stems or leaves	MAGGOTS: Flattened; pink or orange; 1-2.5 mm long	PUPA: Within a gall, in a silky cocoon spun by larva
Puccinia chondrillina Rush skeletonweed rust fungus	Multiple	Dormant spores at the base of flowering stems		INFECTION: Yellowish chlorotic lesions with raised centers form on leaves and stems, becoming orangish-brown pustules that produce large amounts of spores. Spores are dry, powdery, round, and dark brown or rust-colored.		

Photos: a. *Aceria chondrillae* (Eric Erbe, USDA ARS, bugwood.org); b. *Bradyrrhoa gilveolella* (Joseph Milan, BLM); c. *Cystiphora schmidti* (Charles Turner, USDA ARS, bugwood.org); d. *Puccinia chondrillina* (Jennifer Andreas, Washington State University Extension).

Table 4. Comparison of rush skeletonweed biocontrol agent activity according to rush skeletonweed growth stage (plant and biocontrol agent stages will vary by climate and location).

Month	Rush skeletonweed plant life stage	*Aceria chondrillae*	*Bradyrrhoa gilveolella*	*Cystiphora schmidti*	*Puccinia chondrillina*
January	Seedlings or rosettes overwinter; seeds continue to disperse from previous year's stems.	Adults overwinter in galls and in winter rosettes.	Larvae overwinter in feeding tubes.	Larvae or pupae overwinter in galls or soil.	Dormant spores overwinter.
February					
March					
April	Overwintered rosettes begin bolting and branching; second-year and older plants re-sprout from root system and bolt; rosette and stem leaves wither back.	Adults feed on shoot buds and stem tips, inducing the formation of galls. Eggs are laid within the galls. Hatching mites feed within galls. The entire process is repeated through several generations throughout the growing season. A single generation can be completed in 10-12 days.		Pupation occurs in galls or soil; emerging adults lay eggs in leaves of skeletonweed rosettes.	Spores germinate on rosette leaves forming yellowish lesions with raised centers.
May	Flowering occurs; seeds mature; senescence begins.		Larvae pupate in feeding tubes.	Hatching larvae feed on stem and leaf tissues, inducing the formation of galls; pupation occurs within galls; adults emerge from galls using pupal head spines and lay new eggs in stems and stem leaves. The entire process is repeated through four or five generations across the growing season. A single generation can be completed in less than four weeks.	Orangish-brown pustules form and produce abundant powdery brown spores during the growing season; these are easily dispersed by wind and rain, spreading rapidly from plant to plant. There are multiple cycles per growing season.
June			Pupation continues; adults emerge and lay eggs on stems or on soil near skeletonweed plants; new larvae feed into the stem base and on into the roots Adult emergence and egg-laying continue; larvae spin feeding tubes made of silk, sand and frass.		
July					
August					
September			Larvae continue feeding in feeding tubes.		
October		Some mites may spread with wind-dispersed seeds.			Lesions form at the bases of skeletonweed stems; these form teliospores that overwinter in dormancy.
November	Seeds on current-year stems disperse; seedlings germinate with autumn rain; seedlings or rosettes overwinter.	Adults overwinter in galls and in winter rosettes.	Larvae overwinter in feeding tubes.	Larvae or pupae overwinter in galls or soil.	
December					

Chapter 4: Elements of a Rush Skeletonweed Biological Control Program

Before You Begin

Biological control is one of many weed control methods available to land managers, but biological control is not appropriate for areas where rush skeletonweed is not present or where a small number of localized populations occur. Biological control as a control method is best suited to rush skeletonweed populations in the later phases of the invasion curve, where rush skeletonweed populations are experiencing a rapid increase in distribution and abundance, or where rush skeletonweed is widespread and abundant throughout its potential range (asset based protection, Figure 1-3 repeated here in Figure 4-1).

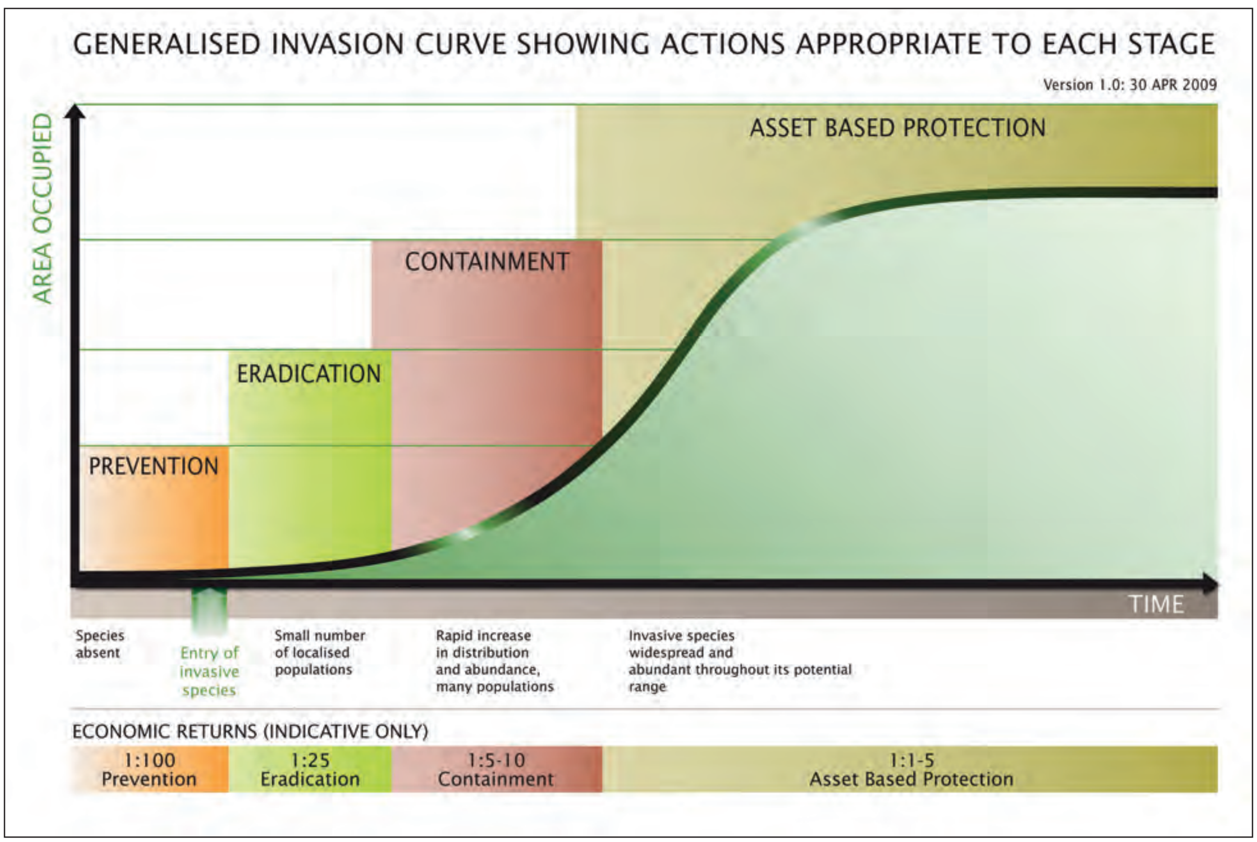


Figure 4-1. Generalized invasion curve showing actions appropriate to each stage. (© State of Victoria, Department of Economic Development, Jobs, Transport and Resources. Reproduced with permission.)

The results of using biological control to treat rush skeletonweed may vary greatly from site to site for a variety of reasons. Land managers should develop treatment programs that complement management activities and objectives unique to the area. This is accomplished by first understanding the scope of the rush skeletonweed problem, defining overall goals for the rush skeletonweed management program, and understanding the control methods available for accomplishing the goals.

Determining the Scope of the Problem

The first step should be to develop a distribution map of rush skeletonweed at a scale that will allow you to address the problem in a manner consistent with your overall land-management objectives and available weed management resources. The most appropriate scale may encompass a large landscape with a variety of site characteristics and land uses managed by many different land owners/managers, all of whom contribute to mapping efforts (Figure 4-2a). In large management areas with significant rush skeletonweed infestations and limited resources, aerial mapping of large patches of rush skeletonweed may be sufficient to identify priority areas for additional survey, mapping, and weed management activities. In other management areas with small, discrete rush skeletonweed infestations, or where an infestation's characteristics affect your ability to meet management objectives, your weed management strategy might have to include more extensive mapping and analysis of the scope of the infestations (e.g. size, density, cover, or location in relation to roads and waterways over time) (Figure 4-2b).

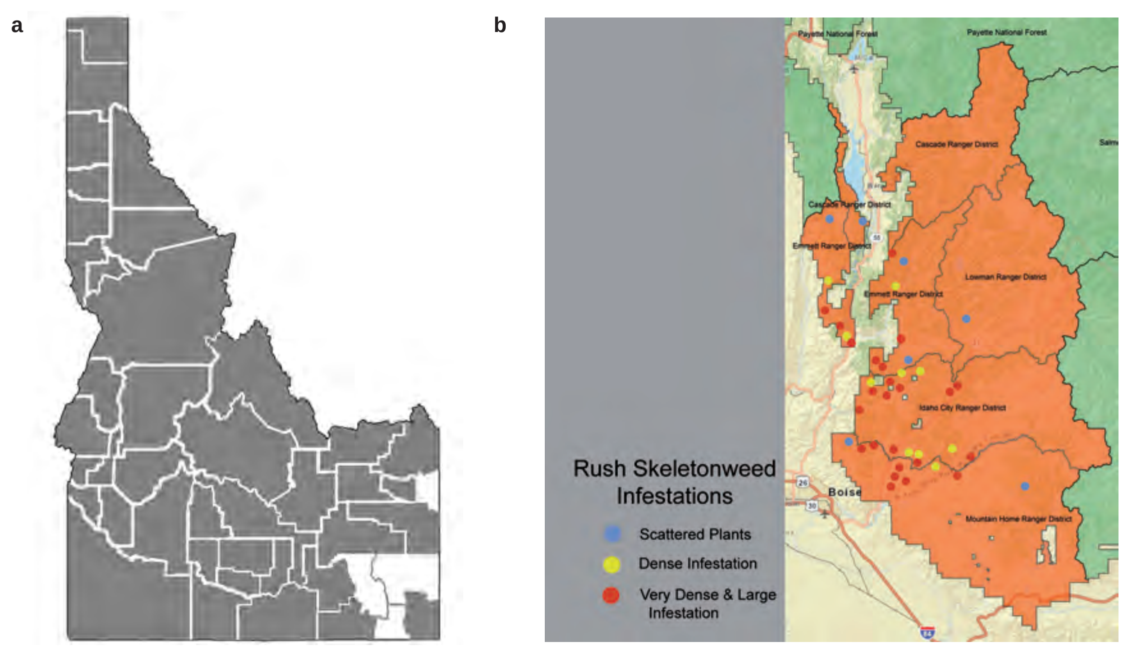


Figure 4-2. Rush skeletonweed data for: a. counties with rush skeletonweed in the state of Idaho (EDDMapS); b. hypothetical infestations in Idaho's Boise National Forest.

In many cases, it may prove useful to check for existing rush skeletonweed distribution data before collecting your own. Several agencies and organizations maintain weed distribution databases, including state agricultural departments, provincial ministries (e.g., British Columbia IAPP Application), invasive plant/species councils, USDA PLANTS database, EDDMapS, and many others. EDDMapS can be particularly useful for land mangers interested in creating rush skeletonweed distribution maps for their area. By visiting www.eddmaps.org and creating a free account, users can view existing distribution maps for rush skeletonweed or other weeds at the state, county, or point level. By selecting the GIS view option, users can view rush skeletonweed data on various backgrounds and zoomed into different scales, add hand drawn labels, boundaries, points and other shapes to the map, perform measurements such as perimeter estimates or distance between points, add new rush skeletonweed data from user shapefiles, edit the management status of various infestations, and print finished maps (see page 62 for more information on EDDMapS).

Defining Goals and Objectives

Goals broadly define the "what" or desired outcome of management; objectives define the "how" or specific activities through which desired outcomes can be achieved. To be effective, objectives must be SMART: specific, measurable, achievable, realistic, and timely. Defining your weed management goals and objectives is the crucial first step in developing a successful biological control program. By defining what you want to achieve, you will be able to determine if, when, and where you should use biological control.

As precisely as possible, you must define what will constitute a successful rush skeletonweed management program. For example, the objective of "…a noticeable reduction in rush skeletonweed density over the next ten years…" might be achievable, but it uses a subjective measurement of success that is open to observer bias. Alternatively, the objective of "…a 50 percent reduction in rush skeletonweed stems over the next three years…" is objectively measurable (and therefore SMART). If your goal is to reduce the abundance of rush skeletonweed, then biological control might be an appropriate weed management tool; however, by itself biological control will not completely and permanently remove rush skeletonweed from the landscape. If your goal is to eradicate rush skeletonweed, then you should plan to employ other weed control techniques instead of, or in addition to, biological control (see Chapter 5 for more details).

Understanding Rush Skeletonweed Management Options

Once you determine the scope of your rush skeletonweed infestations and define your overall program goals, review all the weed control methods available (biological control, physical treatments, cultural practices, and herbicides), and determine the conditions (when, where, if, etc.) under which it might be appropriate to use each method individually or in combination (see Chapter 5). Consult commercial, agency, or university biological control experts, cooperative weed management area partners, or county weed coordinator/supervisors to learn about other rush skeletonweed management activities (herbicide use, grazing, etc.) underway or planned for your area, and the level and persistence of control that might be achieved by each.

Identify the resources that will be available for weed management activities, and determine if they will be consistently available until you meet your weed management program objectives. If resources are not currently available, or will not be available consistently, identify what will happen at the treatment site if planned management activities are not implemented. This information will help you determine the best management activities to use as you initiate and continue your integrated rush skeletonweed management program.

With a map of rush skeletonweed infestations in your management area, an understanding of your land management goals, well defined weed management objectives, and a list of the weed control methods available with the level of control you can realistically expect from each, you can identify sites where biological control would be a good fit, alone or in combination with other control methods.

Developing, Implementing, and Managing a Rush Skeletonweed Biological Control Program

When biological control is deemed suitable for treating your rush skeletonweed infestations, there are several important factors to consider. These include selecting appropriate release sites, obtaining and releasing biocontrol agents, and monitoring the success of the program. Familiarity with all aspects of a biocontrol program before beginning will greatly facilitate its implementation and increase its chances of success. These items are discussed in their own sections below. If problems are encountered following the initiation of a biological control program, refer to the troubleshooting guide in Appendix I for potential solutions.

Selecting Biological Control Agent Release Sites

Establish Goals for your Release Site

You must consider your overall management goals for a given site when you evaluate its suitability for the release of biological control agents. Suitability factors will differ depending on whether the release is to be:

1. a general release, where biological control agents are simply released for rush skeletonweed management,
2. a field insectary (nursery) release, used primarily to mass produce biological control agents for redistribution to other sites, or
3. a research release, used to investigate biological control agent biology and/or the biocontrol agent's impact on the target weed and nontarget plant community.

A site chosen to serve one of the roles listed above may also serve additional functions over time (e.g., biological control agents might eventually be collected for redistribution from a research or general release).

Determine Site Characteristics

For practical purposes, no rush skeletonweed infestation is too large for biocontrol releases; however, it might not be large enough (Figure 4-3a). Very small, isolated patches of rush skeletonweed may not be adequate for biological control agent populations to build up and persist and are often better treated with other weed control methods, such as herbicides or physical control. An area with at least 1 acre (0.40 hectares) of rush skeletonweed is the minimum size to better ensure a successful biological control agent release site, but larger infestations are more desirable (Figure 4-3b), especially if the land manager hopes to someday use the release site as a field insectary. Biocontrol agents disperse more easily in contiguous rush skeletonweed infestations than in infestations with only a few scattered plants and distant patches. If your biological control program goals involve evaluating the program's efficacy, establish permanent monitoring sites before you release any biocontrol agents. The monitoring sites will require regular inspections, so consider the site's ease of accessibility, terrain, and slope.

Note Land Use and Disturbance Factors

Release sites should experience little to no regular disturbance. Abandoned fields/pastures and natural areas are good choices for biological control

Figure 4-3. Rush skeletonweed infestations: a. too small for biological control; b. appropriate for biological control. (a,b: Rachel Winston, MIA Consulting)

agent releases. Sites where insecticides are used should not be utilized for biocontrol agent releases. Such sites include those near wetlands that are subject to mosquito abatement, rangelands that are subjected to grasshopper control, or near agricultural fields or orchards where pesticide applications occur regularly. Roadside infestations along dirt or gravel roads with heavy traffic should also be avoided; extensive dust makes rush skeletonweed plants less attractive to biocontrol agents and silica may kill larvae. Do not use sites where significant land use changes will take place, such as road construction, cultivation, building construction, and mineral or petroleum extraction. If supply of biocontrol agents is limited, prioritize release sites that are not regularly burned or treated with herbicides.

Survey for Presence of Biological Control Agents

Always examine your prospective release sites to determine if rush skeletonweed biological control agents are already present. If a biocontrol agent you are planning to release is already established at a site, you may want to consider making the release at another site where the biocontrol agent is not yet present. If observed biocontrol agent populations are low at a site, you can release additional biocontrol agents at that site to augment the existing population.

Record Ownership and Access

If you release biological control agents on private land, it is a good idea to select sites on land likely to have long-standing, stable ownership and management. Stable ownership will help you establish long-term agreements with a landowner, permitting access to the sites to sample or harvest biological control agents and collect biocontrol agent and vegetation data for the duration of the project. This is particularly important if you are establishing a field nursery site, because five years or more of access may be required to complete biocontrol agent harvesting or data collection. General releases of biological control agents to control rush skeletonweed populations require less-frequent and short-term access; you may need to visit such a site only once or twice after initial release. When releasing biocontrol agents on private land, it may be a good idea to obtain the following:

- written permission from the landowner allowing use of the area as a release site,
- written agreement with the landowner allowing access to the site for monitoring and collection for a period of at least six years (three years for establishment and buildup and three years for collection),
- permission to put a permanent marker at the site, and
- written agreement with the landowner that land management practices at the release site will not interfere with biological control agent activity

The above list can also be helpful for releases made on public land where the goal is to establish an insectary. In particular, an agreement should be reached that land management practices will not interfere with biological control agent activity (e.g. spraying or physically destroying the weed infestation). It is often useful to visit the landowner or land manager at the release site annually to ensure they are reminded of the biological control endeavors and agreement.

You may wish to restrict access to release locations, especially research sites and insectaries, and allow only authorized project partners to visit the sites and collect insects. The simplest approach is to select locations that are not visible to or accessible by the general public. To be practical, most if not all of your sites will be readily accessible, so in order to restrict access you should formalize arrangements with the landowner or manager. This will require you to post no-trespassing signs, install locks on gates, etc. (Figure 4-4).

Figure 4-4. "No disturbance" sign. (Alan Martinson, Latah County Weed Control, and Paul Brusven, Nez Perce BioControl Center)

Another consideration is physical access to a release site. You will need to drive to or near the release locations, so determine if travel on access roads might be interrupted by periodic flooding or inclement weather. You might have to accommodate occasional road closures by private landowners and public land managers for other reasons, such as wildlife protection.

Choosing the Appropriate Biological Control Agents for Release

You should consider several factors when considering which biological control agent to release at a site, including biocontrol agent efficacy, availability, and site preferences (Table 5).

Biocontrol Agent Efficacy

Efficacy refers to the ability of the biological control agent to directly or indirectly reduce the population of the target weed below acceptable damage thresholds or cause weed mortality resulting in control. It is preferable to release only the most effective biocontrol agents rather than releasing all biocontrol agents that might be available for a target weed. Consult with local weed biological control experts, neighboring land managers, and landowners to identify the biocontrol agent(s) that appear(s) more effective given local site characteristics and management scenarios.

Table 5. Summary of general characteristics and site preferences of rush skeletonweed biological control agents released in North America

Biocontrol Agent Characteristics				Site Characteristics	
Species	**Part Attacked**	**Efficacy**	**Availability**	**Favorable Conditions**	**Unfavorable Conditions**
Aceria chondrillae Rush skeletonweed gall mite	All above-ground growth	Reduces flowering and seed production in OR and WA by 50-90%; hindered by parasitism in CA and cold in ID; less abundant/effective in BC	Widespread in OR and WA, established but more limited in CA, ID, MT, WY, and BC	Overwintering rush skeletonweed rosettes; undisturbed sites; low predator populations	Extreme winter temperatures with no fall and winter rosettes; cropland with repeated disturbance; high predator populations
Bradyrrhoa gilveolella Rush skeletonweed root moth	Roots	Too early post establishment to know impact; populations increasing	Limited in ID and OR	Sandy, granitic, or loose-textured soils	Clay, silty, or compacted soils
Cystiphora schmidti Rush skeletonweed gall midge	Stems and leaves	Infested plants are stunted and have reduced seed production; however, populations are hindered by parasitism and predation, so overall impact is low	Limited throughout the NW due to parasitism and predation	Warm, dry, open sites with well-drained soil; low amounts of parasites, grasshoppers and other predators	Cold sites with compacted soil; high amounts parasites, grasshoppers and other predators
Puccinia chondrillina Rush skeletonweed rust fungus	All above-ground growth	Varies by weed genotype and site conditions; most effective biocontrol agent in CA and WA where it decreases plant size and seed production; less effective in ID, OR, and BC	Widespread, established in CA, ID, OR, WA, WY and BC	Significant period of dew (4+ hours) during darkness	Lack of humidity and dew, even during darkness

Biocontrol Agent Availability

All four of the USA-approved biological control agents described in this manual are established in the continental USA; however, availability varies greatly between species and sites. The rust fungus *Puccinia chondrillina* is the most widespread of all biocontrol agents and is readily available for collection in the northwestern USA and British Columbia, though it is more effective on some skeletonweed genotypes and in some locations than others. The mite *Aceria chondrillae* is also established at multiple locations in the northwestern USA and British Columbia, but varies in its abundance and impact. It is most abundant in Oregon and Washington. *Cystiphora schmidti* is hindered by parasitism and predation; although it is established in California, Idaho, Montana, Oregon, Washington, and Wyoming, populations are limited. The root moth *Bradyrrhoa gilveolella* is the least abundant of rush skeletonweed biological control agents. Despite several years of releases, it has only recently been confirmed established in Idaho and Oregon, and populations are slowly increasing. It is believed that all releases of *B. gilveolella* in Canada have failed to establish.

Federal and state/provincial departments or commercial biological control suppliers may be able to assist you in acquiring biocontrol agents not yet available but permitted for use in your area (see Obtaining and Releasing Rush Skeletonweed Biological Control Agents, below). In the USA, state departments of agriculture, county weed managers, extension agents, or federal and university weed biological control specialists should be able to recommend in-state collection sites where appropriate. Remember that in the USA, interstate transport of biological control agents requires a USDA-APHIS-PPQ permit (see Regulations for the Transfer of Rush Skeletonweed Biological Control Agents, page 60). Get your permits early to avoid delays.

Release Site Characteristics

General physical site and biological preferences for each biocontrol agent have been developed from anecdotal observations and experimental data. These are listed in Table 5 to help land managers ensure that biocontrol agents are released in sites with suitable conditions.

Obtaining and Releasing Rush Skeletonweed Biological Control Agents

You can obtain rush skeletonweed biological control agents by collecting or rearing them yourself, having someone collect them for you, or by purchasing them from a commercial supplier. This section provides information on collecting and purchasing rush skeletonweed biocontrol agents, with emphasis on *Aceria chondrillae*, *Cystiphora schmidti*, and *Puccinia chondrillina*. The rush skeletonweed root moth, *Bradyrrhoa gilveolella*, is currently less available in the field.

Factors to Consider when Looking for Sources of Biological Control Agents

You do not need to take a “lottery approach” and release all four biological control agents at a site in the hopes that one of them will work. Some biological control agents will not be available even if you want them, and some have been shown to have little or no effectiveness in certain areas. The best strategy is to release the best agent. Ask the county, state, provincial, or federal biological control experts in your area for recommendations of appropriate biological control agents for your particular project.

If available, biological control agents from local sources are best. Using local sources increases the likelihood that biocontrol agents are adapted to the climate and site conditions present and are available at appropriate times for release at your target infestation. Using locally sourced biocontrol agents also reduces the possibility of accidentally introducing biocontrol agent pathogens or natural enemies to your area. Local sources may include neighboring properties or other locations in your and adjacent counties/ districts. Remember that in the USA, interstate transport of biological control agents requires a USDA-APHIS-PPQ permit (see Regulations Pertaining to the Transfer of Rush Skeletonweed Biological Control Agents, page 60). Get your permits early to avoid delays.

Some USA states, counties, and universities have “field days” at productive insectary sites (Figure 4-5). On these days, land managers and landowners are invited to collect or receive locally collected rush skeletonweed biological control agents for quick release at other sites. These sessions are an easy and often inexpensive way for you to acquire biological control agents. They are good educational opportunities as well, because you can often see first-hand the impacts of various biocontrol agents on rush skeletonweed plant communities.

Figure 4-5. Rush skeletonweed field day. (Joseph Milan, BLM)

Typically, field days are conducted at several sites in a state and on several dates. Although designed for intrastate collection and redistribution, out-of-state participants may be welcome to participate (remember that USDA permits are required for interstate movement and release of biological control agents). Contact county weed supervisors, university weed or biological control specialists, or federal weed managers for information about field days in your region.

Collecting Rush Skeletonweed Biological Control Agents

Planning and timing of collection is critical. For all species, it is usually most efficient to scout the potential collection site well in advance to ensure your desired species is present at suitable densities. The species of biological control agent and weather characteristics at your collection and release site will determine the best time in the season to collect. Ensure that all necessary collection supplies are on hand. Also, accurate identification of the biological control agents is essential. General guidelines for collecting rush skeletonweed biological control agents are listed in the following sections and in Table 6.

For all species, collect only on a day with good weather. Do not collect in the rain; insects will hide and become difficult to find in rainy weather, excess moisture causes adverse effects, and biocontrol agents may drown in wet collection containers. The only exception to this rule is the rust fungus *Puccinia chondrillina*, for which overcast and rainy days are optimal for collection.

Table 6. Recommended timetable and methods for collecting rush skeletonweed biological control agents in North America. Methods are listed in the order of ease of collection and efficacy.

Biocontrol Agent	Biocontrol Agent Stage	Plant Stage	Timing	Method
Aceria chondrillae Rush skeletonweed gall mite	All stages	Stems bolting; stems flowering; plants mature	Late June to September	Move plant stems infected with mites to uninfected sites
Bradyrrhoa gilveolella Rush skeletonweed root moth	Adult	Stems bolting; stems mature	June to August	Sweep adults from foliage in morning and aspirate into collection container; rear adults indoors
Cystiphora schmidti Rush skeletonweed gall midge	Any stage within gall	Stems bolting; stems flowering; plants mature	June to September	Move galled stems to uninfected sites prior to adult emergence; rear adults indoors
Puccinia chondrillina Rush skeletonweed rust fungus	Any stage	All stages	May to October	Move infected plant stems to uninfected sites; vacuum spores, suspend in water, and spray on uninfected leaves prior to dew period

Collection methods

Transferring infested plants: The most common method for collecting gall midge, rust fungus, and gall mite rush skeletonweed biocontrol agents is to transfer infested plants to uninfected sites. Infested stems can be cut, bundled in groups of 20-50, and moved to new sites where those biocontrol agents are not yet established. See the section “Release as many biocontrol agents as possible” on starting on page 57 for detailed instructions on the proper way to utilize bundled plants at new rush skeletonweed sites. Care should be taken not to spread rush skeletonweed roots or seeds to new sites as this may introduce new genetic material. Care should also be taken to avoid spreading other plant or insect species to new sites as this may inadvertently create future problems.

Aspirating: An aspirator is a device used to suck insects from a surface into a collection vial (Figure 4-6a). An aspirator is used to collect insects out of a sweep net (described below), though it can also be used to take adults of the root moth *Bradyrrhoa gilveolella* directly from rush skeletonweed plants. A variety of aspirators can be purchased from entomological, forestry, and biological supply companies, or you can construct them yourself. For the latter, make sure that tubing reaching your mouth is covered by fine-mesh screening, so that insects and small particles are not inhaled (Figure 4-6b).

Sweep netting: Using a sweep net will be the best method for collecting the root moth Bradyrrhoa gilveolella when populations increase sufficiently in the field. A sweep net consists of a conical canvas or muslin bag held open on one end by a sturdy wire hoop 10-15 inches (25-38 cm) in diameter attached to a handle 3 feet (0.9 m) long (Figure 4-7a). They can be purchased from entomological, forestry, and biological supply companies, or you can construct them yourself. As their name implies, these are heavy duty nets used to “sweep” insects off rush skeletonweed.

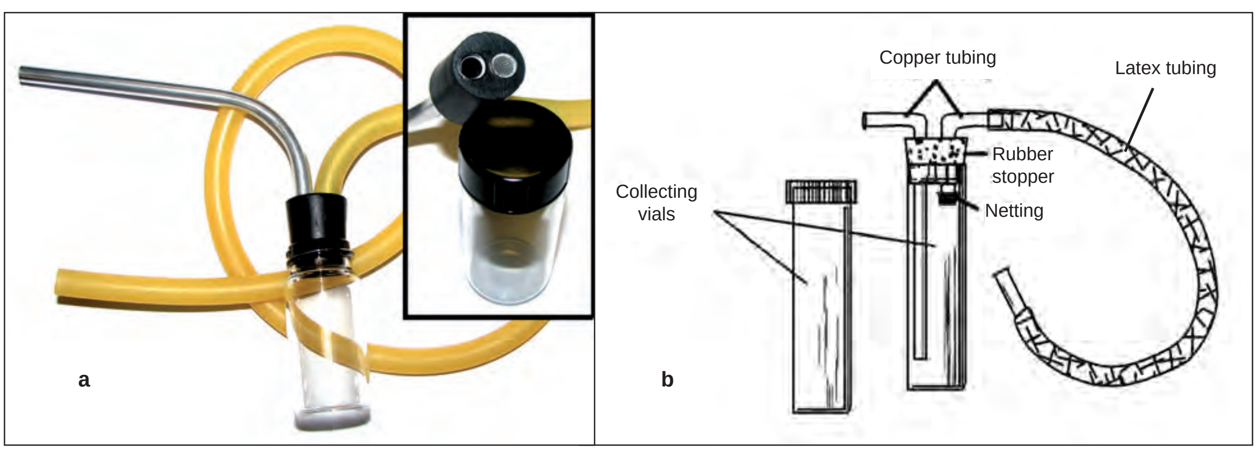


Figure 4-6. Aspirator: a. components (wiki.bugwood.org); b. diagram (Karen Loeffelman, University of Idaho); (a,b: bugwood.org).

A sweep is made by swinging the net through the plant canopy and collecting insects off the foliage (Figure 4-7b). It is best to use no more than 25 sweeps before removing the biocontrol agents from the net. Removing insects at regular intervals reduces the potential harm that could result from knocking biocontrol agents around with debris, and reduces the opportunity for predator insects and spiders swept up with the biocontrol agents from finding and devouring the biocontrol agents.

Methods by species

Gall mite (*Aceria chondrillae*): Rush skeletonweed plants infested with galls can be gathered from late summer through fall. Stems should be cut, bundled in groups of 20-50, tied at both ends, and moved to new sites where the gall mite is not yet established. As galls dry, mites will relocate to uninfested stems. See the section “Release as many biocontrol agents as possible” starting on page 57 for detailed instructions on the proper way to utilize bundled plants at new rush skeletonweed sites. Care should be taken not to spread rush skeletonweed roots or seeds to new sites as this may introduce new genetic material. Care should also be taken to avoid spreading other plant or insect species to new sites as this may inadvertently create future problems.

Root moth (*Bradyrrhoa gilveolella*): Because the root moth is not widely established at present, it may be necessary to obtain the moths from research or professional rearing operations until field populations have built up sufficiently to allow for collection. Once field populations become larger, adults can be swept from vegetation in the morning in spring through late

Figure 4-7. Sweep net: a. closeup (Laura Parsons, University of Idaho); b. being used to collect rush skeletonweed biocontrol agents (Joseph Milan, BLM).

summer. In very early morning, mostly males are active; both sexes are active from mid- to late morning. Take care to collect prior to the majority of egg-laying, or redistribution will be fruitless. Also be aware that sweeping can damage these fragile moths. Aspirating them from the sweep net can greatly reduce damage to the moths.

Alternatively, harvest the roots of infected skeletonweed in the fall and store them at 39-46 °F (4-8 °C). Two to three weeks prior to their normal emergence time, bring them to room temperature in rearing cages or breathable, clear containers. Once they emerge, adults can be transferred to new sites. The latter method is only plausible if precise attack symptoms can be recognized.

Gall midge (*Cystiphora schmidti*): The gall midge is most easily collected by gathering rush skeletonweed stems infested with galls from midsummer through early fall. Stems should be cut, bundled in groups of 20-50, tied at both ends, and moved to new sites where the gall midge is not yet established. Emerging midges will attack the new plants upon emergence. See the section "Release as many biocontrol agents as possible" starting on page 57 for detailed instructions on the proper way to utilize bundled plants at new rush skeletonweed sites. Care should be taken not to spread rush skeletonweed roots or seeds to new sites as this may introduce new genetic material. Care should also be taken to avoid spreading other plant or insect species to new sites as this may inadvertently create future problems.

Transferring infested galls may transfer unwanted parasitoids of the gall midge. To avoid this, gall-infested skeletonweed stems can be collected and midge adults reared out indoors. This can be accomplished by collecting plants infested with galls in the fall and storing them at 39-46 °F (4-8 °C) over the winter. Two to three weeks prior to their normal emergence time, bring them to room temperature in rearing cages or breathable, clear containers. Any parasitoids that emerge should be separated and destroyed. Once midges emerge in spring, they can be transferred to new rush skeletonweed infestations.

Rust fungus (*Puccinia chondrillina*): The preferred method for redistributing the rust fungus is to transfer infested stems. From spring through fall, infested stems should be cut, bundled in groups of 20-50, tied at both ends, and moved to new sites in the evening; uninfected skeletonweed plants should be sprayed with water to increase inoculation success. See the section "Release as many biocontrol agents as possible" starting on page 57 for detailed instructions on the proper way to utilize bundled plants at new rush skeletonweed sites. In fall and spring, whole infected skeletonweed plants can be transplanted to new sites. Care should be taken not to spread rush skeletonweed roots or seeds to new sites as this may introduce new genetic material. Care should also be taken to avoid spreading other plant or insect species to new sites as this may inadvertently create future problems.

Alternatively, spores can be vacuumed from infected rush skeletonweed leaves throughout the growing season suspended in a carrier (typically distilled water and a surfactant) and sprayed on new (uninfected) rush skeletonweed foliage prior to a dew period. Because the methods for spore collection, suspension, and application are varied and more consistent with a bioherbicide, we do not attempt to describe them in this manual. For more information, contact your local biocontrol specialist.

Release Containers for Rush Skeletonweed Biological Control Agents

The manner in which biological control agents are handled during transportation to the release site will affect whether they will survive and multiply at the new site. To reduce mortality or injury, it is best to redistribute the biocontrol agents the same day they are collected.

Transferring biocontrol agents in bundles of long plant stems

Following collection, biological control agents need to be transferred to release containers intended to protect them (and to prevent biocontrol agents from escaping en route). When large sections of infected stems (minus plant propagules such as roots and flowers) are transferred between sites to redistribute the mite, gall midge, or rust, the stems should be stored in large paper bags. Paper bags provide sufficient ventilation while plastic bags may cause moist plant material to rot or down the midges or mites. We do not recommend transferring biological control agents on whole plants as whole plants may be capable of introducing new rush skeletonweed genetic material to the release site.

Transferring biocontrol agents in small plant segments or biocontrol agent adults

When only small infected plant segments are used to transfer the mite or gall midge, or when transferring adult *Bradyrrhoa gilveolella* or *Cystiphora schmidti*, release containers should be rigid enough to resist crushing but also ventilated to provide adequate airflow and reduce condensation. Un-waxed paperboard cartons are ideal; they are rigid, permeable to air and water vapor, and are available in many sizes. As an alternative, you can use release containers made of either light-colored lined or waxed paper (e.g., ice cream cartons or fountain drink cups; see Figure 4-8a) or plastic, providing they are ventilated; simply poke numerous holes in the container or its lid with an ordinary push pin or thumb tack, and cover the holes with a fine mesh screen (Figure 4-8b). Untreated paper bags (lunch bags) work well for transporting biocontrol agents short distances; however, they are fragile and offer little physical protection for the material within, must be sealed tightly to prevent biocontrol agents from escaping, and some biocontrol agents are capable of chewing through them. **Do not use glass or metal release containers**; they are breakable and make it difficult to regulate temperature, airflow, and humidity.

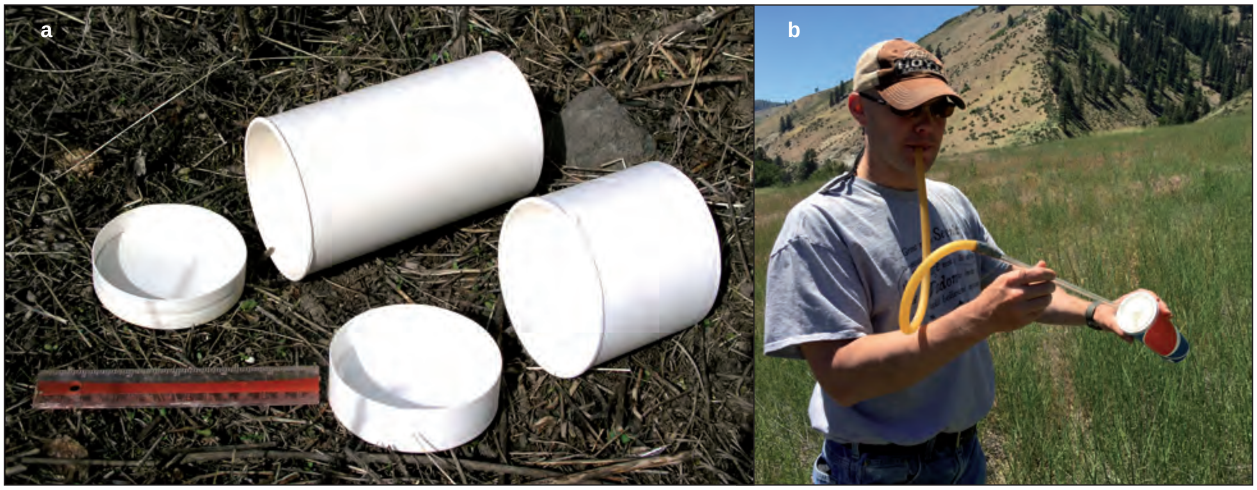


Figure 4-8. Release containers for transporting rush skeletonweed biocontrol agents: a. cardboard (Martin Moses, University of Idaho, bugwood.org); b. fountain drink cup used as the collection container for a homemade aspirator and subsequently closed for transporting the biocontrol agents (Joseph Milan, BLM).

When transferring small plant segments infected with the mite or gall midge, the rush skeletonweed segments should be free of roots, seeds, flowers, dirt, spiders, and other insects and should not be placed in water in the release container. When transferring adult *Bradyrrhoa gilveolella* or *Cystiphora schmidti*, fill release containers two-thirds full with crumpled paper towels or tissue paper to provide a substrate for the insects to rest on and hide in, and to help regulate humidity. Include a few fresh sprigs of rush skeletonweed foliage before adding the biocontrol agents. Again, ensure the sprigs are free of roots, seeds, flowers, dirt, spiders, and other insects. Paper towels or tissue paper can also be added to release containers with small plant segments infested by the mite and gall midge to help regulate humidity and to fill the space, preventing infested plant segments from shifting excessively and becoming damaged.

Seal the release container lids with masking or label tape or with tightly fitting rubber bands. If you are using paper bags, fold over the tops several times and staple them shut. Be sure to label each container with (at least) the biological control agent(s) name, the number of biological control agents in the container, the collection date and site, and the name of the person(s) who did the collecting.

Transporting Rush Skeletonweed Biological Control Agents

Keep the containers cool at all times

Once you collect and package the biocontrol agents, maintain them at temperatures between 50 and 80 °F (10-27 °C). If possible, place the release containers in large coolers equipped with frozen ice packs. Do not use ice cubes unless they are contained in a separate, closed, leak-proof container. Wrap the ice packs in crumpled newspaper or bubble wrap to prevent direct contact with release containers and to absorb any condensation that forms. Place extra packing material in coolers to prevent ice packs from shifting and damaging biocontrol agent containers. As an alternative to coolers with ice packs, electric car-charged coolers may be utilized, provided the cycle is set to cool and not warm. Always keep coolers out of direct sun, and only open them when you are ready to release the biocontrol agents. If you cannot release them immediately, place them in a refrigerator for short-term storage (no lower than 40 °F or 4.4 °C) until you transport or ship them (which should occur as soon as possible and preferably not longer than 48 hours).

Transporting short distances

If you can transport your biocontrol agents to their release sites within 3 hours after collection, and release them the same day or early the next, you need not take any measures other than those already described.

Shipping long distances

If you will be shipping your biocontrol agents to their final destination, use a bonded carrier service with guaranteed overnight delivery (e.g., USPS, FedEx, UPS, or DHL) and send the recipient the tracking number for the package. In such cases, the release containers should be placed in insulated shipping containers with one or more ice packs. Some specially designed foam shippers have pre-cut slots to hold small biocontrol agent containers and ice packs (Figure 4-9). This construction allows cool air to circulate but prevents direct contact between the ice and the release containers. Laboratory and medical suppliers sell foam “bioshippers” that are used to transport

Figure 4-9. Commercially made shipping container. (University of Idaho, bugwood.org)

medical specimens or frozen foods. If neither foam product is available, you can use a heavy-duty plastic cooler, which also may be better suited to large rush skeletonweed stems infected with the rust, mite, or gall midge. **Please note that for safety reasons, dry ice cannot be used for transporting biocontrol agents.**

Careful packaging is very important regardless of the shipping container you use. Ice packs need to be wrapped in crumpled newspaper, wrapping paper, or bubble wrap, and should be firmly taped to the inside walls of the shipping container to prevent them from bumping against and possibly crushing the release containers during shipping. Empty spaces in the shipping container should be loosely filled with crumbled or shredded paper, bubble wrap, packing “peanuts,” or other soft, insulating material. Use enough insulation to prevent release containers and ice packs from shifting during shipment, but not so much that air movement is restricted. Enclose all paperwork accompanying the biocontrol agents (including copies of permits and release forms) before sealing the shipping container. For additional security and protection, you may place the sealed shipping containers or coolers inside cardboard boxes.

Other factors to consider

- Make your overnight shipping arrangements well before you collect your biological control agents, and make sure the carrier you select can guarantee overnight delivery.
- Plan collection and packaging schedules so that overnight shipments can be made early in the week. Avoid late-week shipments that may result in delivery on Friday through Sunday, potentially delaying release of the biocontrol agents for several days.
- Clearly label the contents of containers and specify that they are living organisms.
- Check with a prospective courier to make sure that they can accept this type of cargo and will not treat the packages in ways that could harm the biological control agents. If the courier cannot guarantee that such treatments will not occur, choose a different carrier.
- Contact personnel at the receiving end, tell them what you are shipping and when it is due to arrive, provide a tracking number, verify that someone will be there to accept the shipment, and instruct them not open the container prior to releasing the biocontrol agents. The shipping container should be placed in a refrigerator until the biocontrol agents can be released (as soon after receipt as possible).

Common Packaging Mistakes

Crushing: Secure all material included in the shipping container so that blue ice, bundles of plant material, etc., do not become loose and move around in transit thereby crushing, tearing, or popping open release containers and killing or scattering the biocontrol agents inside.

Escape: Seal release containers securely with rubber bands or easily removable/resealable tape (e.g., masking tape) to prevent mobile biocontrol agents from escaping into the shipping container.

Excess heat: Do not expose release containers to direct sunlight or temperatures above 80 °F (27 °C). Avoid shipping delays that can expose biocontrol agents to high temperatures.

Excess moisture: Remove spilled or excess water in release and shipping containers. Do not ship weed sprigs with any type of water source (e.g., floral foam or tubes) inside release containers. Add crumpled paper towels to release containers to absorb incidental moisture or condensation.

Lack of ventilation: Provide adequate ventilation; use air-permeable release containers or make air holes in plastic containers with push pins or other small diameter tools, covering the holes with a fine mesh screen to prevent the escape of mobile biocontrol agents.

Stress: Provide crumpled paper towels and rush skeletonweed sprigs in containers with adult *Bradyrrhoa gilveolella* or *Cystiphora schmidti* so the adults can shelter; avoid over-crowding.

Purchasing Rush Skeletonweed Biological Control Agents

A number of commercial suppliers provide rush skeletonweed biological control agents. In the USA, county weed managers, extension agents, or university weed or biological control specialists may be able to recommend one or more suppliers. Make sure that a prospective supplier is reputable, can provide copies of required permits, can provide healthy colonies individuals of the species you want (parasite- and pathogen-free), and can deliver them to your area at a time appropriate for field release (you will want to know where and when the biocontrol agents were collected). Avoid purchasing biocontrol agents from a supplier who collects biocontrol agents from an environment significantly different from your planned release location. Interstate shipments of rush skeletonweed biological control agents by commercial suppliers also require a USDA permit, a copy of which should be enclosed in the shipping box (see page 60). Confirm in advance that there is a permit in place for the species you are acquiring as well as the region in which the release will occur. DO NOT purchase or release unapproved or non-permitted biological control organisms. Note that before any biocontrol agents can be taken across national borders, whether collected or purchased, an importation permit from the regulatory agency of the receiving country is required (USDA-APHIS in the USA and CFIA in Canada).

Releasing Rush Skeletonweed Biological Control Agents

Establish permanent location marker

Place a steel fence post or plastic/fiberglass pole as a marker at the release point (Figure 4-10a). Avoid wooden posts; they are vulnerable to weather and decay. Markers should be colorful and conspicuous. White, bright orange, pink, and red are preferred over yellow and green, which may blend into surrounding vegetation. Where conspicuous posts may encourage vandalism, mark your release sites with short, colorful plastic tent/surveyor's stakes or steel plates that can be tagged with release information and located later with a metal detector and GPS. Depending on the land ownership or management status at the release site, it may be necessary to attach a sign to the post or pole indicating a biological control release has occurred there and that the site should not be sprayed with chemicals or be mechanically disturbed (see Figure 4-4 on page 42). Where a sign is appropriate, the landowner/land manager and the local weed management authority (county, state, federal, and/or provincial) should be notified and given a map of the release location.

Record geographical coordinates at release point using GPS

Map coordinates of the site marker should be determined using a global positioning system device (GPS) or a GPS-capable tablet/smartphone. There are numerous free apps available for recording GPS coordinates on a tablet/smartphone (Figure 4-10b). Coordinates should complement but not replace a physical marker. Accurate coordinates will help re-locate release points if markers are damaged or removed. Along with the coordinates, be sure to record what coordinate system and datum you are using, e.g., latitude/longitude in WGS 84 or UTM in NAD83.

Figure 4-10. Biocontrol agent release site tools: a. permanent marker; b. smartphone with free weed and biocontrol agent mapping app iBioControl. (a,b: Rachel Winston, MIA Consulting)

Prepare map

The map should be detailed and describe access to the release site, including roads, trails, and unique landmarks/terrain features that are not likely to change through time (e.g., large rocks or rocky outcrops, creeks, valleys, etc.). Avoid using ephemeral landmarks such as “red bush”, “grazing cows”, etc., and descriptors which may not be obvious to everyone, such as “the Miller place”, or “where the old barn used to be”, etc. Use your vehicle’s trip odometer to measure and record mileage between specified locations on your map, e.g., when you turn on to a new road, at cattle guards along the route, and where you park. The map should complement but not replace a physical marker and GPS coordinates. Maps are especially useful for long-term biological control programs in which more than one person will be involved or participants are likely to change. Maps are often necessary to locate release sites in remote locations or places physically difficult or confusing to access.

Complete relevant paperwork at site

Your local land management agency/authority may have standard biocontrol agent release forms for you to complete. Typically, the information you provide includes a description of the site’s physical location, including GPS-derived latitude, longitude, and elevation; a summary of its biological and physical characteristics and land use; the name(s) of the target weed and biocontrol agent(s) released; date and time of the release; weather conditions during the release; and the name(s) of the person(s) who released the biocontrol agents (see Sample Biological Control Agent Release Form in Appendix II). The best time to record this information is while you are at the field site. Consider using a smartphone and reporting app such as iBioControl. This free application uses EDDMapS (see page 62 for more information) to help county, state, and federal agencies track releases and occurrences of biological control agents of noxious weeds. Once back in the office, submit the information to your local weed control office, land management agency, or other relevant authority/database. **Always keep a copy for your own records**.

Set up photo point

A photo point is used to visually document changes in rush skeletonweed infestations and the plant community over time following the release of biocontrol agents. Use a permanent feature in the background as a reference point (e.g., a mountain, large rocks, trees, or a permanent structure) and make sure each photo includes your release point marker. Pre- and post-release photographs should be taken from roughly the same place and at the same time of year. Label all photos with the year and location.

Release as many biocontrol agents as possible

As a general rule of thumb, it is better to release many individuals of a biocontrol agent species at one rush skeletonweed infestation than it is to spread those individuals too thinly over multiple rush skeletonweed infestations. Releasing all the biocontrol agents within a release container in one spot will help ensure that adequate numbers of males and females are present for reproduction and reduce the risks of inbreeding and other genetic problems. Guidelines for a minimum release size are uncertain for most biocontrol agents, but releases of 50-100 adult Bradyrrhoa gilveolella and 50-100 adult *Cystiphora schmidti* (or more) are encouraged.

Often, a single release will be sufficient to establish an insect population, especially if a large number of individuals are released. The only way to determine if biocontrol agents have established is to inspect them annually for up to 5 years (or more) after releases are made. Subsequent releases may be necessary if initial releases fail to establish. For species or locations where establishment is likely to be slow (e.g., due to high levels of overwintering mortality), planning to make releases on the same site for 2 or 3 consecutive years may increase successful establishment and reduce the time until biocontrol agent impact on target weed populations is seen. If more than one release is available in a given year, be sure to put some distance between releases; 1 km (2/3 mile) is ideal. If possible, make more than one release per drainage or in adjoining drainages; if one of your releases is wiped out by flooding, fire, herbicide application, or other catastrophic disturbance, then biocontrol agents from adjoining releases can repopulate it.

In general, you can release biocontrol agents either in open releases or cages. For open releases, get to the desired release location and open the release container. When releasing adult *Bradyrrhoa gilveolella* or *Cystiphora schmidti*, gently shake out all biocontrol agents in one small area, taking care to dislodge any insects hiding in or clinging to the paper towels in the release containers. When releasing small rush skeletonweed segments infested with *Aceria chondrillae*, *Cystiphora schmidti* galls, or *Puccinia chondrillina*, first ensure the segments have no rush skeletonweed root fragments or seeds and that there are no other insect or plant species in the release containers. Gently shake out all infested plant segments in one small area. Do not scatter biocontrol agents or small plant segments throughout the infestation. Do not walk back over the area where you just made a release.

When releasing by transferring large rush skeletonweed stems infested with *Aceria chondrillae*, *Cystiphora schmidti* galls, or *Puccinia chondrillina*, first ensure collected stems have no rush skeletonweed root fragments or seeds or other insects or plant species. Take bundles of 20-50 stems and remove the ties on one end of each bundle so that stems can be fanned out at the loose end, providing a supportive base (Figure 4-11). Place the fanned bundles upright within dense stands of uninfested rush skeletonweed. In less dense infestations or at windy locations, tying the fanned bundle against uninfested rush skeletonweed may aid in successful establishment. Four to five bundles should be used per site, though more or fewer may be required, depending on the infestation size. When transferring stems infested with the rust *P. chondrillina*, the transfer should take place in the evening, and uninfected skeletonweed plants should be sprayed with water to increase inoculation success.

Figure 4-11. Large rush skeletonweed stems bundled for the redistribution of *Aceria chondrillae*, *Cystiphora*, or *Puccinia chondrillina* and fanned out at the bottom end to provide a supportive base. (Joseph Milan, BLM)

When *P. chondrillina* spores have been vacuum-collected from rush skeletonweed foliage, a suspension can be made by combining spores with a carrier (typically distilled water and a surfactant) and sprayed on new (uninfected) rush skeletonweed foliage prior to a dew period.

Caged releases (appropriate for *Bradyrrhoa gilveolella*) confine biocontrol agents for a period of time so they adjust to the site and easily find one another. They may help increase establishment success at new locations, but they require you to put up and take down equipment. For caged releases, place a mesh bag over a rush skeletonweed plant (Figure 4-12a) or a caged area containing multiple plants (Figure 4-12b). Release the adult moths inside the cage, and secure the bottom of the cage to either the stem or the ground. Cages should be removed within a few days (for plants) or weeks (for areas).

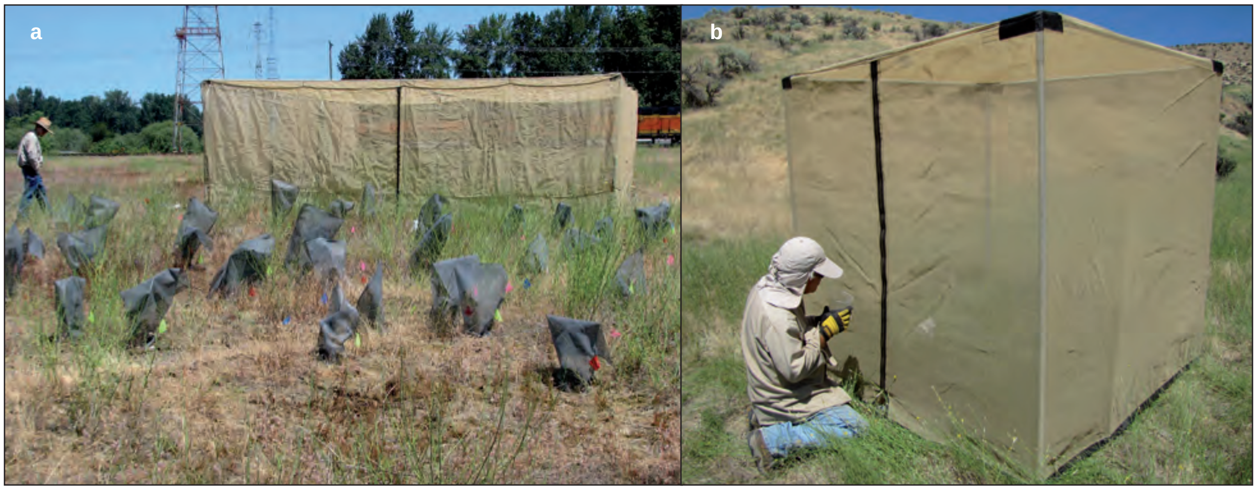


Figure 4-12. Caged releases of *Bradyrrhoa gilveolella*: a. on individual rush skeletonweed plants (foreground) (Gary Brown, USDA APHIS PPQ; bugwood.org); b. in large screen cage with multiple rush skeletonweed plants (Joseph Milan, BLM).

Releases of adult moths and midges or of rust-infested plant material should be made under moderate weather conditions (mornings or evenings of hot summer days, mid-day for cold season releases). Making releases under these conditions reduces the immediate dispersal of stressed insects when they are dumped out of release containers, and the milder temperatures are more conducive to successful rust establishment. Transferring plants infested with first generation overwintering mites, virulent rust strains, and midges that have not been exposed to predation is ideal, especially when transfers are made early in the year. Avoid making releases/transfers on rainy days, unless dealing with the rust, which is aided by moist conditions. If you encounter an extended period of poor weather, it is better to release the biological control agents than wait three or more days for conditions to improve as the biocontrol agents' vitality may decline with extended storage.

Regulations for the Transfer of Rush Skeletonweed Biological Control Agents

USA, intrastate: Generally, there are few if any restrictions governing the collection and shipment of approved biological control agents within the same state; however, you should check with your state's department of agriculture or agriculture extension service about regulations governing the release and intrastate transport of your specific biological control agent. The state of California regulates release permits at the county level.

USA, interstate: The interstate transportation of biological control agents is regulated by the U.S. Department of Agriculture (USDA), and a valid permit is required to transport living biological control agents across state lines. You should apply for a Plant Protection and Quarantine (PPQ) permit from the Animal and Plant Health Inspection Service (APHIS) as early as possible—but at least six months before actual delivery date of your biological control agent. You can check the current status of regulations governing intrastate shipment of weed biological control agents, PPQ Form 526 at the USDA-APHIS-PPQ website. The ePermit process can be accessed by doing an internet search for "USDA APHIS 526 permit application". This allows the complete online processing of biological control agent permit requests.

Canada: Canada requires an import permit for any new biological control agent or shipments from overseas of previously released agents. Permits are issued by the Plant Health Division of the Canadian Food Inspection Agency. Redistribution within a province (or even within Canada) of weed biological control agents that have been officially approved for use in Canada is generally allowed; however, you should consult with provincial and federal authorities and specialists prior to moving any weed biological control agent between areas (e.g., from the prairies to the interior or coast of British Columbia). Accidentally introduced biocontrol agents that have become adventive in a region, or native organisms that may feed on a weed targeted for control should not be moved to new areas without consulting federal authorities and specialists as their host range or potential ecological impacts are not fully known.

Documenting, Monitoring, and Evaluating a Biological Control Program

The Need for Documentation

The purpose of monitoring is to evaluate the success of your rush skeletonweed biological control program and to determine if you are meeting your weed management goals. Documenting outcomes (both successes and failures) of biocontrol release programs will help generate a more complete picture of biocontrol impacts, guide future management strategies, and serve education and public relations functions. Monitoring can provide critical information for other land managers by helping them predict where and when biological control might be successful, helping them avoid releasing ineffective biocontrol agents or the same biocontrol agent in an area where they were previously released, and/or helping them avoid land management activities that would harm local biocontrol agent populations or worsen the rush skeletonweed problem. (See the Code of Best Practices for Classical Biological Control of Weeds on page 8.)

Monitoring activities utilize standardized procedures over time to assess changes in populations of the biocontrol agents, rush skeletonweed, other plants in the community, and other components of the community. Monitoring can help determine:

- If the biological control agents have become established at the release site
- If biological control agent populations are increasing or decreasing and how far they have spread from the initial release point
- If the biological control agents are having an impact on rush skeletonweed
- If/how the plant community or site factors have changed over time

Monitoring methods can be simple or complex. A single year of monitoring may demonstrate whether or not the biocontrol agents established, while multiple years of monitoring may allow you to follow the population of the biocontrol agents, changes in the target weed population and plant community, and changes in other factors such as climate or soil.

Information Databases

Many federal and state/provincial departments have electronic databases for archiving information about weed biological control releases. We have included a standardized biological control agent release form that, when completed, should provide sufficient information for inclusion in any number of databases (see Appendix II).

The U.S. Forest Service (in conjunction with the University of Georgia, MIA Consulting, University of Idaho, CAB International, and the Queensland Government) also maintains a worldwide database for the Biological Control of Weeds: A World Catalogue of Agents and their Target Weeds. The database includes entries for all weed biocontrol agents released to date, including the year of first release within each country, the biocontrol agents' current overall abundance and impact in each country, and more. This database can be accessed at www.ibiocontrol.org/catalog/.

EDDMapS (Early Detection & Distribution MAPping System) is a web-based mapping system increasingly being used for documenting invasive species as well as biocontrol agent distribution in North America. EDDMapS combines data from existing sources (e.g. databases and organizations) while soliciting and verifying volunteer observations, creating an inclusive invasive species geodatabase that is shared with educators, land managers, conservation biologists, and beyond. Information can be added in online forms through home computers and/or apps created for smartphones. For more information on how to utilize or contribute to these tools, visit www.eddmaps.org/about/ and apps.bugwood.org/.

In addition, some states/provinces have county/district weed departments or employ weed biocontrol specialists, often affiliated with state/province departments of agriculture, county extension offices, or Animal and Plant Health Inspection Service Plant Protection and Quarantine (APHIS-PPQ) offices. Contact local entities for more information.

Monitoring Methods

There are three main components to measure in a rush skeletonweed monitoring program: biological control agent populations, rush skeletonweed populations, and the rest of the plant community (including nontarget plants). More detailed monitoring might also examine effects on other biotic community components (such as other insects, birds, mammals, etc.) or abiotic factors (such as erosion, soil chemistry, etc.). Only the three main monitoring components are discussed in this manual.

Assessing biological control agent populations

If you wish to determine whether or not rush skeletonweed biocontrol agents have established after initial release, you simply need to find the biocontrol agents in one or more of their life stages, or evidence of their presence (Table 7). Begin looking for biocontrol agents where they were first released, and then expand to the area around the release site.

Populations of some biocontrol agents take two or more years to reach detectable levels. Thus if no biocontrol agents are detected a year after release, it does not mean they failed to establish. Revisit the site at least once annually for three years. If no evidence of biocontrol agents is found, either select another site for release or make additional releases at the monitored site. Consult with your county extension educator or local biological control of weeds expert for assistance.

Table 7. Life stages/damage to look for to determine establishment of rush skeletonweed biological control agents.

Biocontrol Agent	Life Stage	Where to Look	When to Look
Aceria chondrillae Rush skeletonweed gall mite	Larvae/ Nymphs/Adults	Shoot tips and buds	All growing season (Apr-Oct)
Bradyrrhoa gilveolella Rush skeletonweed root moth	Adults	Ovipositing females on stems or on soil at base of skeletonweed plants	June-August
	Larvae	Within roots	Year-round
Cystiphora schmidti Rush skeletonweed gall midge	Adults	Ovipositing females on stems or leaves	Growing season (Apr-Sep)
	Larvae	Within galls	Year-round
Puccinia chondrillina Rush skeletonweed rust fungus	All	All above-ground growth	Year-round (there can be multiple cycles per year; symptoms and stages will vary)

(continued on next page)

Table 7 (continued). Life stages/damage to look for to determine establishment of rush skeletonweed biological control agents.

Biocontrol Agent	Most Frequently Observed Damage	Appearance
Aceria chondrillae Rush skeletonweed gall mite	Growing tips and buds covered with tiny galls of enlarged plant tissue; stems stunted and deformed	
Bradyrrhoa gilveolella Rush skeletonweed root moth	Adults typically do not cause any direct damage; plants they emerge from may be wilted and stunted	
	Feeding tubes among roots made of silk, sand and frass	
Cystiphora schmidti Rush skeletonweed gall midge	Adults typically do not cause any direct damage beyond the tissue destruction of emerging from galls	
	Leaves and stems covered with purplish colored galls; attacked plants have fewer branches and seed production	
Puccinia chondrillina Rush skeletonweed rust fungus	Yellowish chlorotic lesions with raised centers, becoming orangish-brown pustules that produce powdery, round, and dark brown/rust-colored spores; infected plants stunted and deformed; infected seedlings and rosettes often killed	

a. Stunted and deformed growth caused by galling and feeding of *Aceria chondrillae* (Biotechnology and Biological Control Agency); b. Adult *Bradyrrhoa gilveolella* on skeletonweed stem (Joseph Milan, BLM); c. *B. gilveolella* larval feeding tube (Laura Parsons and Mark Schwarzländer, University of Idaho); d. Adult *Cystiphora schmidti* on skeletonweed stem (Charles Turner, USDA ARS; bugwood.org); e. *C. schmidti* galls on skeletonweed stems (Gary Piper, Washington State University; bugwood.org); f. Rosette leaf infected by *Puccinia chondrillina* (Joseph Milan, BLM).

A systematic monitoring approach is required to determine the changing densities of biocontrol agent populations. The Standardized Impact Monitoring Protocol (SIMP) is one such approach to monitoring biocontrol agent populations, weed populations, and the surrounding plant community over time (Appendix III). This protocol was developed through cooperation among the Bureau of Land Management, the University of Idaho, U.S. Forest Service Forest Health Protection, the Nez Perce Biocontrol Center, and the Idaho State Department of Agriculture. SIMP was designed to be simple, efficient, and sufficiently versatile to allow for the collection of information from the same sites over multiple years. The rush skeletonweed SIMP system is designed to monitor *Bradyrrhoa gilveolella*, but simple presence/absence guidelines have also been developed for the rush skeletonweed mite, midge, and the rust fungus (see Appendix IV). An alternative general biological control agent monitoring form can be found in Appendix V. Existing data sheets may be modified to meet the needs of each land manager by adding extra columns, descriptive classes, etc.

Assessing the status of rush skeletonweed and co-occurring plants

The ultimate goal of a rush skeletonweed biological control program is to permanently reduce the abundance and vigor of rush skeletonweed and enable the recovery of more desirable vegetation on the site. To determine the efficacy of biocontrol efforts, there must be monitoring of plant community attributes, such as target weed distribution and density. Ideally, monitoring begins before biological control efforts are started (pre-release) and occurs at regular intervals after release. There are many ways to qualitatively (descriptively) or quantitatively (numerically) assess weed populations and other plant community attributes at release sites.

Qualitative (descriptive) vegetation monitoring: Qualitative monitoring uses subjective measurements to describe the rush skeletonweed and the rest of the plant community at the management site. Examples include listing plant species occurring at the site, estimates of density, age and distribution classes, visual infestation mapping (as opposed to mapping with a GPS unit), and maintaining a series of photos from designated photo points over time (Figure 4-13a,b). Qualitative monitoring provides insight into the status or change of rush skeletonweed populations; however, its descriptive nature does not generally allow for detailed statistical analyses. Data obtained in qualitative monitoring may trigger more quantitative monitoring later.

Quantitative vegetation monitoring: Quantitative monitoring measures changes in the rush skeletonweed population as well as the vegetative community as a whole before and after a biocontrol agent release using numbers and statistics. It may be as simple as counting the number of rush skeletonweed stems in a small sample area, or as complex as measuring rush skeletonweed plant height, flower and seed production, biomass, species

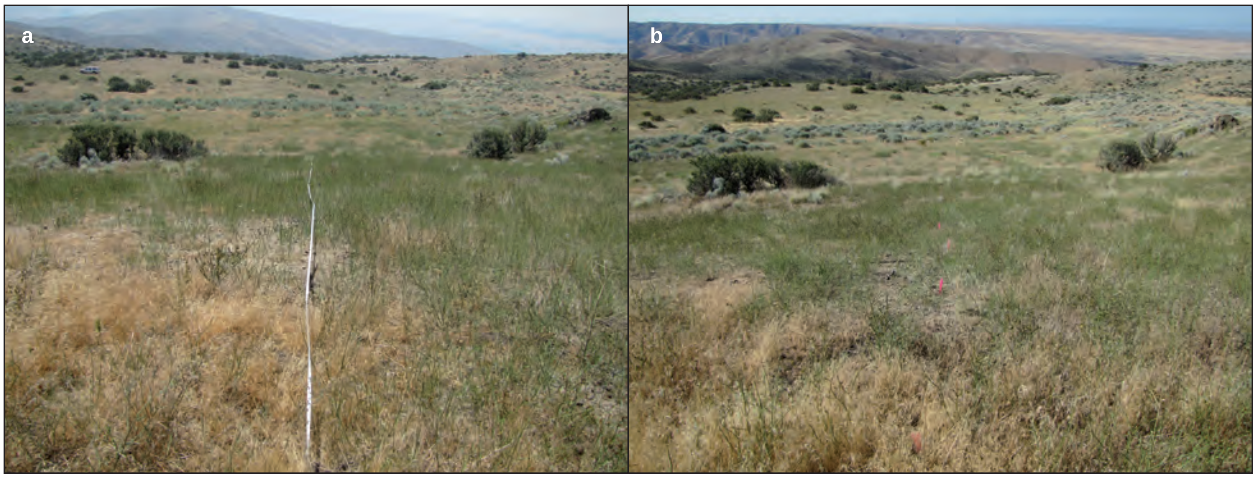


Figure 4-13. Rush skeletonweed biocontrol release site: a. in 2012; b. in 2015 (a,b: Joseph Milan, BLM).

diversity, and species cover (Figure 4-14). Quantitative sampling data can be more readily analyzed using statistical methods and demonstrate significant plant community changes. Pre- and post-release monitoring should follow the same protocol and be employed at the same time of year. Post-release assessments should be planned annually for at least three to five years (and ideally longer than that) after the initial biocontrol agent release.

See Appendix VI for a sample data form where you can record quantitative rush skeletonweed monitoring data along with information on associated vegetation. The SIMP approach described earlier and found in Appendix III is a combination of qualitative and quantitative elements as well as counts for *Bradyrrhoa gilveolella*.

Figure 4-14. Estimating rush skeletonweed coverage. (Rachel Winston, MIA Consulting)

Assessing impacts on nontarget plants

To address possible nontarget attacks on species related to rush skeletonweed, you must become familiar with the plant communities present at and around your release sites and be aware of species related to rush skeletonweed (start with other species in the Asteraceae family and the Cichorieae tribe). You may need to consult with local, state, or regional botanical experts, or review local herbarium records for guidance on areas where related nontarget plants might be growing and additional information on how you can identify them. Care should be taken in the management of your rush skeletonweed biocontrol program to ensure that all closely related native species are identified and monitored along with rush skeletonweed.

If you observe approved biocontrol agents feeding on and/or developing on nontarget plant species, the vegetation sampling procedures described above can be easily modified to monitor changes in density and/or cover of the nontarget species. Concurrently, you may wish to collect additional data, such as the number of biocontrol agents observed on nontarget plants, the amount of foliar attack observed, or the presence of characteristic biocontrol agent damage. Collecting this data over subsequent years can help determine if there is a population level impact or if the nontarget feeding is temporary or of minor consequence to the nontarget species. Please be aware that there are many “look-alike” native insects that feed on related native plants. Correct identification by insect specialists is needed to confirm such records.

If you observe approved biological control agents feeding on and/or developing on native species, collect samples and take them to a biocontrol specialist in your area. Alternatively, you may send the specialist the site data and/or pictures so he or she can survey the site for nontarget impacts. Be sure not to ascribe any damage you observe on native species to any specific species and thus bias the confirmation of attack and the identification of the species causing the attack.

Chapter 5: An Integrated Rush Skeletonweed Management Program

Introduction

The invasion curve (Figure 1-3, repeated here in Figure 5-1) shows that eradication of an invasive species such as rush skeletonweed becomes less likely and control costs increase as an invasive species spreads over time. Prevention is the most cost-effective solution, followed by eradication. If a species is not detected and removed early, intense and long-term control efforts will be unavoidable. Identifying where rush skeletonweed is on the invasion curve in a particular area is the first step to taking management action. Inventorying and mapping current rush skeletonweed populations

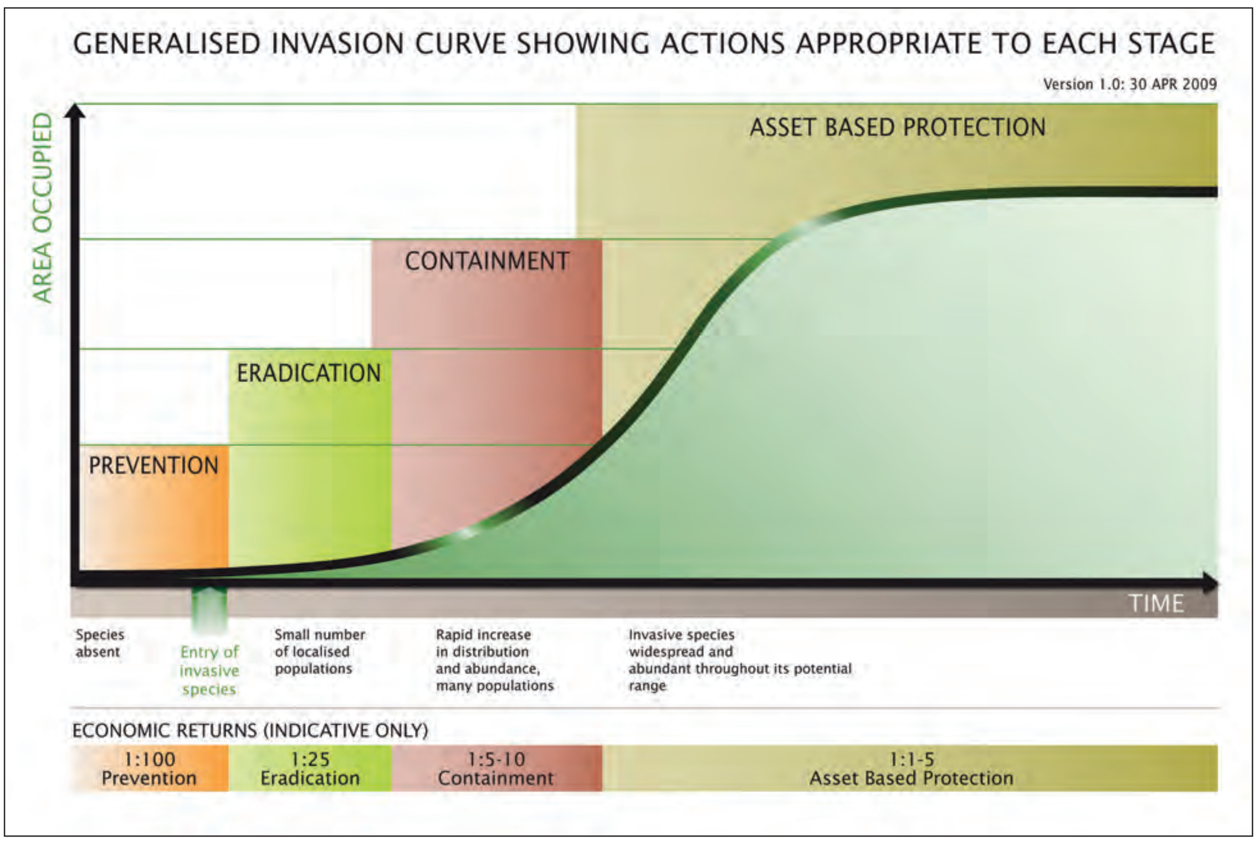


Figure 5-1. Generalized invasion curve showing actions appropriate to each stage. (© State of Victoria, Department of Economic Development, Jobs, Transport and Resources. Reproduced with permission.)

coupled with research efforts to predict where rush skeletonweed is most likely to move enables land managers to concentrate resources in areas where rush skeletonweed is likely to invade, and then to treat individual plants and small populations of rush skeletonweed before it is too late to remove them.

Classical biological control has been applied to many invasive plant species, but biological control is not appropriate for areas on the left side (species absent [prevention] – small number of localized populations [eradication]) of the invasion curve. Biological control as a control method is best suited to rush skeletonweed populations in the later phases of the invasion curve (rapid increase in distribution and abundance [containment] – widespread and abundant throughout its potential range [asset based protection]).

There are several examples in which both single- and multiple-biocontrol agent introductions have successfully controlled the targeted weeds. Where ideally suited, biological control can help maintain rush skeletonweed densities below economically or ecologically significant levels, enabling land managers to live with the weed; however, it may take three to five years or more for biological control to help reduce weed populations to such manageable levels. Furthermore, rush skeletonweed occurs across a wide range of conditions. Some habitats are unsuitable to biocontrol agents, so biological control is not going to work on rush skeletonweed every time at every site. Depending on the infestation, integration with other weed control methods or resorting to other control measures entirely may be required to attain rush skeletonweed management objectives.

A wide variety of successful weed control methods have been developed and may be useful for helping meet management goals for rush skeletonweed. The most successful long-term rush skeletonweed management efforts have a number of common features, including:

- Education and Outreach
- Inventory and Monitoring
- Prevention
- Weed Control Activities: A variety of rush skeletonweed control activities which are selected based on characteristics of the target infestation and planned in advance to use the most appropriate method or combination of methods at each site, including:
    - Biological control
    - Physical treatment
    - Cultural practices
    - Chemical treatment

Integrated Pest Management (IPM) incorporates all efforts noted above, and addresses several aspects of land management, not just how to get rid of weed populations. Land managers or landowners engaged in IPM take the time to educate themselves and others about the threat invasive species pose to the land and how management may facilitate invasion. IPM requires land managers to regularly inventory and map the land they manage, identifying areas where the vegetation is not meeting their management objectives and identifying reasons why. When a weed infestation is found, IPM dictates that land managers map it and make plans to address it utilizing control methods most appropriate for their particular infestation. After initiating control activities, IPM encourages land managers to monitor the site to determine if the control was successful in subsequent years. If re-treatment or additional treatments are necessary, these are applied in a timely manner with appropriate post-treatment monitoring to ensure that management objectives are being met.

Integrated Pest Management programs undertaken on a landscape level over many years can at times prove logistically difficult, expensive, and time-consuming. The concept of Cooperative Weed Management Areas (CWMA) was created in western North America in order to erase jurisdictional boundaries as an impediment to weed control and make a landscape IPM approach to weed management more feasible and successful. CWMAs consist of federal, state and local land managers, as well as concerned private landowners, within a designated zone who join efforts against exotic plants, pooling and stretching limited resources and manpower for managing invasive species and protecting/restoring habitat. Cooperation between neighboring CWMAs helps transfer knowledge and experience between heavily treated regions and places not yet as impacted by rush skeletonweed. Sharing successes and failures in rush skeletonweed management saves time and funding and reduces the incidence of negative impacts from management efforts, such as herbicide resistance. Numerous CWMAs exist throughout the western states of the USA and are excellent sources of information, experience, and resources for treating rush skeletonweed infestations using an IPM approach.

Components of Successful Integrated Pest Management Programs to Manage Rush Skeletonweed

Though each component of IPM is an important tool for managing rush skeletonweed, it is important to note that these components work best when used in a combined approach. Rather than applying only one tool per site (e.g., applying herbicides at one infestation, mowing at another, and using biological control at still another), the most effective IPM strategy is to employ as many tools as possible at a single site in order to maximize the efficacy of each tool and ultimately reduce rush skeletonweed infestations. Education, inventorying/mapping, and prevention are important and applicable across all landscapes, whether or not rush skeletonweed is already present. When rush skeletonweed is established and control methods are warranted, long-term management success is greatly improved when control methods are identified according to infested habitat type, land use, ownership, and available resources and then integrated where appropriate. As described above, biological control is most appropriately used on large infestations where multiple years may be required before impacts are realized. During this time, chemical and physical control methods are best applied to smaller new or satellite populations where immediate eradication is warranted, and to the edges of large infestations to prevent further spread. Cultural control methods work to enhance the growth of more desirable vegetation and are best applied as complements to all other control methods.

The components of rush skeletonweed IPM are described individually below. Because the focus of this manual is the biological control of rush skeletonweed, the potential to integrate biocontrol with other weed control methods is described at the end of each control method's section.

Education and Outreach

Education and outreach activities increase public awareness of noxious weeds, the problems they cause, their distribution, and ways to manage them (Figure 5-2). Ideally, education and outreach activities also foster cooperation and collaboration across land ownership boundaries to facilitate the development of a landscape-level weed management response. Education efforts should be an important component of any weed management plan, regardless of the target weed or weed control method employed.

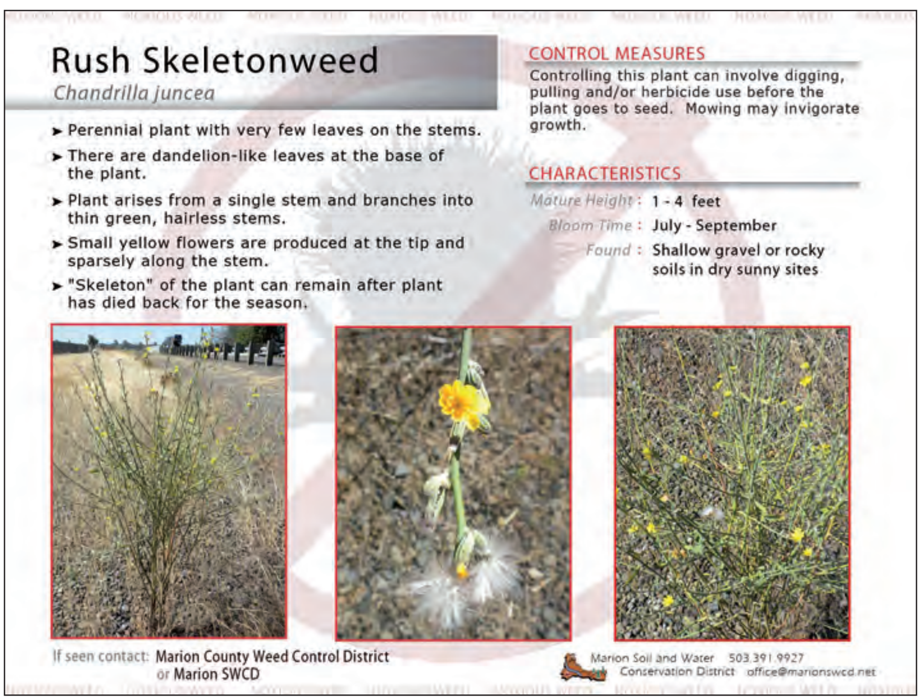


Figure 5-2. Rush skeletonweed education brochure. (Marion Soil and Water Conservation District, Marion, Oregon)

Rush skeletonweed education and outreach should focus on conveying to the public:

- the threat rush skeletonweed poses
- how to identify rush skeletonweed in different stages
- ways in which they can help in rush skeletonweed management

By educating land managers and landowners, recreationalists and the public about the threat of rush skeletonweed, enabling them to identify infestations, and enlisting them in mapping and management efforts, it becomes possible to cooperatively develop successful weed management responses at the landscape level.

Inventory and Mapping

Inventory and mapping are key elements of a successful pest management program. It is imperative to accurately characterize the size and extent of weed infestations before control activities are identified, prioritized, and implemented because the best treatment methods are often determined by the size and location of the infestation. Education and outreach activities that foster collaboration between adjacent landowners are particularly useful when developing landscape-level maps of weed infestations. Once land managers and landowners fully understand the threat rush skeletonweed poses to their land, they are often more willing to participate to ensure that their land is inventoried and accurate maps of rush skeletonweed are developed so the best control activities can be implemented.

Rush skeletonweed infestations are often mapped by foot, vehicle, horse, or airplane using a global positioning system unit (GPS) and a geographical information system (GIS), though hard copy maps made by hand are suitable for some locations. Different infestations are best monitored by different means. Small infestations can be very difficult to spot, given the morphology of RSW stems which make the plant difficult to distinguish from neighboring vegetation (Figure 5-3a). These infestations are often best spotted with small-scale search operations such as those done by foot or on horseback (Figure 5-3b). Larger infestations are more easily spotted by their gray-green appearance, monoculture tendencies and (during late summer) the presence of numerous scattered yellow flower heads (Figure 5-3c). Large infestations can be seen from hovering aircraft and helicopters, though identification may remain difficult as surrounding vegetation still determines the ease with which rush skeletonweed can be distinguished. Whichever monitoring method is deemed most appropriate for the targeted infestation and terrain, a 65 foot (20 m) buffer should always be searched in all directions from the outermost rush skeletonweed plants. Given the rhizomatous nature of this species, this buffer is required in order to ensure the entire population is inventoried and all daughter plants are contained within the surveyed area.

Figure 5-3. Rush skeletonweed: a. skeletal nature (Steve Dewey, Utah State University, bugwood.org); b. mapping an infestation with GPS (Joseph Milan, BLM); c. infestation in flower (Rachel Winston, MIA Consulting).

An increasing number of free smartphone and tablet apps help make accurate, detailed, and versatile weed mapping available to anyone (e.g., the apps available from EDDMapS, see page 62 for more information). Inventory efforts should document the following for each infestation: location coordinates, boundaries, estimated density (number of stems of target weed per area, e.g. square meter or square yard), land usage, treatment history, disturbance history (e.g., fire, flooding), habitat type (desert, upland, shrubland, grassland), and date. Photos of the infestation and a list of co-occurring species are also very useful. Documenting inventory and mapping efforts enables land managers to determine if all known rush skeletonweed infestations have been treated, and facilitates post-treatment monitoring. In turn, this allows land managers to judge the effectiveness of various treatment methods. See Chapter 4 for suggested techniques of monitoring infestations.

Prevention

Prevention activities focus on areas not currently infested by rush skeletonweed with the goal of keeping these areas weed-free. Though rush skeletonweed is already present throughout much of northwestern North America, there are many sites where it is absent or remains at low densities, and entire counties/states/provinces where rush skeletonweed has not yet invaded. Inventory and mapping efforts help identify the precise borders of these existing rush skeletonweed infestations as well as identify weed free areas. Preventing introduction and spread of rush skeletonweed to uninfested areas is more environmentally desirable and cost-effective than treating large-scale infestations.

Rush skeletonweed is spread by the movement of seed on wind, water, hay, motorized equipment, livestock, wildlife, or by root-fragments being deposited in uninfested areas. Preventing the spread of rush skeletonweed requires cooperation among all landowners and land managers. In areas where rush skeletonweed is not yet present, it is important to ensure that possible invasion avenues are identified and management actions taken to reduce the risk of spread. This includes minimizing soil disturbances and regularly monitoring uninfested sites to confirm that they have remained uninfested.

Cultivation, soil erosion (especially following flooding events and prescribed or wildfire), road grading, recreational activities (e.g., riding dirt bikes or four wheelers), and overgrazing all weaken existing plant communities, decrease plant cover, and cause disturbance, conditions that favor rush skeletonweed establishment and persistence (Figure 5-4). Because such activities are also potential ways of spreading rush skeletonweed seeds, they should either be avoided or closely monitored in skeletonweed-prone areas. Where grazing does occur, proper livestock management (such as strategic timing and stocking rates) will allow grazed vegetation to recover and competitive plants to increase which, in turn, will help prevent the establishment of rush skeletonweed. If possible, livestock should be kept off weed-infested land when they are most likely to spread viable seeds (e.g., after seed formation). If it is not possible to avoid driving vehicles and machinery (e.g., logging, construction, or rangeland fire-fighting equipment) through rush skeletonweed infestations, it is crucial that a thorough cleaning take place before equipment leaves the contaminated area.

Prevention and exclusion activities are typically paired with education efforts. Examples of exclusion efforts include weed-free forage programs, state and provincial seed laws, and mandatory equipment cleaning when leaving infested sites and before entering uninfested sites.

Figure 5-4. Overgrazing and erosion. (Paul Bolstad, University of Minnesota, bugwood.org)

EDRR

An early detection and rapid response (EDRR) program is a specific protocol for tracking and responding to new infestations. It relies heavily on education and outreach activities to be effective. An EDRR program targets areas where rush skeletonweed may spread. It consists of three complementary activities: 1) educating land managers and the public on weed identification and mapping techniques, 2) enlisting their aid in immediate and thorough detection of the weed, and 3) initiating rapid response eradication efforts at all verified locations of the weed.

The most cost-effective strategies for dealing with rush skeletonweed are prevention and EDRR. Because rush skeletonweed is particularly difficult to control once established, it is imperative that every effort be made to inventory regularly and immediately eliminate all early invaders.

Weed Control Activities

Biological Control

Biological control involves the use of living organisms, usually insects, mites, or pathogens, to control a weed infestation and regain the balance among coexisting plant species. Classical biological control focuses on the introduction of host specific natural enemies from the invasive weed's native range. This method of rush skeletonweed management is the most economical and suitable for larger infestations (tens to thousands of acres). For small patches (less than 4 acres or 1.6 hectares) of new satellite (those growing outside of well-established) rush skeletonweed infestations, more aggressive control methods should be utilized (e.g. physical control or herbicides). Refer to Chapter 3 for detailed descriptions of the biological control agents currently approved for use on rush skeletonweed and Chapter 4 for how to implement a rush skeletonweed biological control program in your area.

Physical Treatment

Physical treatment utilizes hand pulling, mowing, or tilling to remove or disrupt the growth of weeds and is the oldest method of weed control. Physical methods have had variable success in controlling rush skeletonweed but are labor-intensive and not suitable for the more rugged and inaccessible sites where skeletonweed has invaded. Due to rush skeletonweed's ability to regenerate from severed root fragments, extreme care must be taken to ensure physical control methods are carried out under the appropriate conditions so that rush skeletonweed populations do not increase as a result of control efforts. Regardless of the physical method employed, it is imperative that all equipment used be thoroughly cleaned following use to prevent the spread of rush skeletonweed seeds or propagating root fragments.

Hand pulling

Hand pulling is most appropriate in the EDRR stage of a rush skeletonweed infestation or on satellite populations occurring outside larger containment areas. Hand pulling can provide successful control of small rush skeletonweed infestations (under 1 acre or 0.4 ha) if applied persistently. It is especially effective on young plants; seedlings and rosettes growing for less than five weeks since germination are not capable of full regeneration from severed roots. As plants age, or for plants growing in compacted soils, hand pulling can result in increased rush skeletonweed populations due to regeneration from the severed roots of pulled plants. To account for this plant response, small populations of older rush skeletonweed individuals must be pulled several times a year and often for multiple years. Multiple hand-pulling sessions will also control new rush skeletonweed individuals sprouting later in the growing season from seeds lying dormant in the short-lived seed bank.

It is important to remove as much of the rush skeletonweed root as possible, while minimizing soil disturbance. When rush skeletonweed plants are in flower or seed, cut off and bag all flower stalks prior to pulling. Otherwise, the jarring action of pulling may dislodge and distribute seeds at the site. **All roots, stems, flowers and seeds should be securely bagged and taken to the trash or a transfer site to prevent possible rush skeletonweed vegetative growth or seed dispersal from pulled material.** Re-seeding the open space resulting from rush skeletonweed removal with seeds of desirable vegetation can provide competition to decrease rush skeletonweed seedling germination and persistence.

Because hand pulling removes the biocontrol agent's host from the site, this control method is not compatible with biological control. Hand pulling is most appropriate for small infestations where immediate eradication is feasible, while biological control is more appropriate for much larger, established infestations where the management goal is containment or asset based protection. One way to successfully combine these two methods is to release biological control agents in a large, main infestation while employing hand pulling to remove individual plants and to control small, satellite patches arising outside of the main rush skeletonweed infestation.

Mowing

Mowing rush skeletonweed infestations (Figure 5-5a) can sometimes exacerbate the problem by stimulating skeletonweed re-growth (and subsequent flowering) and reducing competition from surrounding vegetation. Regular mowing throughout the growing season utilizes much of the stored root reserves, and over time decreases the root regenerative capacity, rosette formation, and seed production. Frequent mowing of rush skeletonweed is not feasible in either the crop or rangeland setting where rush skeletonweed is so problematic in the western North America, but it may provide control to rush skeletonweed along roadsides and rights-of-way. Alternatively, mowing can be used to reduce nontarget plant cover and litter prior to fall herbicide applications, as this will improve coverage of the chemical on fall rush skeletonweed rosettes. When mowing is used as a form of rush skeletonweed control, it is important that mowing treatments occur before seed production because mowing can facilitate seed dispersal. This can be especially difficult to time properly in populations where plants flower at different times.

The destructive nature of mowing is damaging to the gall midge, *Cystiphora schmidti*, which utilizes stems and leaves for all stages of its life cycle. Mowing is more compatible with the root moth, *Bradyrrhoa gilveolella*, as larvae and pupae are mostly underground the majority of the year. Adult *B. gilveolella* can fly away during mowing events. Mowing may actually help distribute the gall mite (*Aceria chondrillae*) and the rust fungus (*Puccinia chondrillina*), and both biocontrol agents can re-establish on rush skeletonweed plants recovering from mowing efforts.

Tilling

One of the most common physical weed control methods in grain crops has been and continues to be cultivation prior to sowing of cereal seed. Because of rush skeletonweed's ability to regenerate from severed roots, cultivation as part of a crop and fallow system often historically had the opposite of the desired effect, actually leading to a dramatic increase of rush skeletonweed infestations in some regions. It has since been determined that cultivation can be used successfully for the control of rush skeletonweed, if the timing, frequency, and depth of cultivation are accurately applied.

Root fragment growth and development can only occur when there is sufficient moisture available in the soil and sufficient energy reserves in the root. Fragmentation occurring in dry soil often fails to grow new plants. The deeper the root or root fragment is within the soil, the greater the amount of energy reserves required to produce a shoot. If the root is cut again before the energy from shoot production is regained, its reserves are further depleted, and the plant is weakened and often killed. Consequently, deep (10 inches or 25 cm) and frequent cultivation of dry soil can help decrease rush skeletonweed populations (Figure 5-5b). In a wheat fallow rotation

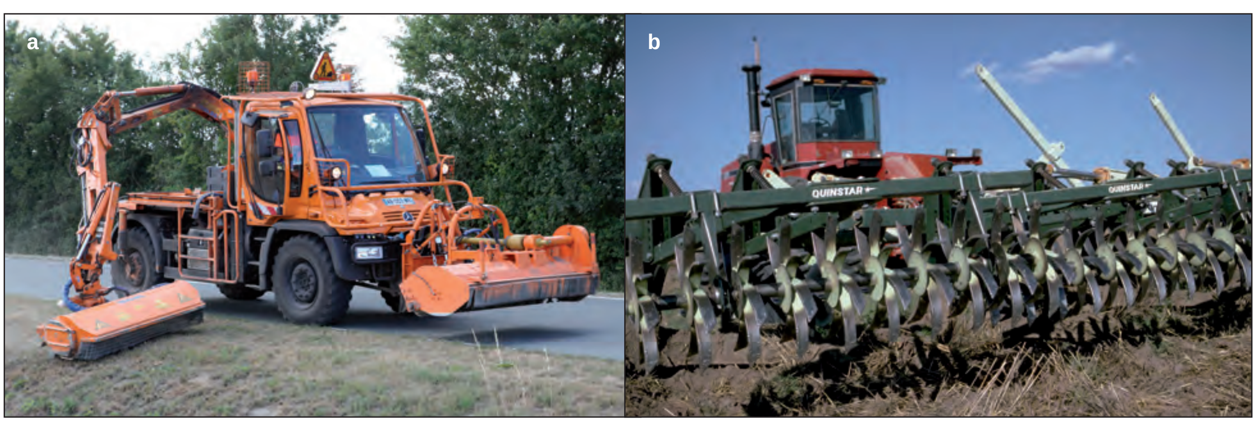


Figure 5-5. Physical weed treatments: a. roadside mowing (Joost J. Bakker, IJmuiden); b. tilling (Howard F. Schwartz, Colorado State University, bugwood.org).

or in the extensive rangeland system rush skeletonweed has invaded in western North America, frequent and deep cultivation are not feasible. Where cultivation is possible, be aware that shallow tilling in more moist soil may only increase the problem.

Because cultivation destroys aboveground growth and can slice rush skeletonweed roots into numerous fragments, this form of weed control is often destructive to all four species of rush skeletonweed biocontrol agents. Repeated tilling is most applicable in a crop setting, where chronic disturbance and the need to attain immediate control make biological control fundamentally incompatible.

Cultural Practices

Cultural methods of weed control (including burning, grazing, flooding, and seeding with competitive species) can enhance the growth of desired vegetation, which may slow the invasion of noxious weeds onto a site. Regardless of which method is used, all cultural control techniques are more successful when combined with other control methods, such as hand pulling prior to re-seeding or burning prior to applying herbicides.

Flooding and burning

For rush skeletonweed management, flooding is typically not applicable due to the arid locations rush skeletonweed frequently infests. Burning has largely been found to be ineffective. Observations of various researchers and land managers indicate that fire is more apt to increase infestations than hinder rush skeletonweed’s spread. While the aboveground biomass of rush skeletonweed burns readily in very hot fires, the deep rhizomatous root system is unlikely to be damaged and will recover post-fire. Furthermore, rush skeletonweed is capable of producing numerous windblown seeds whose establishment success is aided markedly by disturbance of the soil, such as following a fire. While fire often exacerbates the problem, it has been used intentionally in some locations to burn off plant litter in order to make the re-sprouting rush skeletonweed easier to see when applying herbicides (Figure 5-6a). When prescribed fire kills off competing vegetation, it will only increase the skeletonweed problem, even with subsequent herbicide applications. Revegetation with desired vegetation is recommended wherever fire is utilized to aid in rush skeletonweed chemical control.

High temperature fires destroy the galls and any individuals therein of the gall mite (*Aceria chondrillae*) and the gall midge (*Cystiphora schmidti*). Fire can also kill the urediniospores of *Puccinia chondrillina*. In fires where the roots of rush skeletonweed are not damaged, burning should not affect the larvae and pupae of *Bradyrrhoa gilveolella*.

Grazing

Most domestic livestock (including goats, sheep, horses, and cattle) and some species of wildlife will graze rush skeletonweed growing in the young rosette stage. Goats are the only species frequently observed feeding on the tougher stems of flowering plants. Sheep are believed to be the most effective against rush skeletonweed and have been shown to reduce or even prevent skeletonweed seed production (Figure 5-6b). The best results have been found with continuous grazing rather than rotational grazing. When livestock are moved as part of rotational grazing, rush skeletonweed quickly recovers, bolts, and spreads. Continuous grazing keeps the plant from bolting when other green feed is scarce; however, this heavy feeding is considered by many to be overgrazing.

Heavy grazing due to greater numbers of animals present is no more effective for controlling rush skeletonweed than is moderate grazing, because heavy grazing decreases the competitive ability of desired plant species. In the Intermountain West of the USA, some ranchers have found that while dense populations of rush skeletonweed often require intensive control efforts, scattered rush skeletonweed plants across well-managed pastures pose no serious decrease in livestock carrying capacity, and even supply late season forage when fall rosettes are available for livestock after grasses and other desirable species have senesced and died back for the season.

Although grazing can be effective for controlling rush skeletonweed under the right management circumstances, the difficulties and costs associated with proper livestock management often limit this control method. Where it is feasible to utilize livestock to manage rush skeletonweed, it’s important that the animals do not graze during seed set, as this can assist in the distribution of skeletonweed seeds.

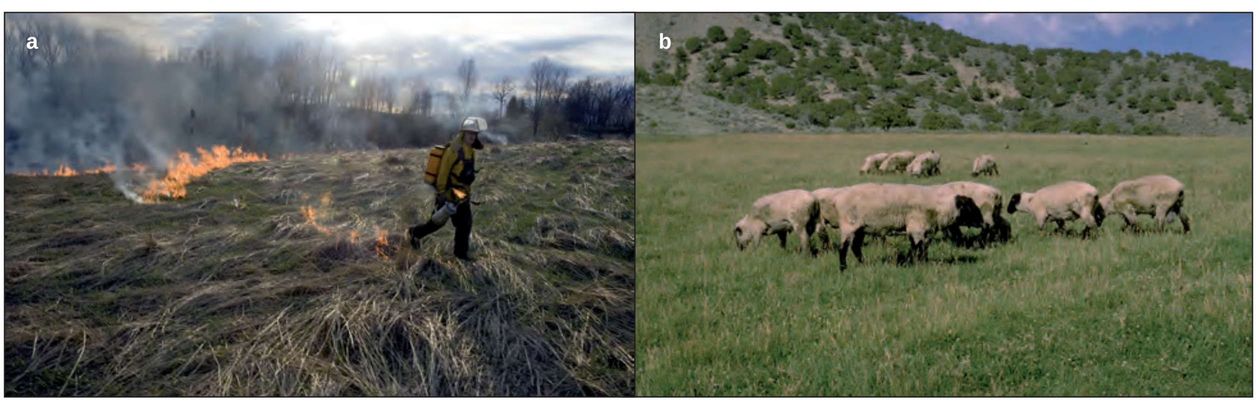


Figure 5-6. Rush skeletonweed management techniques: a. prescribed fire (David Cappaert, Michigan State University); b. grazing sheep (Howard F. Schwartz, Colorado State University); (a,b: bugwood.org).

The combination of grazing with biological control is largely unknown, though it can be assumed that grazing animals would wish to avoid plants heavily infested with the rust (*Puccinia chondrillina*) or the gall mite (*Aceria chondrillae*). Feeding on rush skeletonweed stems and leaves infested with the gall midge (*Cystiphora schmidti*) would destroy populations of that insect; however, grazing that did not disturb the soil or roots of rush skeletonweed should not hinder the life cycle of the root moth, *Bradyrrhoa gilveolella*.

Seeding competitive species

Where rush skeletonweed is established and then suppressed by one or more control methods, reinvasion by rush skeletonweed or other undesirable species is likely if the ecological niche they occupied remains unfilled. Successful long-term management requires the establishment and maintenance of desirable competitive species to avoid reinvasions.

Rush skeletonweed is very sensitive to competition for light and resources during early growth stages. In agricultural settings, when cool season annual crop or pastoral plants emerge before rush skeletonweed, their dense shade restricts the growth of skeletonweed seedlings and adult plant rosettes. This can reduce rush skeletonweed populations by as much as 63 percent in four years. In addition to competition for light, certain species hinder the growth of rush skeletonweed through other mechanisms. Deep-rooted perennials such as alfalfa (*Medicago sativa*, Figure 5-7a) compete with rush skeletonweed for much-needed soil moisture over the summer months. Alfalfa and other legumes such as sub-clover (*Trifolium subterraneum*, Figure 5-7b) also improve the soil nitrogen status by fixing their own nitrogen. This increases growth and competition from additional desirable pastoral species previously limited by nitrogen availability. Adding nitrogen and/or superphosphates artificially by up to 150 lb/acre (170kg/ha) has a

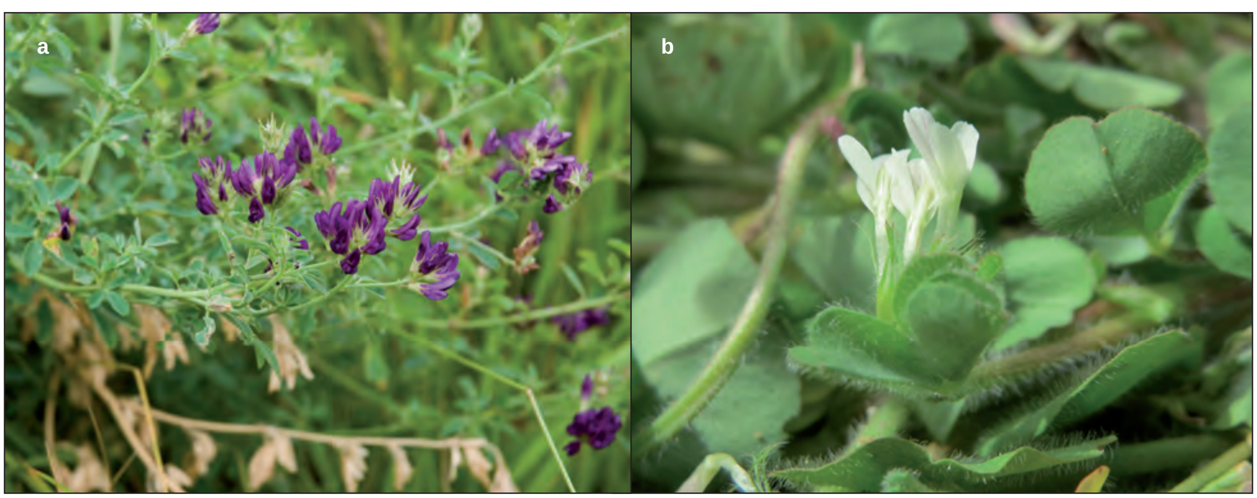


Figure 5-7. Legume species that compete well with rush skeletonweed in a pastoral setting: a. Alfalfa (*Medicago sativa*, Olivier Pichard); b. Sub-clover (AnRo0002).

similar effect, reducing rush skeletonweed rosette densities by an average of 80 percent. The level of management required to maintain dense stands of shading and/or nitrogen-fixing species is often difficult to achieve, especially in the vast natural and rangeland habitat rush skeletonweed has invaded in western North America.

In more natural settings, the most suitable plant species to use for competition with rush skeletonweed depends on habitat, site conditions, climate, management goals, and future land use. Ideally, planted seeds should contain a mix of species, some of which should be quick to germinate and others to provide more long-term competition to rush skeletonweed seedlings. Utilizing ecologically equivalent species (those with root and growth patterns similar to rush skeletonweed) may provide the best competition. Inventorying nearby sites that are uninvaded by rush skeletonweed may provide insight into the best replacement species. Consult your local county extension agent or Natural Resource Conservation Service (NRCS) representative for additional help in determining the best alternatives in your area. Further suggestions for ecoregions throughout the United States may be found on the Native Seed Network website (please see Chapter 5 References for the URL). Likewise, the “links” section of the USDA PLANTS website offers numerous revegetation guideline manuals specific to different regions of both the United States and Canada. This site also provides access to a program and fact sheets that utilize soil, plant, and climate data to select plant species that are site-specifically adapted, suitable for the selected practice, and appropriate for the goals and objectives of the revegetation project.

Control of rush skeletonweed prior to seeding more desirable species is important because established skeletonweed plants are highly competitive, and they spread rapidly and far via wind-carried seed. Seeding of competitors should take place immediately following exposure of soil to maximize their competitive abilities. For example, seeding should occur in bare soil following burning or after young skeletonweed plants have been hand pulled or killed with herbicides. Because high populations of rodents can reduce the success of re-seeding, erecting a raptor perch/pole may discourage rodent habitation and help ensure seeded species successfully germinate and establish.

Incorporating biocontrol agents with re-seeding can be difficult, primarily because the methods used to establish a productive stand of competitive species are not always compatible with the establishment and survival of biological control agents. Any method used to initially reduce rush skeletonweed stems and leaves to promote the growth of competitive species hinders the survival of all four biological control agents. Consequently, many successful revegetation programs establish competitive species first, using biological control agents after the seeded species have become established and rush skeletonweed begins to reappear. For example, rush skeletonweed plants growing in the presence of competitive leguminous species while simultaneously infected by the rust often have significant reductions in density and biomass. Alternatively, revegetation projects can target only a small portion of the infestation annually, leaving a reservoir of rush skeletonweed plants to support biocontrol agent populations. In some settings, it may be the biological control agents that open up the competing plant canopy, allowing for subsequent re-seeding to occur.

Chemical Control

Many herbicides are registered for use on rush skeletonweed growing in a variety of locations. Herbicide usage is most effective on small infestations, including newly established populations and recently established satellite patches arising from nearby older, larger rush skeletonweed infestations. If utilized appropriately, herbicides are also useful on the leading edge of large, advancing rush skeletonweed infestations.

Herbicides may be too costly to be of practical use in treating extensive infestations of rush skeletonweed and, similar to physical and cultural control methods, are also impractical in hard-to-access and environmentally sensitive areas. Repeated herbicide applications will be required over time as rush skeletonweed stems can re-sprout from their root system if not completely killed, and new rush skeletonweed plants can germinate from the seedbank. Potential nontarget damage to associated vegetation must also be considered when using herbicides. For these reasons, herbicides are best used as part of a larger, integrated pest management program that employs regular (annual) inventory and mapping, re-treatments when necessary, and incorporates other weed control methods in areas where herbicides are less likely to be cost effective or the most appropriate control method choice.

Herbicides are generally applied in one of two ways: spot or broadcast applications. Spot treatments are used for individual rush skeletonweed plants or small patches (Figure 5-8). In spot applications, an appropriate herbicide is applied to the foliage of target plants only, thus reducing nontarget effects. Broadcast treatments spray an appropriate herbicide over an entire area to treat larger weed infestations. Broadcast treatments should be used with caution as many herbicides may also impact plants that land managers may want to retain. If a broadcast treatment should kill all plants in a treated area, the bare soil remaining often allows rush skeletonweed to reinvade from the seedbank, creating a larger infestation than was there originally. Selective herbicides are those that target selected species (e.g., broad-leafed forbs vs. grasses) while leaving other species virtually unharmed. Utilizing selective herbicides in spot treatments helps reduce the nontarget impacts of herbicide applications, and is the recommended approach for treating rush skeletonweed infestations with chemical control. The herbicide label should always be referenced to help determine the chance of nontarget species damage.

Figure 5-8. Herbicide-spraying equipment for spot-treating small patches of rush skeletonweed in rangeland. (Leon Slichter, Idaho County Weed Control)

Most herbicides currently registered for the control of rush skeletonweed work best when applied while the weed is actively growing, especially when the weed is in the rosette stage in spring or fall. Extra care should be taken in making spring applications when rosettes of related species are present and may look similar to those of rush skeletonweed. Fall applications are generally more effective as herbicides are more readily translocated to roots during that season, and the stress of the subsequent winter increases plant mortality. Fall applications have the added advantage of competing vegetation having died back already, making rush skeletonweed individuals easier to see and treat. **The overall low number of leaves hinder the uptake of herbicides, so adding a surfactant to the herbicide mix is highly recommended regardless of the season of application.**

Some genotypes of rush skeletonweed are known to react to herbicides differently, though susceptibility studies for North American forms of rush skeletonweed are still needed. Some of the most widely used herbicides to combat rush skeletonweed in North America include:

Broadleaf selective herbicides

- **Aminopyralid** is one of the most frequently used and effective herbicides for rush skeletonweed. It can be applied in the spring or early summer on rosettes or bolting plants or in the fall on rosettes. It is effective on all parts of the plant. Aminopyralid can be very damaging to desirable forbs and trees, especially legumes. This herbicide can be applied near some tree species where dicamba and picloram cannot be used, but is not registered for use in forestry, and care should still be taken to verify whether a given species is tolerant of having aminopyralid applied with its dripline. Additional information regarding the known tolerance of various tree species to aminopyralid is available on the manufacturer's website. This herbicide does not kill grasses, sedges, cattails, or other monocots when applied post-emergence at broadcast label rates, **but it demonstrates some pre-emergence control of bromes dependent on application timing**. It has a long soil residual period, and broadcast applications may reduce re-growth from remaining skeletonweed roots or seedlings for 1-2 years following application. Aminopyralid is frequently combined with 2,4-D to increase translocation and efficacy.
- **Picloram** is also frequently used for the control of rush skeletonweed, often in a mix with 2,4-D. It should be applied to actively growing rush skeletonweed rosettes in spring or fall. It is effective on all parts of the plant. Picloram has a long soil residual period, which will reduce re-growth from remaining skeletonweed roots or seedlings for 2-3 years following application. **Picloram is mostly safe for use on grasses (young monocots may be affected, check the product label for additional information), but it will kill desirable legume species.** Picloram is less useful in hot, sunny conditions or in sandy soil because it is degraded by sunlight and can leach below the root zone in sandy soils.
- **Aminocyclopyrachlor + Chlorsulfuron**. Aminocyclopyrachlor is a relatively new broadleaf selective herbicide which is currently being packaged for sale with chlorsulfuron in uncultivated non-agricultural land, industrial sites, and natural areas. Aminocyclopyrachlor + chlorsulfuron should be applied to actively growing plants in the spring, and is effective on all parts of the plant, as well as having soil residual activity. **Low rates of aminocyclopyrachlor can kill nontarget tree and shrub species, so do not apply within underneath the dripline of trees or shrubs, to a distance equal to the height of the species of concern. Aminocyclopyrachlor may also injure a number of desirable grass species depending on the product rate.** It has the potential to be mobile in the soil, and may demonstrate residual activity several years after application.

- **Clopyralid** should be applied to rush skeletonweed rosettes in spring or fall. It is often mixed with other herbicides to increase weed control results. While it can provide control of all rush skeletonweed growth, it is less effective and has less soil residual than either aminopyralid or picloram, and may require additional monitoring and re-treatments compared to treatments that utilize those products. It is more selective than aminopyralid and picloram, affecting only four plant families, and does not kill grasses, sedges, cattails, or other monocots. **It will still kill desirable legume species and other forbs, so its use should be carefully considered in relation to existing native species, or those that may be reintroduced to a site when used in conjunction with broadleaf revegetation efforts.**
- **Dicamba** should be applied to actively growing rush skeletonweed rosettes in spring or fall. Dicamba alone is usually not the most effective herbicide for the control of rush skeletonweed because although it kills aboveground growth, plants re-sprout from the roots, and repeated applications are required. There is some residual activity of dicamba that is useful against the seedbank. Dicamba is often mixed with other herbicides (especially 2,4-D) to increase weed control results. When mixed with diflufenzopyr, dicamba is accumulated in the plant and is more effective on the root system. **Dicamba will likely kill desirable broadleaf species, including legumes.** Alone, it does not kill grasses, sedges, cattails or other monocots (though increased effects may be observed when it is used in combination with diflufenzopyr).
- **2,4-D** should be applied to rush skeletonweed rosettes in spring or fall. It is a broadleaf herbicide so will not harm grasses, sedges, cattails, or other monocots. 2,4-D is not the most effective herbicide for the control of rush skeletonweed as there is no soil activity, and though aboveground growth is killed, plants re-sprout from the roots and from the soil seedbank. Repeated applications are required. 2,4-D has a low cost, so is often combined with other herbicides that offer more complete and/or residual control, such as aminopyralid or picloram.

Non-selective herbicides

- **Imazapyr** can be applied anytime rush skeletonweed is actively growing. **It is a non-selective herbicide and should only be used in spot treatments and in situations where loss of nontarget vegetation is acceptable.** It is soil-active with a long residual activity, so is effective in preventing seedling germination. Imazapyr's soil residual activity varies with the rate applied, and may still provide weed control or harm new plantings anywhere from 3 months to 2 years post application. Even when used as a spot treatment, **imazapyr may harm other plants rooted in the general area or even downhill, depending on soil conditions and precipitation**.

- **Glyphosate** is typically applied in spring or early summer during the actively growing bud stage. It has had variable results for the control of rush skeletonweed. It has no residual activity in the soil, and repeated applications are often required. **It is a non-selective herbicide and should only be used in spot treatments and in situations where loss of nontarget vegetation is acceptable.** Glyphosate may temporarily result in bare ground. Glyphosate use should be accompanied by revegetation of desirable species.

When herbicides are used for the control of rush skeletonweed, it is important that the applicator adhere to all label instructions to ensure the usage, surfactant requirement, application rate, application timing and location/site of herbicide application fall within label recommendations. Not all herbicides are registered for use on rush skeletonweed in all settings (including on or near water), or for use in each state of the USA and in Canada. Some herbicides are restricted use and can only be applied by a certified and licensed applicator, and then only under specific conditions. Herbicide treatments can vary widely depending upon geographic location, climatic conditions and rate of application. Please consult your local weed control authority, county agricultural extension agent, or forest invasive coordinator to learn which herbicides work best for rush skeletonweed control and when to apply them in your area.

If land usage of treated areas includes grazing practices, consult the herbicide label for any grazing restrictions that might be applicable.

Heavy herbicide use will reduce the rush skeletonweed stems and leaves on which the gall mite, gall midge, and rust fungus rely, thus hindering establishment of these species. In order to guarantee that biological control agent populations remain viable as the rush skeletonweed infestations are reduced, plants should either be sprayed late in the growing season or some of the infested area should not be treated with herbicides to serve as "refuges" for biological control agents. The actions of herbicides and the root-feeding larvae of the rush skeletonweed root moth (*Bradyrrhoa gilveolella*) may be complementary in certain locations or habitats, though hard evidence is lacking.

The advantages and disadvantages of the most common rush skeletonweed control methods are summarized in Table 8.

Use Herbicides Safely!

Read the herbicide label, even if you have used the herbicide before. Follow all instructions on the label.

Wear protective clothing and safety devices as recommended on the label.

Bathe or shower after each herbicide application.

Be cautious when you apply herbicides. Know your legal responsibility as an herbicide applicator. You may be liable for injury or damage resulting from herbicide use.

Follow all storage and disposal instructions on the herbicide label.

Table 8. Comparison of rush skeletonweed management options

Control Method	Advantage	Disadvantage	Compatibility with Biocontrol
Biological Control	Sustainable – biocontrol agents generally do not have to be reintroduced once established	Measurable changes in weed densities may take many years (eradication is not the goal)	All four biocontrol agents are believed to be compatible with each other. Their impacts on each other are largely unknown.
	Most economical option for large infestations	Some risk of undesirable effects on nontarget plants	
	Public acceptance is generally higher than with other weed control methods	Permanent; cannot be undone	
	Selective	Not successful in all situations	
Physical Control (hand pulling)	Reduces seed production	Expensive and time intensive	Applicable only to very small infestations where biocontrol is not recommended. Hand pulling is not directly compatible with any biocontrol agent; however, biocontrol can be applied to large, main infestations while hand pulling can be used on surrounding small, satellite populations.
	Useful for small infestations that must be quickly eradicated	Must be repeated regularly due to constant threat of re-establishment from seedbank or plants re-growing from root fragments	
Physical Control (mowing)	Repeated mowing may reduce seed production; leaf removal reducing photosynthesis impedes root carbohydrate storage essential for plant persistence and vigor	May spread rush skeletonweed if done during flowering or seeding; or lead to compensatory growth if done infrequently	Not compatible with *Cystiphora schmidti*. Most likely *Bradyrrhoa gilveolella* stages may escape damage. Mowing likely aids in the dispersal of *Aceria chondrillae* and *Puccinia chondrillina*.
		Expensive and time intensive; requires proper timing and equipment	
Physical Control (tilling)	Kills rush skeletonweed plants and propagules when done frequently, at sufficient soil depth, and in dry soil	Only applicable in limited crop settings	Not compatible with all four biocontrol species. Tilling is only suitable in certain crop settings, where biocontrol is not recommended in general.
		May spread rush skeletonweed if done infrequently, at shallow soil depth, or in moist soil	
		Expensive and time intensive; requires proper timing and equipment	

(continued on next page)

Table 8 (continued). Comparison of rush skeletonweed management options

Control Method	**Advantage**	**Disadvantage**	**Compatibility with Biocontrol**
Cultural Control (flooding & burning)	Not recommended for rush skeletonweed management		
Cultural Control (grazing)	Allows use of the land even with heavy rush skeletonweed infestations	Cannot be used in many natural areas such as national parks and wilderness areas	Compatibility with biocontrol largely unknown. Livestock would likely wish to avoid skeletonweed infested with *Aceria* and *Puccinia*. Grazing *Cystiphora*-infested stems would destroy the gall midge. Grazing may be compatible with *Bradyrrhoa* if skeletonweed roots are not trampled/damaged.
		Nonselective; can exacerbate the problem	
	Can be used (under the right conditions) in combination with biological or chemical control methods	Can be expensive	
		Kills only above-ground growth; rush skeletonweed can recover rapidly post-grazing	
Cultural Control (re-seeding)	Can be used to restore native or more desirable species	Expensive for large areas	Compatible if biocontrol agents are introduced after competitive species are established. Also compatible if re-seeding is done only on small sections of the infestation annually, leaving rush skeletonweed “refuges” for the biocontrol agents. In some settings, it is biocontrol that may make re-seeding feasible.
	Can be self-perpetuating	May be ineffective if existing rush skeletonweed stand is dense	
Chemical Control	Fast acting	Expensive for large areas; repeat applications and monitoring often required	Herbicides are applicable only to small infestations, which are unsuitable for biocontrol. Compatible when using biocontrol on a main infestation and herbicides on surrounding small, satellite infestations. Somewhat compatible if herbicides are applied late in the growing season. Herbicides used in spring and summer interfere with the food source of *Aceria*, *Cystiphora*, and *Puccinia*. May be compatible with *Bradyrrhoa*, though this remains unknown.
	Successful for reducing rush skeletonweed densities in some settings, especially in combination with other control methods	May harm desirable vegetation	
	If applied correctly and repeatedly, has the potential to eradicate some populations of rush skeletonweed	Public resistance to chemical controls	
	Useful along transportation vectors (roads, trails, occasionally waterways)	Regulations or policies may prohibit use in some areas	

Glossary

abdomen	The last of the three insect body regions; usually containing the digestive and reproductive organs
achene	A small, one-seeded fruit that does not split at maturity
adventive	Species that arrived in the geographical area from elsewhere by any means, but is not self-sustaining and whose numbers are only increased through non-reproductive means, unlike a naturalized species
alternate	Where leaves appear singly at stem nodes, on alternate sides of the stem
annual	A plant that sprouts, flowers, and dies all in the same year
antenna (pl. antennae)	In arthropods, one of a pair of appendages on the head, normally many jointed and of sensory function
apomictic plant	A plant that produces seeds without fertilization
app	A self-contained program or piece of software designed to fulfill a particular purpose; an application, especially as downloaded by a user to a mobile device
basal	Located at the base of a plant or plant part
biennial	A plant that flowers and dies between its first and second years and does not flower in its first year
bioherbicide	A biologically based control agent for weeds, often using a fungus, bacteria, virus, or other pathogen.
biological control	The reduction in the abundance of a pest through intentional use of its natural enemies (predators, parasitoids, and pathogens)
bolting	Plant stage at which the flower stalk begins to grow
bract	A small, leaf-like structure below a flower
capitulum (capitula pl.)	Seed head of a plant in the sunflower family
caterpillar	The larval stage of a moth or butterfly
chrysalis	The pupal stage of a moth or butterfly
cocoon	A silk case that moth or butterfly larvae spin to contain the chrysalis

community	A naturally-occurring group of different species of organisms that live together and interact as a more or less self-contained ‘unit’
complete metamorphosis	A life cycle with four distinct stages (egg, larva, pupa, adult)
compound eyes	Paired eyes consisting of many facets, or ommatidia, in most adult Arthropoda
coordinates	A set of numbers used to specify a location
density	Number of individuals per unit area
diapause	Period of dormancy in insects
dicot	Plant with two seed leaves upon germination, including most common flowering species, excluding grasses, sedges, cattails, lilies and orchids
dissemination	Dispersal. Can be applied to seeds or insects
emergence (insect)	Act of adult insect leaving the pupal exoskeleton, or leaving winter or summer dormancy
enemy release hypothesis	Hypothesis stating that exotic plants can become invasive by experiencing less regulation (than native plants) by enemies in their introduced habitat. This relative release allows the exotic species to increase in abundance and distribution
eradicate	To get rid of something completely
erect	Grows upright and vertical as opposed to prostrate (spreading on the ground)
exoskeleton	Hard, external skeleton of the body of an insect
exotic	Originating in a distant foreign country; not native
field insectary	An area where host plants or animals are abundant and biological control agents are released and propagated with or without additional human manipulation
floret	One of the small, closely clustered flowers forming the head of a composite flower in the sunflower family
flower head	A special type of inflorescence consisting of numerous florets that actually look like one flower
forb	Herbaceous plant (does not have solid woody stems)
frass	Plant fragments, usually mixed with excrement, deposited by feeding insects
gall	A plant tumor; a localized proliferation of abnormal plant tissue that is induced by an insect, nematode, fungus or other organism and usually exhibits a characteristic shape and color; gall-making insects usually live and feed within the gall
genus (pl. genera)	A taxonomic category ranking below family and above species and consisting of a group of species exhibiting similar characteristics. The genus name is followed by a Latin adjective or epithet to form the name of a species

GPS	Global Positioning System; a space-based navigational system providing location and time information by using four or more satellites
head	Insect segment with the mouthparts, antennae, and eyes
herbivory	Feeding on plants
host	The plant or animal on which an organism feeds; the organism utilized by a parasitoid; a plant or animal susceptible to attack by a pathogen
host specificity	The highly-evolved, often obligatory association between an insect and its host (i.e. weed). A highly host-specific insect feeds only on its host and on no other species
inflorescence	The flowering part of a plant
instar	The phase of an insect’s nymphal or larval development between molts
invasive	Tending to spread prolifically and undesirably or harmfully
involucre	A circle of bracts under an inflorescence
larva (pl. larvae)	Immature insect stage between the egg and pupa (examples include grubs, caterpillars and maggots)
litter	Dead plant material, such as leaves, bark, needles, and twigs, that has fallen to the ground
margin (of leaf)	The edge of a leaf. Margins typically fall within a handful of categories and are useful in plant identification
membranous	Thin and transparent
molting	Process of insect development that involves shedding its exoskeleton and producing another for the next instar
monocot	Plant with only one seed leaf upon germination, including grasses, sedges, cattails, lilies, and orchids
monoculture	An area vegetated by a single plant species
NAD 83	North American Datum, the official datum used for the UTM geographic coordinate system in North America
native	Of indigenous origin
node	Part of the stem of a plant from which a leaf, branch, or root grows
nontarget effect	When control efforts affect a species other than the species they were enacted to control (can be positive or negative)
noxious weed	A weed whose control is mandated, and whose movement is regulated by federal or state law
nymph	Immature form of invertebrates that undergo gradual metamorphosis. Resembles adults
oviposit	To lay or deposit eggs
pappus	A tuft of hairs, scales, or bristles at the base of an achene in flowers of the sunflower family

perennial	A plant that lives for more than two years
petiole	Leaf stalk that attaches it to a plant stem
plant cover	The portion of the vegetative canopy in a fixed area attributable to an individual or a single plant species
pupa (pl. pupae; v. pupate)	Non-feeding, inactive insect stage between larva and adult
qualitative	Measurement of descriptive elements (e.g., age class, distribution)
quantitative	Measurement of quantity; the number or amount (e.g., seeds per capitula)
receptacle	Part of the stem to which the flower is attached
rhizome	A modified stem of a plant that grows horizontally underground, often sending out roots and shoots from its nodes
root crown	Part of a root system from which a stem arises; where a plant's stem meets the roots
rosette	A compact, circular, and normally basal cluster of leaves
senescence	Final stage in a plant's life cycle
seed head	Part of the stem to which the flower is attached
species	A fundamental category of taxonomic classification, ranking below a genus or subgenus and consisting of related organisms capable of interbreeding
surfactant	Often applied with an herbicide mix to help bring the herbicide into closer contact with the leaf surface in order to aid absorption
synchrony	Occurring at the same time (e.g. plant flowering and insect oviposition)
taxonomy	The classification of organisms in an ordered system that indicates natural relationships. The science, laws, or principles of classification; systematics
thorax	Body region of an insect behind the head and abdomen, bearing the legs and wings
toothed	Leaf margin that is regularly incised, such as for a saw
transect	A straight line of varying length along which plants are periodically sampled individually or in quadrants
trichome	Epidermal outgrowths on plants. There are various kinds, including numerous types of hairs and scales
UTM	Universal Transverse Mercator, a grid-based geographic coordinate system
vegetative reproduction	Reproduction in plants other than by seeds, such as from rhizomes, stolons, and from nodes on lateral, often creeping, roots
viability	The proportion of propagules (e.g., seeds) that are alive and can germinate
weed	A plant growing where it is not wanted
WGS 84	The World Geodetic System, a datum for latitude/longitude geographic coordinate systems

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Appendix I: Troubleshooting Guide: When Things Go Wrong

This guide is intended to assist those who encounter problems when establishing a biological control program. It identifies the probable cause of typical problems and offers solutions.

Problem	Probable Cause	Solution
Biological control agents unhealthy or dead when received	Physical damage to biocontrol agents in transport	Provide adequate packing material to minimize movement of containers and ice packs.
	Drowning	Do not put water in containers during transport; prevent accumulation of excess moisture; too much plant material causes condensation.
	Excess or prolonged heat or cold	Keep containers cool at all times; use coolers and ice packs; avoid exposure to direct sunlight while in transit.
	Starvation	Put rush skeletonweed foliage (no flowers, seeds, or roots) in containers.
	Release delay	Transport or ship biocontrol agents immediately after collection.
		Release biocontrol agents at new site immediately upon arrival or receipt of biocontrol agent.
	Parasitism and/or disease	Check source biocontrol agents. Ensure the insect population is disease-free when collecting or receiving shipment.
Reproductive problems	Biocontrol agents past reproductive stage	Collect at peak activity (i.e. insects are mating and ovipositing).
	Sex ratio: not enough males or females	Collect at peak activity; observe mating among target biocontrol agents before collecting; males often emerge earlier than females.
	Biocontrol agents not synchronized with the rush skeletonweed growth stage	Biological control agents require the weed to be at specific growth stage for optimal oviposition; collect biocontrol agents from sites with plants in similar stages.
Few biological control agents collected	Collection at wrong time	Refer to Table 6 for collection time and technique.
	Collection technique	Biological control agents can be killed/damaged during sweeping or aspirating so sweep lightly; avoid debris.
	Conditions at time of collection wrong	Refer to the Chapter 4 section “Collecting Rush Skeletonweed Biological Control Agents” for guidelines on desirable weather conditions.
	Population insufficient	Only collect from well-established populations.
Biocontrol agents not found after release	Site is unsuitable or too small	Refer to the Chapter 4 section “Selecting Biological Control Agent Release Sites.”
	Not enough biocontrol agents released	Release as many biocontrol agents as is feasible to ensure survival and reproduction.
	Pesticide use/mowing in area	Select sites where land usage does not interfere with biological control agent life cycles.
	Released on wrong species	Ensure rush skeletonweed is targeted, and the correct biocontrol agent is used.
	Released at wrong time	Release only during the correct plant stage and in the cool hours of the day. Refer to Table 6 for guidelines.
	Biocontrol agents not well adapted to conditions	Release field-collected biocontrol agents from local sources wherever possible rather than greenhouse-reared adults or insects collected from distant locations.
	Ants or other predators preyed upon biocontrol agents	Release only at sites with no obvious ant mounds or high insect predator populations (e.g. mice, voles).
Cannot locate release site	Location marker not obvious	Use bright-colored wooden, metal, or plastic stake.
	Site destroyed	Communicate with all direct and neighboring land users.
	Map poorly/incorrectly drawn	Check map; redraw with more detail or add landmarks; GPS.

Appendix II: Sample Biological Control Agent Release Form

Released By:______________________ **Release Date:**___/___/___ (mm dd yy) **State:** ______ **County:**________

Biocontrol Agent:________________________________ **# Released:**____________

Target Weed:________________________________ **Date Collected:**___/___/___ (mm dd yy)

Source of Biocontrol Agents:________________________________

Biocontrol Agent Life Stage (circle): Larvae Adults

Land Ownership (circle): Private County State USFS BLM COE BOR BIA/Tribe TNC Other (specify) ______________

Legal: T_____ R_____ Sec_____ Q_____ **Lat:** Deg____Min____Sec_____ **Long:** Deg___Min_____Sec____

UTM: UTM Datum Zone________ UTM Year________ UTM Easting: ____________ UTM Northing: ____________

ENVIRONMENT

Temperature (°F):________ **Wind:** Calm, Light, Moderate, Strong, Gusty **Wind Direction:** N S E W

Weather (circle): Clear Ptly Cloudy Cloudy Rain Snow **Release Time:**____________ AM/PM

Site Aspect (circle): N, NE, E, SE, S, SW, W, NW **Elevation:**______________________

Site Slope: Flat (0-10%) _______ Gentle (10-30%) _______ Moderate (30-60%) _______ Steep (>60%) _______

Topographic Position (circle): Valley Bottom Terrace Lower Slope Mid/Upper Slope Crest

Disturbance: (check all that apply, circle most prevalent) Cultivation ___ Fire ___ Flood ___ Grazing ___ Logging ____
Roads____ Mining ____ Recreation ____

SITE CHARACTERISTICS

Site Name:______________________ **Size of Infestation** (acres):______ **Estimated % Weed Cover:**__________________

Est. Weed Height (cm): _______ **Est. Weed Density** (# per meter sq.): _______ **Dominant Plant:**______________________

Distribution of Weed: Isolated_____ Scattered_____ Sc-Patchy_____ Patchy_____ Continuous_____ Linear_____

Phenology: Seedling % ____ Rosette % ____ Bolt % ____ Bud % ____ Flowering % ____ Seed % ____ Dormant % ____

Vegetation Type (check):		**Estimate % Cover:**	
Grassland	☐	Tree	____________
Pasture	☐	Shrub	____________
Dry Meadow	☐	Forb	____________
Moist Meadow	☐	Grass	____________
Shrubland Steppe	☐	Litter	____________
Conifer Forest	☐	Bare Ground	____________
Conifer Forest	☐	Rock	____________

Soil Texture: (check) Sand___ Silt___ Clay___ Gravel___ Loam___

Appendix II: Sample Biological Control Agent Release Form (Side 2)

CONTACT PERSON:
Name: ______________________
Address: ______________________
City: ______________________
State: ______________________
Phone: _____ - ______ - __________
e-mail: ______________________

LEGAL LANDOWNER:
Name: ______________________
Address: ______________________
City: ______________________
State: ______________________
Phone: _____ - ______ - __________
e-mail: ______________________

Road Map to Site

Site and Vegetation Map

Comments:

Appendix III: Standardized Impact Monitoring Protocol (SIMP) for *Bradyrrhoa gilveolella*

Overview:
A critical part of successful weed biological control programs is a monitoring process to measure populations of biological control agents and the impact that they are having on the target weed. Monitoring should be conducted on an annual basis for a number of years. The Idaho State Department of Agriculture, in conjunction with the University of Idaho, Nez Perce Biocontrol Center, and federal land management agencies, has developed the monitoring protocol below to enable land managers to take a more active role in monitoring the progress and weed control ability of the rush skeletonweed root moth, *Bradyrrhoa gilveolella* (BRGI) in efforts to control rush skeletonweed, *Chondrilla juncea*. This monitoring protocol was designed to be implemented by land managers in a timely manner while providing data which will enable researchers to better quantify the impact of BRGI on rush skeletonweed throughout the state.

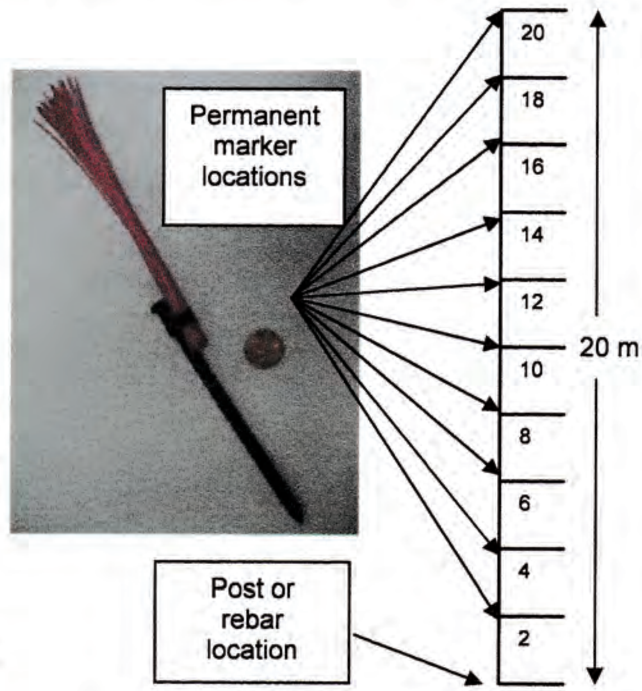
Rush Skeletonweed:
Rush skeletonweed is a long-lived perennial capable of reproducing by seed or vegetative regrowth. Flowers are self-fertile and contain 10 to 12 bright yellow florets. An individual plant is capable of producing up to 20,000 seeds that can remain viable in the soil for up to a year. Plants are typically 1.5 to 3 feet tall with multiple spreading, nearly leafless, light-green stems characterized by stiff, downward pointing hairs located on the lowermost 2 to 3 inches of the plant. Rush skeletonweed has a taproot that can reach depths of 7.5 ft into the soil, enabling it to thrive under a variety of climatic conditions. This weed is commonly found on sandy soils in areas of drought that have been disturbed by grazing, recreation, or fire.

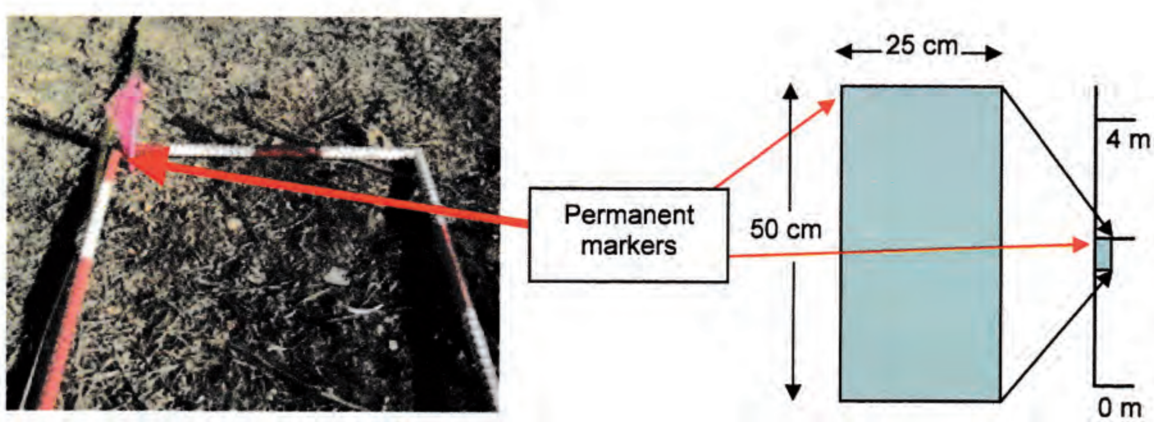
Rush Skeletonweed Root-boring Moth (BRGI):

BRGI is a recently approved biological control agent for rush skeletonweed. The moths overwinter as late instar larva or pupa and emerge from their shelter tubes, made of silk, latex, and frass, in May and June for winter generation adults or August to September for summer generation adults. Adults are 0.5 inches long, have a 1 inch wing span, are creamy buff in color, and have three distinct horizontal bands on their front wings. Females produce nearly 300 eggs and lay them on plant rosettes or in the soil. Six to 10 days later, the larvae hatch, penetrate the soil, and begin feeding externally on the roots while spinning their tubes. The larvae destroy the cortical and vascular tissues of the roots, depleting carbohydrate reserves, adversely impacting plant vigor and overwintering ability, and exposing the plants to soilborne plant pathogens. Larvae complete development in 45-60 days and the pupal period lasts seven to 10 days.

Monitoring:
The Statewide Biological Control monitoring protocol is based upon a permanent 20 meter vegetation sampling transect randomly placed in a suitable (at least 1 acre) infestation of rush skeletonweed and sweep net samples of BRGI. Annual vegetation sampling will allow researchers to characterize the plant community and the abundance and vigor of rush skeletonweed. Sweep net samples of BRGI adults will provide researchers with an estimate of BRGI population levels.

Permanent Site Set-up:
To set up the vegetation monitoring transect, you will need: 1) a 25 x 50 cm Daubenmire frame made from PVC (preferred) or rebar; 2) a 20 m tape measure for the transect and plant height; 3) 10 permanent markers (road whiskers and 16 penny nails – see picture below); 4) a post (stake or piece of rebar) to monument the site (see pictures for examples of field equipment); and 5) 30-45 minutes at the site during the **3rd week of June**. To set up the transect, place the 20 m tape randomly within the infestation. Mark the beginning of the transect with a post. Place permanent markers every 2 m (for a total of 10 markers) beginning at the 2 m mark and ending with the 20 m mark on the tape measure. Place the Daubenmire frame parallel to the tape on the 50 cm side with the permanent marker in the upper left corner starting at 2 m (see pictures). **Refer to the "sweep" data sheet for how to conduct monitoring.** Repeat the frame placement at 2 m intervals for a total of 10 measurements (one at each permanent marker).





Please see the following site for more information and downloadable forms:
http://www.agri.idaho.gov/AGRI/Categories/PlantsInsects/NoxiousWeeds/Bio_Control.php

Monitoring biological control agents is an essential component of a successful biological control program. Monitoring data can be used to accurately document the impact of this weed management practice. This monitoring form has been endorsed by the Nez Perce Biocontrol Center, University of Idaho, Forest Health Protection, Bureau of Land Management, and Idaho State Department of Agriculture. The monitoring information from this form will be used to document vegetation cover, target weed density, and biological control agent abundance. When conducted annually, this monitoring data will document changes that occur over time.

Standardized Impact Monitoring Protocol (SIMP) Biological Control Monitoring Form

General Information:

Observer(s):		Date:	Landowner:
Permanent site? Y N	Site name:		Weed:
Biological control agent:		Insect Stage:	
Lat/Long: N ° '	W ° '	UTM Datum:	UTM E:
		UTM Year:	UTM N:

Weed Infestation:

Size in acres:	Picture taken?	Yes No	If Y, picture direction:

Vegetation cover (all in %, rows add to 100%):

Frame	Target weed%	Other weed%	Forb/shrub%	Perennial Grass%	Bare ground%	Litter%	Moss%	Total%
1								
2								
3								
4								
5								
6								
7								
8								
9								
10								

Target weed size/density:

Frame	Number of Stems	Height of tallest stem (cm)
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		

Biological control agent:

10 sweeps repeated 6 times (for AP, GA, LA, CYAC & OBER) **OR** a 3 minute timed count repeated 6 times (for MEJA, ACMA galls & URCA galls)

Count site	Insect (or gall) count
1	
2	
3	
4	
5	
6	

Notes:

A step-by-step guide for completing the SIMP biological control monitoring form:

General Information:

- Observer(s) – Who are you?
- Date – Today's date.
- Landowner – Who is the landowner/land manager?
- Permanent? – Is this a permanent monitoring site?
- Site name – Which site are you monitoring? This could have a specific name if it is a permanent site.
- Weed – Which target weed are you are monitoring?
- Biological control agent – Which biological control agent you are monitoring?
- Insect Stage – What is the developmental stage of the agent are you monitoring (egg, larva, nymph, pupa adult)?
- Lat/Long OR UTM – What are the GPS coordinates of the site you are monitoring? If UTM (preferred), what datum and year is your coordinate system?

Annual grass – note stems which are typically solitary or in a few stemmed tufts

Vegetation Cover (all in %, rows add up to 100%) – All percentages are to be estimated to the nearest 5%. If there is a trace of any of the vegetation you monitoring in the frame, round up to 5%.

- Frame – Which frame number are you working on (1= 2m, 2= 4m, ...,10 = 20m)?
- Target weed % – What is % cover of the target weed to the nearest 5%?
- Other weeds % – What is the % cover of any other weeds in the frame to the nearest 5%? <u>Count undesirable annual grasses as weeds</u>
- Forb/Shrub % – What is the % cover of native forbs/shrubs in the frame to the nearest 5%?
- Grass % – What is the % cover of perennial grass to the nearest 5%?
- Bare Ground/Litter % – What is the % cover of bare ground/litter to the nearest 5%?

Target Weed Size/Density

- Frame – Which frame number are you working on (1=2m, 2=4m,...,10=20m)?
- Number of stems – How many stems of the target weed are in the frame?
- Height of tallest stems (cm) – How tall is the tallest stem of the target weed in the frame (in cm)?

Perennial grass – note the multiple stem base with multiple year's growth

Biological Control Agent

- Count location – Identify 6 sites at least 5 paces away from the vegetation transect but within the same weed infestation.
- # of insects per 10 sweeps – How many insects are in your net after 10 sweeps of the surrounding vegetation? Take one step between each sweep. Repeat 5 more times (for a total of 6 sweep sites, 60 sweeps) moving at least 2 steps away from the last sweep location **<u>(for AP, CYAC, GA, LA, & OBER)</u>**.
- # of biological control insects or galls per 3 min. count – How many biological control agents or galls do you see in a 3 minute period? Carefully approach the plants and be sure to count insects one time only. Please repeat 5 times (for a total of 6) moving at least 4 paces away from the first count location **<u>(for, MEJA, ACMA galls & URCA galls)</u>**.

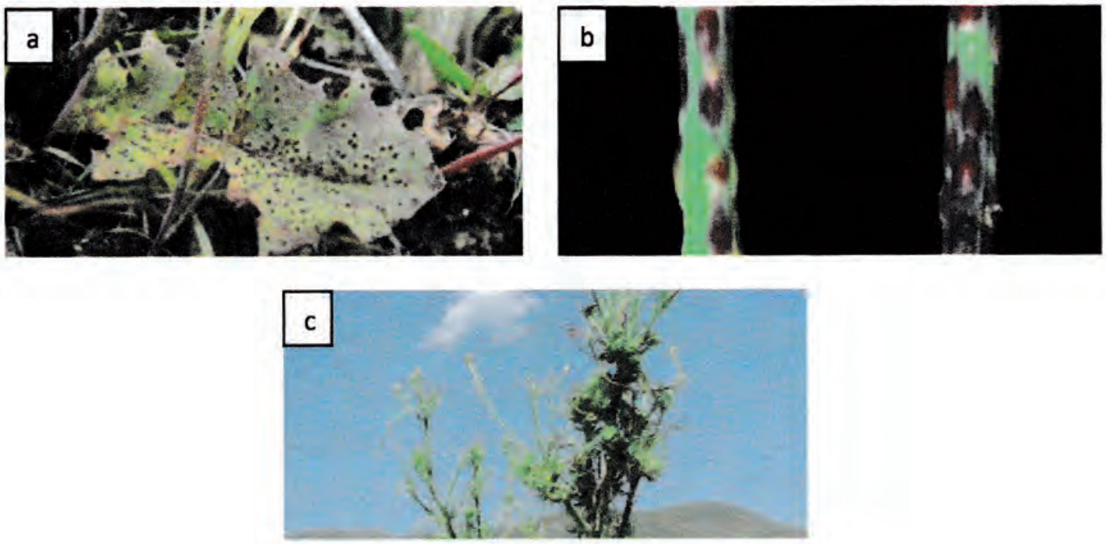
Appendix IV: Standardized Impact Monitoring Protocol (SIMP) for *Puccinia chondrillina* (the rust), *Cystiphora schmidti* (the midge), and *Aceria chondrillae* (the mite) on Rush Skeletonweed

Overview:

A critical part of successful weed biological control programs is a monitoring process to measure populations of biological control agents and the impact that they are having on the target weed. Monitoring should be conducted on an annual basis for a number of years. The Idaho State Department of Agriculture, in conjunction with the University of Idaho, Nez Perce Biocontrol Center, and federal land management agencies, has developed the quick monitoring protocol below to enable land managers to take a more active role in monitoring the progress and weed control ability of the three well-established biological control agents for rush skeletonweed: The rust (*Puccinia chondrillina*), the midge (*Cystiphora schmidti*), and the mite (*Aceria chondrillae*) in efforts to control rush skeletonweed, *Chondrilla juncea*. This monitoring protocol was designed to be implemented by land managers in a timely manner while providing data which will enable researchers to better quantify the presence/absence of typical biological control agents for rush skeletonweed throughout the state.

Rush Skeletonweed:

Rush skeletonweed is a long-lived perennial capable of reproducing by seed or vegetative regrowth. Flowers are self-fertile and contain 10 to 12 bright yellow florets. An individual plant is capable of producing up to 20,000 seeds that can remain viable in the soil for up to a year. Plants are typically 1.5 to 3 feet tall with multiple spreading, nearly leafless, light-green stems characterized by stiff, downward pointing hairs located on the lowermost 2 to 3 inches of the plant. Rush skeletonweed has a taproot that can reach depths of 7.5 ft into the soil, enabling it to thrive under a variety of climatic conditions. This weed is commonly found on sandy soils in areas of drought that have been disturbed by grazing, recreation, or fire.



Rush Skeletonweed Biological Control Agents:
Signs of rust presence can be found by locating pustules on rosettes or the stem (photo a above). Towards the end of the summer, the rust appears as black, leathery eruptions on the flower shoot typically near the base of the plant. Presence of the midge in the field can be indicated by galls formed on the stems and leaves of rush skeletonweed. Yellowish or maroon-colored galls are spherical and marginally raised on the leaf surface and stretched and more elevated on the stems (photo b above). To examine for the mite, look for galls which appear as clusters of tiny hyperplastic buds. The galls range in size from ½ - 2 inches in size and create a characteristic deformed appearance (photo c above).

Biological Control Agent Monitoring:
To conduct the monitoring, go into an infestation of rush skeletonweed and examine the rosettes, stems, and flower buds. This can occur from May (best time to find the rust – usually on rosettes) through July (when the midge and the mites are much more conspicuous on the stems and flower buds, respectively). For ten minutes, examine plants and look for the presence of the rust, the midge, and/or the mite. If these biological control agents are present, they will likely spread on their own. If they are not present, contact a biological control specialist to obtain these agents.

Appendix V: General Biological Control Agent Monitoring Form

SITE: ____________ **STATE:** ______ **COUNTY** ____________ **DATE:** ______ ______ ______
year month day

DATA COLLECTOR: ________________________ **TIME:** ____________
First and last name

LAT/LONG: N _____°_______' **W** ____°_______' **UTM DATUM:** __________ **UTM YEAR:** ________
UTM E: ____________ **UTM N:** ____________

ELEVATION: ___________ **TEMPERATURE:** ________ **WEATHER:**
____

INSECT COUNTS:

Chart A: Agent abundance	
1	1-10
2	11-25
3	26-100
4	100-500
5	>500

Species	Method	# insects (use Chart A)
Bradyrrhoa gilveolella	Randomly select 25 plants, dissect root tissue and count larvae	
Aceria chondrillae *Cystiphora schmidti* *Puccinia chondrillina*	In 5 one-minute intervals, count the number of plants you find infected with each species	

Rush Skeletonweed:

Chart B: Damage Class	
0	<1%
1	1-5%
2	6-25%
3	> 25%

Chart C: Cover Class	
0	<1%
1	1-5%
2	6-25%
3	26-50%
4	51-75%
5	76-95%
6	>95%

Quad #	% damage (use Chart B)*				Rush Skeletonweed	Plants					
	BG	AC	CS	PC	% cover (use Chart C)	# bolting plants	# flowering plants	Height 4 tallest plants (cm)			
1											
2											
3											
4											
5											
6											
7											
8											
9											
10											

*** BG=Bradyrrhoa gilveolella, AC=Aceria chondrillae, CS=Cystiphora schmidti, PC=Puccinia chondrillina**

Notes:

Instructions for Appendix V: General Biological Control Agent Monitoring Form

Materials needed: 20 meter tape measure (65 ft), 0.2 x 0.5 m (0.2 x 0.55 yard) quadrat frame, stopwatch, sweep net, monitoring form, pencils, clipboard, camera, and GPS unit to relocate transects.

General: The purpose of this monitoring activity is to estimate the abundance of rush skeletonweed and its biocontrol agents at the site, and to record measurements of a sample of rush skeletonweed plants. Conduct the monitoring when the biocontrol agents are at their peak. Monitoring is easier with two people, one to make the observations and the other to record data.

To set up the transect, place the 20-meter tape randomly within the infestation. Mark the beginning of the transect with a post or stake. Place permanent markers every 2 meters (for a total of 10 markers) beginning at the 2-meter mark and ending at the 20-meter mark. Place the quadrat frame parallel to the tape with the permanent marker in the upper left corner starting at 2-meters. Repeat the frame placement at each of the next 2-meter intervals for a total of 10 measurements (one at each permanent marker).

1. **Site information:** Fill out the site information at the top of the form.

2. **Biocontrol agent counting:** Use the chart for the method to count biocontrol agents. Carefully approach the site and avoid disturbing the vegetation. Adult moths (BG) often fly off once you touch stems (or even as you approach the quadrat). Use Chart A to record the category of abundance (1-5) for insects encountered (BG) or number of plants infected (AC, CS, PC).

3. **Locate the transect and position the quadrat:** After you have completed the biocontrol agent counts, locate the transect using the GPS coordinates and the permanent marker.

4. **Position the quadrat:** Position the quadrat along the transect, as close to the ground as possible, carefully positioning the quadrat along that transect line. Be sure not to damage the plants. The quadrat should be in the same location as the previous year’s quadrat. Move stems in or out of the frame area so that all stems originating inside the quadrat are included.

5. **Estimate feeding/infection damage:** Examine the rush skeletonweed plants for any damage to the leaves, shoots, flower heads, etc., such as malformed shoots due to mite galling. Standing over the frame, estimate the percent of damage over the entire quadrat, using Chart B to determine the category of damage.

6. **Estimate percent cover:** Standing over the frame, estimate how much of the quadrat is covered by rush skeletonweed. Use cover estimates in Chart C to estimate percent cover class.

7. **Count plants:** Count the number of rush skeletonweed plants, beginning at one corner of the quadrat and working systematically across the quadrat. Count the number of mature (floral) and immature (vegetative) plants.

8. **Measure plants:** Select the four (4) tallest rush skeletonweed plants in each quadrat (if there are fewer than 4 plants/quadrat, measure all that are present). Measure the stem height (to the closest cm).

9. **Other observations:** Record any general observations or useful information; disturbances, grazing, fire, etc., for the sample quadrat or the site in general.

Appendix VI: Rush Skeletonweed Quantitative Monitoring Form—Associated Vegetation

SITE: ____________ **STATE:** ______ **COUNTY** ____________ **DATE:** ______ ______ ______
year *month* *day*

DATA COLLECTOR: ______________________ **TIME:** ____________
First and last name

LAT/LONG: N ____°______' **W** ____°______'

UTM DATUM: ________ **UTM YEAR:** ______
UTM E: ________ **UTM N:** ________

ELEVATION: ________ **TEMPERATURE:** ______ **WEATHER:** ____________

Chart A: Cover Class	
0	<1%
1	1-5%
2	6-25%
3	26-50%
4	51-75%
5	76-95%
6	>95%

Percent of Quadrat (Use Chart A; total for each column is 100%)

	Q1	Q2	Q3	Q4	Q5	Q6	Q7	Q8	Q	Q10
Vegetated										
Soil, litter										
Rock										

Vegetation Cover (Use Chart A; total for column may exceed 100% due to overlapping of vegetation)

	Q1	Q2	Q3	Q4	Q5	Q6	Q7	Q8	Q	Q10
Rush skeletonweed										
All other vegetation:										
Forbs										
Grasses and Sedges										
Woody plants										

Individual Species (names) (Check if present or use Chart A to indicate percent cover)

	Q1	Q2	Q3	Q4	Q5	Q6	Q7	Q8	Q	Q10

Instructions for Appendix VI: Rush Skeletonweed Quantitative Monitoring Form—Associated Vegetation

Materials needed: 1 meter stick, 1.0 m^2 quadrat frame, data sheets, pencils, clipboard, camera, and GPS unit to relocate quadrats.

General: The purpose of this activity is to estimate the abundance of other vegetation in the community, and to record measurements of rush skeletonweed plant attributes. Monitoring is easier with two people, one to make the observations and the other to record data.

1. **Site information:** Fill out the site information at the top of the form.

2. **Position the quadrat:** Position the quadrat frame as close to the ground as possible, carefully positioning the quadrat along that transect line. Be sure not to damage the vegetation. Each quadrat should be in the same location as the previous year's quadrat of that same number.

3. **Estimate amount of vegetation:** Standing over the frame, estimate how much of the quadrat is vegetated, and how much is not vegetated (bare ground, rock, etc). Use cover estimates in Chart A to estimate percent cover.

4. **Estimate percent cover of vegetation:** Standing over the frame, estimate how much of the quadrat is covered by rush skeletonweed, how much is covered by other forbs, grasses, or shrubs. Use cover estimates in Chart A to estimate percent cover. Because vegetation can naturally overlap, it is possible to have a combined total percent cover to exceed 100%.

5. **Estimate percent cover of individual species:** Standing over the frame, estimate how much of the quadrat is covered by individual species, other than rush skeletonweed. Use this section to track specific species, for example perennial grasses, native forbs, etc.

6. **Other observations:** Record any general observations or useful information, such as disturbances, grazing, fire, etc.