

Biology and Ecology of Sickleweed (*Falcaria vulgaris*)

in the Fort Pierre National Grassland of South Dakota

BY

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in the Fort Pierre National Grassland of South Dakota

This thesis is approved as a credible and independent investigation by a candidate for the Master of Science degree and is acceptable for meeting the thesis requirements for this degree. Acceptance of this thesis does not imply that the conclusions reached by the candidate are necessarily the conclusions of the major department.

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Abstract

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In the last two decades the exotic plant sickleweed (*Falcaria vulgaris* Bernh., Apiaceae) has invaded, and come to dominate, large areas of the Fort Pierre National Grassland (FPNG) in central South Dakota, USA. Currently sickleweed is estimated to infest over 3200 ha of FPNG. The purpose of this study was to examine several of the biological and ecological traits that may contribute to the invasiveness of sickleweed in FPNG. Specifically, I researched germination characteristics of sickleweed seed collected from FPNG, the effect of sickleweed on other grassland vegetation in FPNG, and vegetative reproduction and regeneration of sickleweed from roots.

Germination trials conducted at the SDSU Seed Testing Laboratory found that sickleweed seed had a high degree of germination (70-90%) in all tested conditions. Germination response was higher at cooler temperatures with no effect of light conditions or storage temperature (at $P \leq 0.05$ level). Germination was significantly lower (at $P \leq 0.05$ level) after 18 months of storage. In simulated field conditions with soil collected from FPNG, sickleweed had higher germination in bare soil taken from a patch of sickleweed than in vegetated, unclipped western wheatgrass soil (at $P \leq 0.05$ level).

Plot biomass data revealed that in dense sickleweed stands (>40 stems/m²), sickleweed (unclipped plots) reduced the biomass of all vegetation when compared to control (no sickleweed) plots (at $P \leq 0.05$ level). Total biomass harvested from plots with sickleweed clipped at soil surface for one year was not significantly different (at $P \leq 0.05$ level) than total biomass harvested from unclipped and control plots. Native grass biomass was significantly reduced (at $P \leq 0.05$ level) by the presence of sickleweed in both treatment plots when compared to control plots. Biomass of introduced grasses and forbs was not significantly affected by treatments.

To determine sickleweed root generative capacity, root segments, 4 cm in length, were cut from the proximal, middle, and distal portions of green house grown sickleweed taproots and buried under 5 cm of FPNG soil in a SDSU greenhouse. After 2 months shoot generation was high (68-82%) in all regions with no significant difference in response by portion of root ($P=0.383$), which suggests that sickleweed is capable of surviving taproot excision and generating new shoots from small portions of taproot. During the course of this experiment new shoot production from lateral roots was observed in greenhouse grown plants, a phenomenon not previously detected in the field. Lateral spread via rootstocks in a field setting was confirmed when sickleweed shoots began emerging in the second growing season up to 1 m away from potted sickleweed plants transplanted from FPNG into a local garden. The results of this study indicate that sickleweed at FPNG is a perennial plant with high germination capabilities, reproducing and spreading by sprouting from rootstocks, and negatively impacts the production of other vegetation, especially native grasses.

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Introduction

In the last two decades the exotic plant sickleweed (*Falcaria vulgaris* Bernh., Apiaceae) has invaded large areas of the Fort Pierre National Grassland (FPNG) in central South Dakota, USA. In 1992, South Dakota Game, Fish, and Parks Department botanist Dave Ode first collected sickleweed on FPNG (personal communication, C. A. Taylor Herbarium specimens). At that time, Ode described the plant as being restricted to the west half of the Alkali West allotment, an area of approximately 65 ha. The pasture was in long term rest (Kelly Fuoss, personal communication) and was described by Ode as being in good condition with high species diversity. By 2001, sickleweed had spread to the point where managers began efforts to control the weed due to concerns that the aggressive spread and dense growth of sickleweed was negatively impacting native grassland species. A prescribed fire was conducted during late August 2005 in an invaded area to evaluate the effect of fire on sickleweed. However, post fire observations indicated that fire did not reduce the abundance of sickleweed (Anthony DeToy, former FPNG District Ranger, personal communication). Herbicide treatments with Dupont Telar XP® were initiated in 2004 and have reduced the density, foliar cover, and frequency of the weed in treated areas (US Forest Service unpublished data). As early as 2005, the U. S. Forest Service estimated that 3200 ha were infested at FPNG (Carol Erickson, USFS, personal communication), with new patches continuing to be found. In 2006, sickleweed was also discovered on the Buffalo Gap National Grassland (BGNG), roughly 160 km west of FPNG, and is now estimated to infest more than 40 ha near Wall,

SD (Terri Harris, USFS, personal communication). In 2008, herbicide treatments were initiated on the BGNG population.

Basic biological and ecological information, which may provide insight on management alternatives for sickleweed, is lacking. Oftentimes, what is published about sickleweed conflicts with what is observed in the field. At FPNG sickleweed is strictly a perennial plant, however, the species is described as annual, biennial, and perennial in its native range (Tutin et al. 1968). Sickleweed was found to have 80-100% germination at temperatures ranging from 10-25°C (Kew Gardens Seed Information Data Base), yet controlled environment germination trials conducted on soil collected from heavily infested sites of FPNG produced no sickleweed seedlings (US Forest Service unpublished data). Sickleweed is generally described as having a straight tap root with no mention of lateral vegetative reproduction via the root system. However, some sources describe sickleweed as capable of spreading laterally by root sprouting (Larina 2008, South Dakota Dept. of Ag. 1967, Gress 1923). At FPNG small infestations generally appear as circular patches (Figure 1), a characteristic commonly found in exotic species that spread laterally via rootstocks, e.g., Canada thistle (*Cirsium arvense* (L.) Scop.), leafy spurge (*Euphorbia esula* L.), and smooth brome (*Bromus inermis* Leyss.).

Objectives

The goal of my study was to determine biological traits of sickleweed that may contribute to its establishment and spread at FPNG. I also examined how the presence of



Figure 1. A circular patch of sickleweed (center of photo, whitish-green in color) with several smaller, more distant patches in Mallard (Middle) allotment at Fort Pierre National Grassland.

sickleweed adversely impacted grassland vegetation at FPNG. These data can help guide Forest Service grassland managers in decisions regarding sickleweed control measures and how to manage new areas of sickleweed invasion outside of FPNG and BGNG.

These data will also be useful in future studies of sickleweed. My results will provide baseline information on sickleweed biology and ecology in northern mixed grass prairie. Hopefully, this will reduce time and money spent determining basic biological information about the plant when it invades new areas, and hasten research efforts regarding effective sickleweed control strategies.

My specific objectives were to:

- Determine germination characteristics of sickleweed.
- Assess sickleweed's impact on grassland vegetation.
- Determine if sickleweed is capable of generating new plants from root segments and via branch roots.

Literature Review

The terms 'exotic', 'invasive', and 'weed' in literature are often loosely defined, with many interpretations provided by many authors; and often the terms are used interchangeably within articles (Colautti and MacIsaac 2004). Attempts to standardize terminology (Colautti and MacIsaac 2004) have not resolved the conflicting usage. In reviewing the literature, most authors neglected to define the term or terms they used, compounding the issue.

The term ‘exotic’ is usually used for a species that have invaded an ecosystem in recent history, at least by evolutionary standards, and generally through the intentional or unintentional actions of humans (Pimentel et al. 2000, Agrawal and Kotanen 2003). The interpretation of ‘weed’ is generally based on a species’ impact on humans (Baker 1974, Pimentel et al. 2000, Sutherland 2004), which usually involves negative agricultural impacts.

‘Invasive’ is a term usually used to describe plant species that readily invade ecosystems to the detriment of native vegetation (Callaway and Aschehoug 2000, MacDougall and Turkington 2005; invasive used within titles although other terms were used within text). An invasive species has been defined by the National Invasive Species Council (2006) as “an alien species whose introduction does or is likely to cause economic or environmental harm or harm to human health.” They further qualify their definition by adding that for a species to be considered invasive it has to do more harm than good; consequently, a species, such as smooth brome (*Bromus inermis* Leyss.), which readily invades natural areas to the detriment of native species, is not considered invasive because of its forage value. I have avoided the use of the term ‘invasive’ in this paper because of this ‘official’ definition, since economics have no bearing on a species’ invasiveness, i.e., its ability to invade ecosystems. I have also avoided use of the term “weed” where possible because many native species that have no negative impact in natural settings are often labeled as weeds because they are well suited to disturbed habitats, such as agricultural fields. For the purpose of this review, I have used the term ‘exotic’ species in describing the bulk of my literature cited, as most of the species

described are indeed introduced from another continent, and this term does not misinterpret the cited author's work when they may have actually used different terminology.

Exotic plant species can negatively impact ecosystem processes such as primary succession (Vitousek 1990), nutrient cycling, acquisition, and utilization (Vitousek 1990; Ehrenfeld 2003; Zedler and Kercher 2004), trophic structure (Vitousek 1990; Levin et al. 2006), and natural disturbance frequency and intensity (Vitousek 1990; Brooks et al. 2004). Exotic plant species have also been found to contribute to the decline of threatened and endangered species (Wilcove et al. 1998) and to reduce habitat use by native ungulates (Trammell and Butler 1995).

The monetary cost associated with exotic plants in the United States is large. Pimentel et al. (2000, 2005) reported monetary losses caused by several exotic plants to crops, pastures, and lawns in the United State to be in excess of \$30 billion based on reported control costs and estimated losses of crop and forage production. Duncan et al. (2004) estimated the costs associated with 16 exotic plants in the western U.S. They estimated potential losses as high as \$16 billion for individual species based on economic losses, including job losses due to reduced farm and ranch income, and societal losses due to lost ecosystem functions and recreational opportunities. Despite the economic losses, control efforts have not kept pace with the spread of exotic plants. The US National Park Service estimated that \$79 million dollars would be needed for their exotic plant control efforts between 1996 and 2000, but that only \$6.5 million was made available (Randall 1996).

Much attention has been focused on exotic species biology and ecology due to their large scale impacts, and a number of ideas have been explored to explain their aggressiveness. For example, Elton (1958) hypothesized that areas with low species richness likely do not have all available resources being used by plants and, therefore, are more susceptible to invasion. This hypothesis has been debated (Levine and D'Antonio 1999), but widely supported in theory and practice (MacArthur and Levins 1967; Tilman 1997). Stohlgren et al. (1999, 2003), however, contradicted this paradigm at multiple spatial scales when they found that areas with high species richness also had high levels of exotic species invasion. Ortega and Pearson (2005) conducted research that supports both sides of the argument. In their study, they described weak competitors (exotics that generally do not attain high abundance, usually annual forbs) as generally similar to native annuals in responding to small scale disturbances and invading areas of high diversity without affecting native species composition. In contrast, strong invaders (those that often dominate invaded areas, usually perennial forbs) were negatively correlated with species diversity. Ortega and Pearson (2005) also found that *Centaurea maculosa*, a strong forb invader, had a large effect on native grass dominance, shifting the dominant functional group from grasses to forbs. Ortega and Pearson also mentioned that many exotic species studies are conducted post-invasion, without benefit of pre-invasion data, which may skew reports of changes in species diversity.

Davis et al. (2000) hypothesized that disturbances cause fluctuating resources in native plant communities, which reduces the uptake of resources by existing species due to reduction of cover or by directly causing mortality or debilitating the existing plants.

These resources are then available for exotic species, providing a window of opportunity for their establishment. This hypothesis could account for the findings of Hierro et al. (2006), who conducted experiments with *Centaurea solstitialis* in its native and introduced ranges. These researchers found that *C. solstitialis* responded better (greater establishment and taller growth) to disturbance in its introduced ranges. However, the study also suggested that soil biota had a strong influence on their results. Callaway and Aushehaug (2000) and Vivanco et al. (2004) studied the allelopathic interactions of *Centaurea* (*C. maculosa* and *C. diffusa*) with North American natives. They found root exudates of these *Centaurea* species gave the plants a competitive advantage in resource uptake. These findings may have been influential in the conceptualization of the novel weapons hypothesis (Callaway and Ridenour 2004). The hypothesis asserts that exotic plants potentially possess powerful biochemical weapons that ‘naïve’ native plants are not evolved to compete against, thus giving such exotic species a competitive edge that allows them to dominate communities.

MacDougall and Turkington (2005) asked the question “Are invasive species drivers or passengers of change in degraded ecosystems?” They established study sites in the oak savanna of British Columbia, and removed all above ground biomass and as much below ground biomass of *Poa pratensis* L. and *Dactylis glomerata* L. as possible for 3 years. However, the removal of these exotic dominants did not result in an increase of historically native dominants. This supported the idea that exotic species are passengers of degraded ecosystems. The authors concluded that competitive ability alone is not enough to explain the dominance of exotic species.

Another hypothesis proposed to explain why exotics often become invasive is the enemy release hypothesis (Colautti et al. 2004, Agrawal et al. 2005). This hypothesis states that when a plant invades a new location it has escaped its specialized consumers, which gives the invader a competitive advantage over natives that are impacted by their own specialized consumers. This hypothesis is the basis for classical biological control research and has led to the successful control of several exotic plant species by the intentional introduction of their specialized consumers (McFadyen 1998). While this hypothesis is well accepted, Agrawal et al. (2005) showed mixed results in testing the hypothesis. For example, Agrawal and Kotanen (2003) found that native herbivores can, in some cases, impact exotic species more than natives.

Certain plant characteristics, including seed attributes, growth form, armament, etc., are often cited to help explain the ability of exotic plants to invade and dominate an area. Sutherland (2004) compared weeds and non-weeds (both exotic and native) from several large databases to determine if certain traits were more common in weeds. While his results were mixed for many traits, some did stand out, including armament (specialized defenses such as spines, toxic sap), short generation time, and generalized pollination. Some traits, such as vegetative reproduction and wind pollination, were expected to be characteristic in weeds but were actually more characteristic of non-weeds in his study. Rejmanek and Richardson (1996) found that seed size, seed crop size, and juvenile time period could all help explain invasiveness based on successful and unsuccessful introductions of various *Pinus* species. Daehler (2003) reviewed the literature to determine if exotic plants had performance advantages (ability to grow

larger, spread faster, lower construction costs, etc.) over native species. He found that in specific conditions many exotics have an advantage, but only rarely did exotics have a performance advantage over a wide range of environmental conditions.

Sickleweed is an exotic plant species that has recently shown the ability to invade a northern Great Plains grassland. Sickleweed is the sole member of its genus in the family Apiaceae (Umbelliferae). Like many members of the family, it produces a compound umbel of small, white, 5-petaled flowers. Each flower produces two, one-seeded brown mericarps with tan longitudinal ribs. The plant can reach a height of 1 m, and during fall senescence, the stem naturally abscises at the nodes and the plant segments tumble in the wind to disperse the seeds. Sickleweed's leaves are serrated with 'sickle-shaped' leaflets that have toothed cartilaginous margins. The leaves are alternate with a basally sheathing petiole and can be simple (mainly the basal leaves) to mostly pinnate with 3-5 leaflets. Sickleweed has a solid stem, which is unusual for Apiaceae, and a deep, fleshy tap root reaching more than a half meter in depth and with dormant adventitious buds over most of its length.

Sickleweed is native to Eastern Europe and Western Asia where it is sometimes used as a food plant and there is some interest in potential medicinal and antifungal properties (Qasem and Abu-Blan 1996, Turan et al. 2003, Khazaei and Salehi 2006). In the U.S., sickleweed has been recorded in sixteen states (USDA PLANTS Database 2010). The few published findings of sickleweed (in Kansas and Louisiana) mention only that the species was found and make no reference to it being invasive (Gates 1938, Thomas & Raymond 1987).

Sickleweed was documented in South Dakota as early as 1961 (C. A. Taylor herbarium specimen collected June 6, 1961 in a grain field near Tyndall, Bon Homme County, submitted by an extension agent and determined by Wayne G. Wright. The plant was described as a spreading, rhizomatous weed.). In 1964, SDSU agronomist records reported it infesting “few acres in southeastern counties and one infestation in the Black Hills region” (Michael Moechnig, SDSU Extension Weeds Specialist, personal communication). Of these, an infestation in Clay County was selected for herbicide trials by the SDSU Cooperative Extension Office. Most herbicides tested against sickleweed were largely ineffective; however, Tordon® provided 90-99% control (Michael Moechnig, personal communication). Use of this herbicide in combination with conversion of the area from alfalfa to row crops reportedly eliminated the population (Michael Moechnig, personal communication). In the 1970s, there were additional reports of infestations in several southeastern counties (Michael Moechnig, personal communication). Two of these were roadside infestations reportedly treated with Tordon® and eradicated. Another is located near a greenhouse on the South Dakota State University campus where it has persisted since the 1970s despite frequent mowing.

The SDSU Weed Extension and Evaluation Demonstration (WEED) Project testing has shown that DuPont™ Telar® XP and Escort® XP herbicides are effective in controlling sickleweed. The recommendation from SDSU Extension Weed Specialists is to apply either of these herbicides at a rate of either 0.7 oz or 1 oz per acre in the spring for the most effective control (Moechnig et al. 2007). Similarly, US Forest Service monitoring data (unpublished) found that two successive yearly applications of Telar®

XP at a rate of 0.7 oz or 1 oz per acre significantly reduced the presence of sickleweed. Sickleweed plot frequency was reduced by 68% to 14%; foliar cover was reduced from 19% to 1%; and density was reduced from 645 stems/m² to 24 stems/m².

Both chemicals belong to the DuPont™ sulfonylurea family of herbicides. Telar XP® is chlorsulfuron (DuPont 2009) and Escort® XP is metsulfuron (DuPont 2007). Both herbicides inhibit acetolactate synthase (ALS), an enzyme needed for the production of the branched amino acids isoleucine, leucine, and valine (DuPont 2007, 2009). The herbicides are primarily absorbed through the leaves and accumulate in meristematic areas of the plant (DuPont 2007, 2009). One to 4 weeks following herbicide application, yellowing of plant parts occurs with full effects often not evident until the following spring (DuPont 2007, 2009). Recommended application rates of both herbicides are low (<4 oz of chemical per acre) for control of most target species (DuPont 2007, 2009). Other Apiaceae reportedly controlled by these herbicides are wild carrot (*Daucus carota*), poison hemlock (*Conium maculatum*), and wild parsnip (*Pastinaca sativa*) (DuPont 2007, 2009).

Literature on sickleweed is limited and sometimes conflicting. For instance, the life habit of sickleweed is apparently variable and often described differently in different regions. For example, Kaul et al. (2006) describe sickleweed as biennial or perennial in Nebraska. Shishkin et al. (1968) and Davis (1972) describe it as annual, biennial, or perennial in Europe and Turkey, respectively. But Clapham et al. (1957) describe sickleweed only as perennial in the British Isles.

Another unclear or variable aspect of sickleweed is whether or not it reproduces vegetatively by sprouting from underground rootstocks. Gress (1923) described a sickleweed infestation in Pennsylvania as spreading via lateral root growth. Similarly, the 1967 edition of South Dakota Weeds (South Dakota Dept. of Ag. 1967) describes sickleweed as reproducing from rootstocks and includes a drawing that depicts lateral roots branching off the main taproot. No other publications mention this, including later editions of South Dakota Weeds (South Dakota Dept. of Ag. 1972). At FPNG, new sickleweed infestations generally appear as small circular patches, appearing much like new infestations of the strongly rhizomatous smooth brome, and suggest lateral spread via the root system. Efforts to confirm this in the field are hampered by the depth to which sickleweed taproots penetrate and the inability to trace fine lateral roots in dense clay soil.

The Royal Botanical Gardens of Kew, England, Seed Information Database lists germination data for sickleweed (Royal Botanical Gardens, 2008). In their germination trials, seeds were placed in 1% agar medium and then exposed to 8 hours light and 16 hours darkness per day at constant temperatures of 10, 15, 20, and 25°C. The database indicates 100% germination at 15 and 25°C, and 85 and 83% germination at 10 and 20°C, respectively. No further details are given. The database also refers to a seed bank study (Thompson et al., 1997) in which sickleweed seeds are described as persisting for less than 1 year in the soil. B and T World Seeds, a French company that sells sickleweed seeds, reports that germination can be improved by ‘smoke’, a proprietary butenolide compound produced by heated plant material (Matthew Sleight, B and T World Seeds,

personal communication). In the fall of 2006, the US Forest Service Rocky Mountain Research Station collected 100, 1-kg soil samples from FPNG to assess sickleweed's representation in the seed bank of a heavily infested area of FPNG (Jack Butler, US Forest Service, personal communication). The soil samples were transported to the research laboratory in Rapid City, SD, cold stratified at 5°C for 120 days, and placed into a germination chamber with germination conditions of 16 hours light at 30°C and 8 hours dark at 10°C. A total of 55 species germinated from the soil samples, however, no sickleweed germinated.

Study Area

The land that constitutes the Fort Pierre National Grassland (Figure 2) was originally homesteaded in the late 1800's by ranchers (USDA NRCS 1980, 1987, 1999). By 1900, most of the landowners had changed from livestock production to farming, breaking native sod to plant crops such as wheat. Much of the land that was initially broken for agricultural production was marginal for farming. During the dust bowl era, many of these farmers went bankrupt because the marginal soil could not support crops during the extended drought of the 1930s (USDA NRCS 1980). Under 'New Deal' legislation the government bought back a total of 4.4 million ha of degraded land across the nation, of which almost 1.6 million ha would eventually become national grasslands (Moul 2006). On June 20, 1960, FPNG was one of 19 national grasslands designated to be managed by the US Forest Service. The Forest Service is mandated to manage the national grasslands for multiple uses including grazing, wildlife production, and

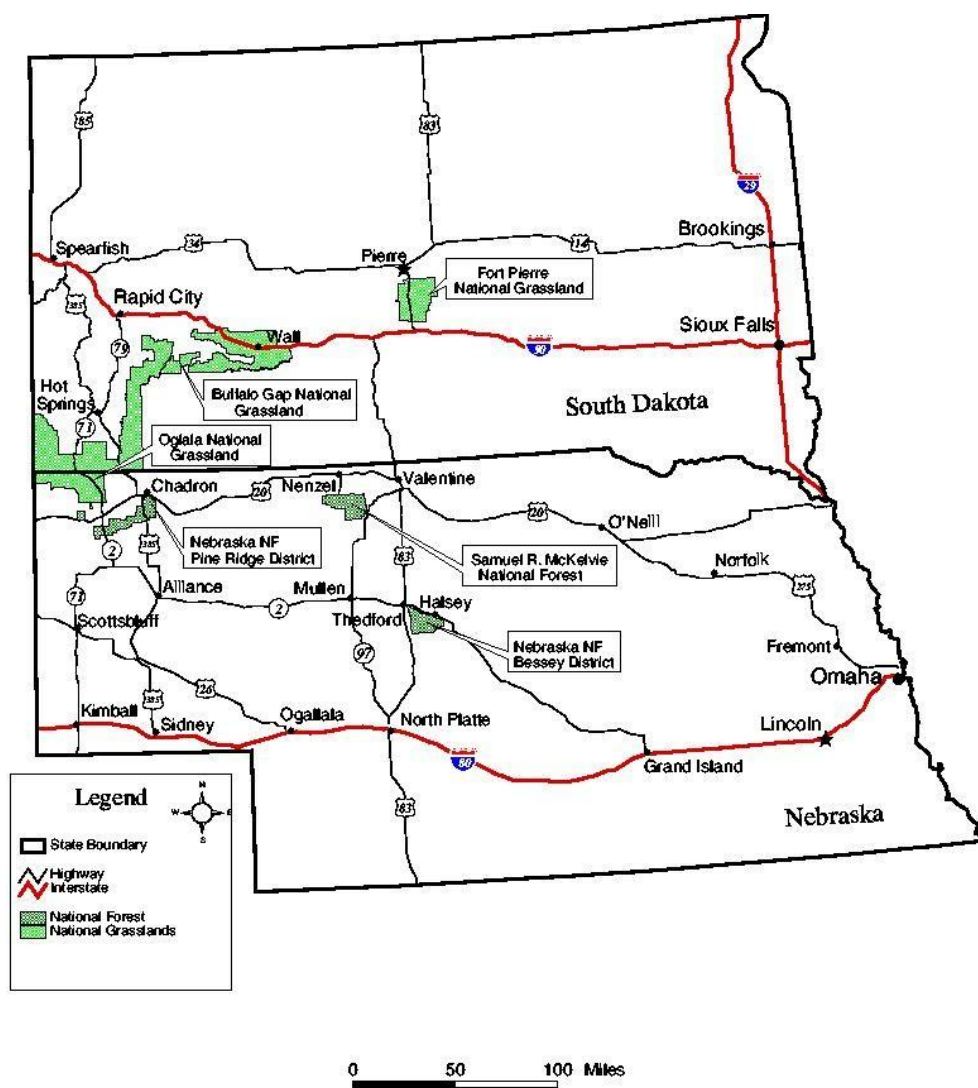


Figure 2. Locations of Fort Pierre National Grassland and other National Grasslands and Forests administered by the Nebraska National Forests and Grasslands, Chadron, NE district (Taken from US Forest Service Website, http://fs.usda.gov/Internet/FSE_MEDIA/fsm9_027827.jpg).

recreation (Moul 2006). FPNG is a 46,977 ha public grassland located in central South Dakota (approx. N44° 16' to N43° 58' and W100° 03' to W100° 28') about 30 km south of Pierre, South Dakota, and straddling Jones, Lyman, and Stanley counties.

Sixty-five million years ago the area that is now FPNG was part of a shallow inland sea. The shale derived clays that make up the soil were developed from the sediment that had been at the bottom of this sea. Weathered into fine clays by geological forces, the major soils of the grassland are Milboro and Opal-Sansarc associations of the Vertisol order, which are medium to well-drained with low permeability (Schumacher 1987, Faulkner 1999). The area is composed of gently rolling hills and mixed grass prairie ranging in elevation from 470 to 680 m. FPNG lies within the Bad River watershed, which drains into the Missouri River.

Average temperatures for the grassland are 22°C in summer and -7°C in winter. Average annual precipitation is 45 cm with most occurring between April and September (USDA NRCS 1987, 1999). The temperature range is extreme in the grassland as summer high temperatures regularly exceed 38°C and winter lows often fall below -29°C (SDSU Coop Weather 2009).

Vegetation of FPNG is characteristic of the wheatgrass-needlegrass prairie type (Johnson and Larson 2007). The dominant native grasses include western wheatgrass (*Elymus smithii* (Rydb.) Gould), green needlegrass (*Nassella viridula* (Trin.) Barkworth), needle-and-thread (*Hesperostipa comata* (Trin. & Rupr.) Barkworth), porcupine grass (*Hesperostipa spartea* (Trin.) Barkworth), buffalo grass (*Buchloe dactyloides* (Nutt) Englem.) and blue grama (*Bouteloua gracilis* (Willd. ex Kunth) Lag. ex Griffiths).

Vegetation on shallow hillsides is often more characteristic of the eastern tall grass or northern mixed grass prairie types (Johnson and Larson 2007) and is dominated by big bluestem (*Andropogon gerardii* Vitman), little bluestem (*Schizachyrium scoparium* (Michx.) Nash), and sideoats grama (*Bouteloua curtipendula* (Michx.) Torr.).

Introduced species comprise a major component of the grassland vegetation. Unfortunately, early records of what species were planted on the grassland when the land was retired from farming are unavailable or nonexistent. Introduced species such as crested wheat grass (*Agropyron cristatum* (L.) Gaertn.), smooth brome (*Bromus inermis* Leyss.), and alfalfa (*Medicago sativa* L.) were undoubtedly in the mix because of their ease of establishment and good forage potential. These species currently persist and have come to dominate many areas of the grassland and are continuing to invade native vegetation. Management objectives of the U. S. Forest Service include reducing the abundance of non-native plants and restoring more natural vegetation (Kelly Fuoss, personal communication).

Materials and Methods

Sickleweed Germination Characteristics

Germination experiments were conducted in the SDSU Seed Laboratory (seed lab) and focused on the effects of temperature, light, storage temperature, and age of seed on sickleweed germination. Additional germination trials were conducted in a SDSU greenhouse and focused on whether sickleweed germination is affected by soil type and competition with western wheatgrass.

Seeds were hand collected at FPNG on 20 September 2008 and 22-23 September 2009. Seeds were only collected from plants that had abscised from the crown or nearly so. Seeds were placed in brown paper bags and transported to the seed lab where they were separated from stems using a ribbed rubbing board while the remaining inert material was separated from seeds using graduated sieves. The seeds were stored in small manila envelopes. Half of the seeds by volume were stored dry at -15°C (freezer storage) while the other half were stored dry at 25°C (room temperature storage).

For my first germination experiment, seeds were placed in clear plastic seed boxes on wetted filter paper 6 months after initial (2008) seed collection. This experiment was conducted using a randomized complete block design with 4 replications of 50 seeds in each block. Blocks consisted of 5 temperature regimes, 2 light treatments, and 2 storage conditions within 5 germination chambers. All chambers had a 24-hour photoperiod with a light cycle of 8 hours light and 16 hours darkness, although half of the seed boxes were covered with aluminum foil to evaluate the influence of light on germination. Three of the chambers had constant temperatures of 10, 15, and 25°C. The remaining 2 chambers had alternating temperatures, one maintained 15°C during the dark cycle and 25°C during the light cycle, while the second chamber maintained 20°C during the dark cycle and 30°C during the light cycle. Germinated seeds were first counted and removed after 7 days and the remaining seeds were counted again after 30 days. The counts were summed for total germination per replicate. This experiment was repeated 12 months after seed collection with identical methods as part of experiments 2 and 3 as discussed below. The purpose of this experiment was to determine the effects of

temperature, light, and storage temperature on sickleweed germination. Data from this experiment was used in conjunction with later trials to assess the effect of age of seed on sickleweed germination (see experiment 3).

My second germination experiment was the inclusion of freshly collected seed (2009) as an additional treatment with the 12-month trial of my first germination experiment. The freshly collected seeds were subjected to the same treatments as the first experiment with the exception of storage conditions, since this test was run immediately after seed cleaning. The objective of this test was to 1) determine if an after-ripening period is necessary for germination, because the first experiment was run on seed aged 6 months, and 2) evaluate the influence of year of seed production on germination under the various conditions.

My third experiment was a germination trial conducted with the seeds collected in 2008 and aged 18 months in storage. This trial used the same methods as previous experiments except that it was conducted at only 2 temperature levels (constant 15°C and 25°C) with light. These germination conditions were chosen based on the results from the first germination experiment. Data from this trial with data from the first experiment were used to determine if sickleweed germination was reduced after ageing 18 months.

My fourth germination experiment was a greenhouse inter-seeding experiment to evaluate sickleweed germination responses to four soil treatments: 1) unclipped western wheatgrass sod, 2) clipped western wheatgrass sod, 3) bare soil from non-infested sites, and 4) bare soil from sickleweed infested sites. The purpose of this experiment was to determine if sickleweed germination was affected by competition with western

wheatgrass, and if soil from sickleweed patches produced higher sickleweed germination.

In August 2009, 20 clumps of western wheatgrass sod (plants and soil) were collected from FPNG in an area without sickleweed. The sod was placed in 20-cm diameter pots in the greenhouse. Pots were weeded of non-western wheatgrass species, watered regularly, and given Miraclegro® monthly, as per label instructions, for 3 months before the experiment began to allow plants to recover and develop a healthy and pure sod. Just prior to seed addition, 10 of the wheatgrass pots were clipped at 15 cm and maintained at that height for the duration of the experiment. This was done to simulate grazing to a height typical of FPNG grazed allotments (USDA USFS 2001). Soil without plants (bare soil) was also collected from the grassland in September 2009 for use in this experiment. Two batches of soil were collected, one from the same exact location as the wheatgrass sod collection (control) and the other from an isolated sickleweed patch within 100 m of where the control soil was taken (sickleweed soil). The soil was stored dry for 1 month prior to the experiment at which time the soil was placed in 20-cm diameter pots in the greenhouse. Twenty-five sickleweed seeds were sown into all pots at a depth of 1 cm and the location of each seed was marked with a toothpick. Pots were watered to field capacity biweekly and seedlings were counted at 1 week and again at 1 month as with other germination trials. Temperatures in the greenhouse ranged between 21 and 27°C during this experiment. After 1 month, germination counts were completed and seedling heights were also recorded as an index of plant vigor.

Germination data were subjected to binomial probability analysis using SAS v9.2 (SAS Institute 2008) and the PROC-LOGISTIC program. PROC-LOGISTIC was

selected for analyzing germination data since this test is intended for binomial data, such as whether or not a seed has germinated. Binomial responses violate the assumption of a normally distributed population as required by ANOVAs in programs such as PROC-GLM or PROC-ANOVA when used without transformed data. In PROC-LOGISTIC the significance of the main effects are determined by the Chi-squared goodness of fit value, which is also used to determine significance at the desired P value. In the case of this study if germination temperature, for example, produces germination levels significantly different than 50-50 odds (i.e. the flip of a coin) at $P \leq 0.05$. If the Chi-squared value is significant the odds ratio point estimate and confidence intervals are used to determine the significance of the individual treatments. The point estimate is the computed regression coefficient of the ratio of the odds of successes in one treatment over the odds of success in the other treatment. A point estimate of 1 indicates odds of success in the first treatment vs. odds of success in the second treatment is the same as 50-50 odds, <1 indicates lower odds of success in the first treatment, and >1 indicates higher odds of success in the second treatment. For example, a point estimate of 2 for temperature A vs. temperature B would indicate that temperature A has twice as likely a chance of producing a success than temperature B. The 95% confidence intervals are used to determine the treatment significance at $P \leq 0.05$ level. If the 95% confidence intervals do not overlap with 1, the estimate is significantly different than a 50-50 odds of success between the two treatments at $P \leq 0.05$.

In the third experiment (18 months) the data from the two temperature treatments were analyzed separately due to an age*storage temperature interaction in the 15°C

treatment. Height response to treatment effects in greenhouse trials were analyzed using standard ANOVA procedures and utilizing the PROC-GLM program. Post hoc analysis was run using Bonferroni t-tests on treatments with $P \leq 0.05$. The Bonferroni test, while having lower power, maintains an experimentalwise error rate of $P \leq 0.05$.

Effect of Sickleweed on Other Rangeland Vegetation

A primary concern of grassland managers is the effect sickleweed may have on forage production of native grasses and other rangeland vegetation. This experiment was conducted to assess sickleweed impacts on native vegetation in densely infested areas. Four permanent study sites were established on the grassland in July 2008. Two were located about 1 km apart in the Alkali (Home) Allotment with one in each of the Grass Creek (#1) and Reservation Road allotments (about 4.5 and 7 km, respectively, from the Alkali plots; Figure 3). At each site a 40-m treatment transect was placed in a dense patch of sickleweed (>40 stems/m²), and a 20-m control transect was located within 50 m of the treatment transect in similar vegetation but without sickleweed. Transects were oriented east-west and ends were marked with rebar stakes. Plots (0.5 m X 0.5 m) were placed at 1-m intervals along the north sides of the transects. Sickleweed was clipped out of every other plot along the treatment transect and from a 0.5-m buffer around the clipped plots. Clipping was done monthly as needed for the duration of this experiment. No clipping was done in or around plots within the control transect. All vegetation cover in the quadrats was recorded by species using cover classes as described by Daubenmire (1959). At approximately peak biomass in 2009 (25 June and 15 July depending on the

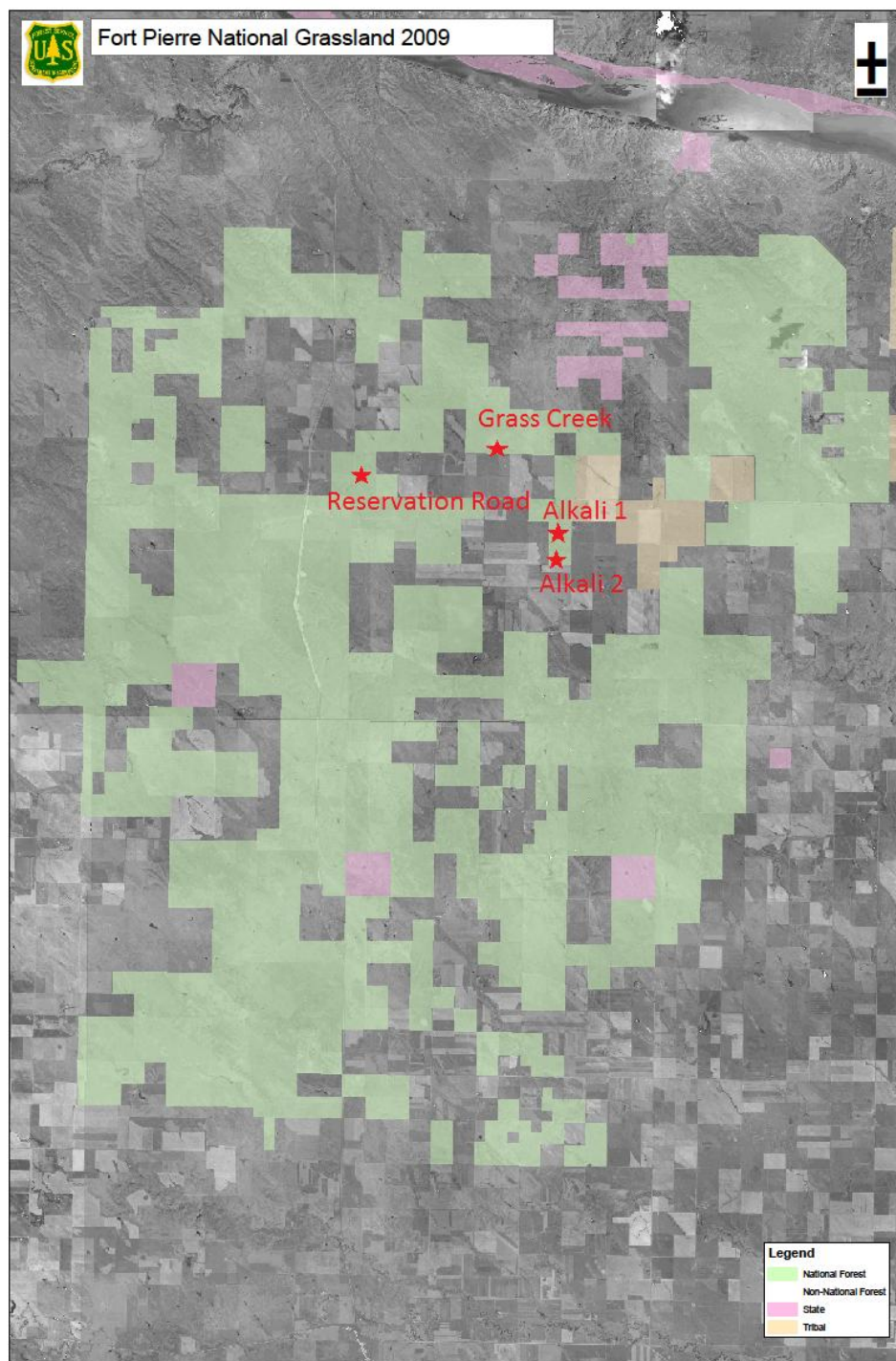


Figure 3. Map of FPNG showing locations of the four permanent study sites.

site), foliar cover and sickleweed density were again recorded and then all vegetation was clipped out by functional group (native grass, introduced grass, and forbs other than sickleweed). Functional group biomass measures were also summed for a total biomass other than sickleweed response. Biomass was dried at 70°C for 7 days and weighed to the nearest 0.1 g.

Biomass response to treatment effects, clipped sickleweed, unclipped sickleweed, and no sickleweed (control), were analyzed using analysis of variance procedures with SAS v9.2 (SAS Institute 2008) and the PROC-GLM program. Post hoc analysis was run using Bonferroni t-tests on treatments with $P \leq 0.05$. Species richness and diversity were calculated from plot cover data and analyzed using SAS v9.2 (SAS Institute 2008) utilizing the PROC-MIXED procedure. PROC-MIXED was selected because it allows for a combination of fixed and random effects in the model and it allows for adjusting correlation due to sampling the same set of plots during both sampling seasons. In this analysis the control, clipped, and unclipped treatments were the fixed effects and the sites were the random effects. Post hoc analysis utilized least square means test with the Tukey adjustment. The Tukey adjustment was used because it, like the Bonferroni test, also maintains a $P \leq 0.05$ experimentalwise error rate.

During the course of this research I also collected voucher specimens of vascular plants from throughout FPNG to better document the vascular flora of FPNG. The resulting species checklist comprises Appendix B. Nomenclature follows Kaul et al. (2006). Voucher specimens were deposited in the C.A. Taylor Herbarium at South Dakota State University.

Vegetative Reproduction and Regeneration from Roots

This experiment was conducted to evaluate re-growth potential of sickleweed roots in response to subsurface soil disturbance and injury. Anecdotal field observations suggested a correlation between populations of pocket gophers (*Geomys* and/or *Thomomys* spp.) and sickleweed infestations (Carol Erickson, personal communication). The tentative, casual explanation for the correlation involved gophers being attracted to areas of sickleweed infestation and feeding on the rootstocks, yet little if any sickleweed mortality was observed. Conversely these areas appeared to have higher sickleweed abundance suggesting that roots excised from below could survive and produce new vegetative growth from both the root above and below the excision. When coupled with the potential for small root fragments, which may have also been able to produce new shoots, to be transported within the burrows, this was considered a possible explanation for dense infestations in areas with high gopher abundance.

Plants for this experiment were grown from sickleweed seeds (collected from FPNG) planted in two v-shaped, Plexiglas, 50(l) x 20(w) x 40(d)-cm root boxes filled with potting soil. Plants were allowed to grow for 1 year prior to the start of this experiment. The root boxes were watered regularly and given Miracle Gro® monthly as per label instructions. Ultimately, this resulted in a large number of sickleweed plants with roots reaching the bottom of the root boxes. In preparation for this experiment, the root boxes were disassembled and soil was gently washed off the roots. The main taproots were then cut into 4 cm sections from three regions: proximal (1 cm below crown), middle (10 cm below crown), and distal (20 cm below crown). Only main

taproots large enough to provide all three sections were used for this experiment. The taproot sections were placed into 20-cm diameter pots by root region, with 6 pots per region, and 10 root sections per pot. The root sections were placed on top of 5 cm of soil taken from FPNG, and covered with an additional 5 cm of FPNG soil. Pots were watered weekly. After 2 months, the root segments were removed from the pots and classified as alive and with a shoot to soil surface, alive with a shoot not reaching the surface, or dead and showing no evidence of new growth.

Shoot regeneration by region response to cutting was subjected to the Kruskal-Wallis analysis of variance procedure (ANOVA) using SAS v9.2 software (SAS Institute 2008) and the PROC- NPAR1WAY program. Kruskal-Wallis tests were used to analyze these data due to the small sample size. While there were three response variables recorded, in the analysis the responses were reduced to alive or dead since it was evident that any of the living taproot segments producing shoots would have reached the surface if given more time.

Results and Discussion

Sickleweed Germination Characteristics

Sickleweed showed high levels of germination (Figure 4) under all tested laboratory conditions. Germination temperature significantly affected the germination of sickleweed (Table 1). In my first two seed lab experiments (6- and 12-month trials, 2008 and 2009 collected seeds), sickleweed germinated significantly better in the 10, 15 and 15/25°C treatments when compared to the 25°C treatment, the 20/30°C treatment was not

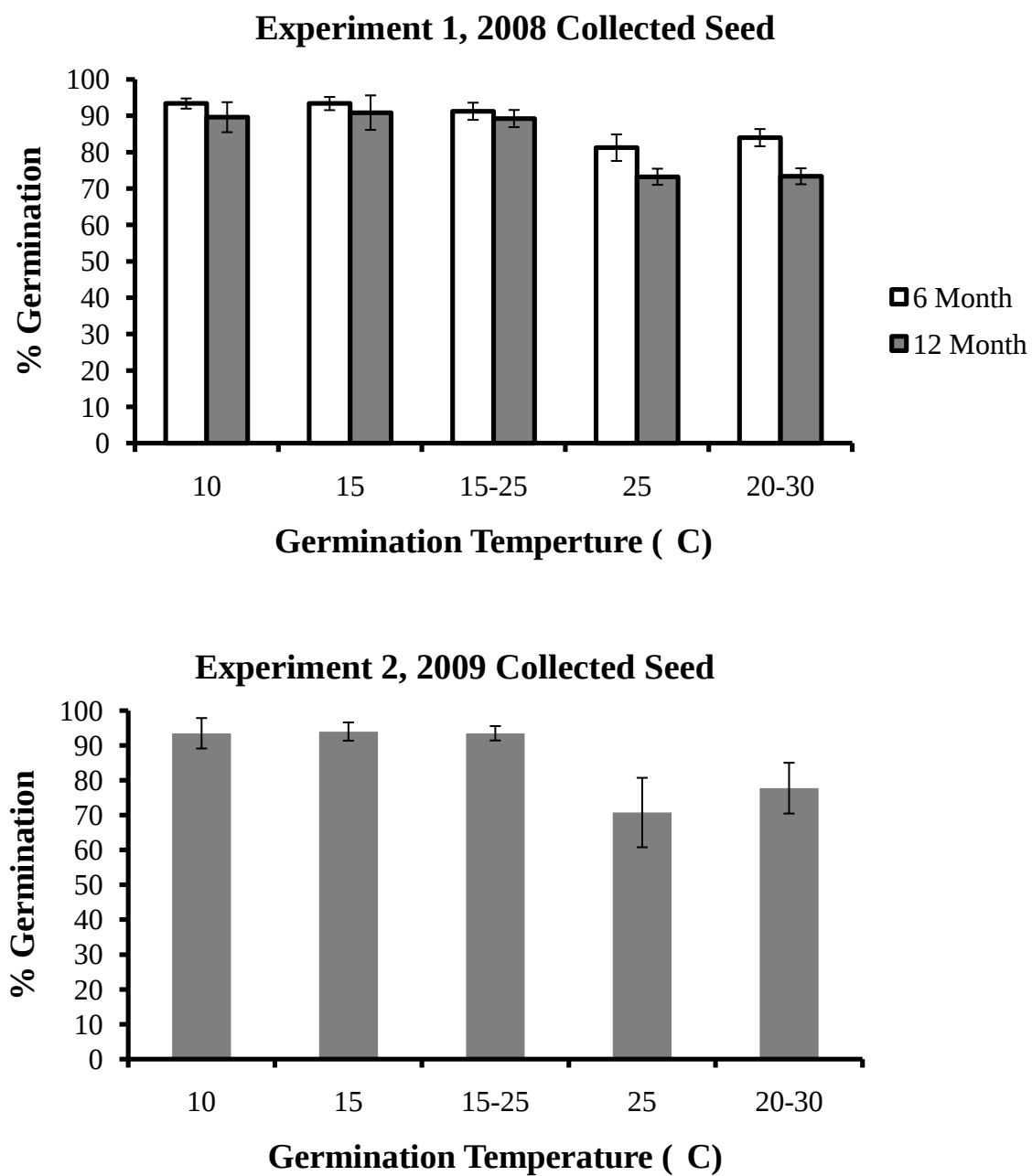


Figure 4. Mean germination (+/- standard deviation) by temperature for experiment 1 (2008 collected seed aged 6 and 12 months) and experiment 2 (freshly collected seed in 2009).

Table 1. Results of logistic analysis conducted on all sickleweed germination data from experiments 1 and 2. Temperatures in °C, ‘F’ indicates freezer storage, ‘RT’ indicates room temperature storage, and ‘N’ indicates 2009 collected seeds.

Source	Parameter Estimate	Standard Error	Wald Chi-square	P-value
Temperature 10 vs. 25	0.494	0.071	48.077	<0.001
Temperature 15 vs. 25	0.578	0.073	62.148	<0.001
Temperature 15/25 vs. 25	0.366	0.068	28.704	<0.001
Temperature 20/30 vs. 25	-0.646	0.053	147.724	<0.001
Storage F vs. RT	-0.072	0.042	2.884	0.090
Storage N vs. RT	0.141	0.053	7.218	0.007
Light vs. dark	-0.031	0.030	1.066	0.302

Source	Odds Ratio Estimate	Confidence Limits	
		Lower	Upper
Temperature 10 vs. 25	3.618	2.989	4.380
Temperature 15 vs. 25	3.934	3.234	4.785
Temperature 15/25 vs. 25	3.184	2.649	3.828
Temperature 20/30 vs. 25	1.157	0.998	1.343
Storage F vs. RT	0.998	0.877	1.136
Storage N vs. RT	1.234	1.044	1.459
Light vs. dark	0.941	0.838	1.056

significantly different than the 25°C treatment (Table 1). Light and storage temperature had no significant effect on germination of 2008 collected seed in these experiments. Similar germination temperature and light effects were found for seeds collected in both 2008 and 2009. The freshly collected 2009 seed had significantly lower germination than room temperature stored seed, however confidence intervals overlapped with 2008 collected seed in freezer versus room temperature storage treatments (Table 1), therefore this difference most likely resulted from the reduction in germination at warmer germination temperatures after ageing 12 months (see experiment three results).

The Kew Gardens Seed Information Database report on sickleweed germination differs from my results. Germination percentages reported by the database were similarly high (between 80 and 100%), but there was no clear trend in germination temperature difference in percent germination. Their data had high response at warmer temperatures (100% germination at 25°C) and low responses at cooler temperatures (85% germination at 10°C). However, comparisons are difficult to make since no information is given about sample size, age of seed, etc.

In my experiment with 18-month old seed, sickleweed germination was reduced compared to earlier trials (Table 2). Sickleweed germination was significantly reduced with age ($P=0.027$ at 15°C and $P<0.001$ at 25°C) but not storage temperature ($P=0.079$ for 15°C and $P=0.312$ for 25°C); even though room temperature storage resulted in lower germination in all but one case, 15°C during the 6-month trial. That particular case resulted in a significant age by storage temperature interaction ($P=0.027$) due to the 3.5% higher germination for room temperature stored seed aged 6-months, whereas the 12- and

Table 2. Results of the logistic regression analysis applied to the germination data for sickleweed seeds subjected to 2 temperatures (15°C and 25°C) and 2 seed storage conditions (F=Freezer, RT=Room Temperature). Confidence limits for Odds Ratio Estimates are given at the 95% probability level.

Treatment	Temperature Treatments	
	15° C	25° C
	Parameter Estimate (standard error, P)	Parameter Estimate (standard error, P)
Age of Seed	-0.047 (0.021, P < 0.027)	-0.064 (0.014, P < 0.001)
Storage of Seed	-0.518 (0.294, P = 0.079)	-0.047 (0.021, P < 0.312)
Age*Storage Temperature Interaction	0.047 (0.021, P = 0.027)	0.005 (0.014, P = 0.750)
Treatment	Odds Ratio Estimate (upper, lower confidence limits)	Odds Ratio Estimate (upper, lower confidence limits)
Storage F vs. RT at Age=6	0.626 (0.309, 1.268)	1.568 (0.984, 2.497)
Storage F vs. RT at Age=12	1.105 (0.731, 1.671)	1.658 (1.261, 2.179)
Storage F vs. RT at Age=18	1.950 (1.079, 3.523)	1.752 (1.177, 2.608)

18-month old room temperature stored seeds had lower germination (-2% and -6%, respectively). There was no consistent trend of higher germination in room temperature stored seed during the first or second germination experiments, so this was likely due to natural variation in seed germinability.

The results of the 18-month trial are consistent with the findings of Thompson et al. (1997, as cited by Kew Gardens Seed Information Database) who reported that sickleweed seed persisted for less than 1 year in the soil. In my experiments sickleweed seed lost as much as 30% viability after 18 months under room temperature storage conditions. The warmer germination temperature also seemed to lower germination of older seed, with room temperature stored seed producing 11% reduction in germination after 12 months in storage and the 16.5% reduction after 18 months in storage. Viability was somewhat preserved by freezer storage, even though there was not a significant difference, which is to be expected as this is a common practice when storing seed for long periods of time.

Sickleweed germination in the inter-seeding experiment was not as high as in the germination chambers (Table 3). The greenhouse experiment yielded germination between 54% and 24%. Treatments significantly affected sickleweed germination ($P=0.037$). Sickleweed seed germinated significantly better in the sickleweed soil treatment (54% germination) than the unclipped wheatgrass treatment (24% germination). The clipped (34% germination) and control (30% germination) treatments were not significantly different from the other treatments. This confirms that the degree

Table 3. Odds ratio estimates of logistic regression applied to germination response to soil and vegetation treatment. Germination was significantly affected by treatments ($P=0.037$).

Odds Ratio Estimates for Germination			
Treatment Effect	Point Estimate	95% Wald Confidence Limits	
		Lower	Upper
Clip vs. Unclipped wheatgrass	0.496	0.107	2.307
Control vs. Unclipped wheatgrass	0.822	0.179	3.775
Sickleweed Soil vs. Unclipped wheatgrass	0.104	0.020	0.545

of germination observed under ideal conditions in the seed lab would not be expected in the field. However, with up to several thousand seeds produced per plant (personal observation) and high sickleweed densities at FPNG, my lowest germination response of 23.6% would still produce many seedlings. While not statistically significant, differences in germination between clipped and unclipped wheatgrass treatments may coincide with Davis et al. (2000) where they suggest that herbivory reduces resource uptake by native plants (simulated through clipping), thus making more resources available and usable for invasive species to become established.

Sickleweed seedling heights (Table 4) were significantly affected by treatments ($P < 0.001$). Seedling heights were tallest in the control treatments (5.2 cm) and shortest in the unclipped wheatgrass treatments (3.4 cm). The sickleweed soil (4.8 cm) and clipped wheatgrass (4.7 cm) were not significantly different from each other but were significantly different than control and unclipped wheatgrass treatments.

It is not surprising that seedling height was the shortest in competition with unclipped western wheatgrass as this treatment would be expected to have the most competition for resources such as light, water, and nutrients. Conversely, the control treatment lacked inter-specific competition for resources and would be expected to have the tallest seedlings. While not significantly different, the treatment with the highest germination (sickleweed soil) did not have the tallest seedlings; which may indicate that intra-specific competition is an important factor in sickleweed seedling competition.

My germination results follow what other research has generally reported for Apiaceae. Germination is often quite high in laboratory settings (Baskin and Baskin

Table 4. Mean sickleweed seedling height in response to soil treatment and competition with western wheatgrass. Means followed by different letters indicate significant differences in germination and height at the $P \leq 0.05$ level.

Treatment	Average Sickleweed	
	Seedling Height (cm)	Standard Deviation
Control	5.2 ^A	1.2
Sickleweed Soil	4.8 ^B	1.4
Clipped Wheatgrass	4.7 ^B	0.8
Unclipped Wheatgrass	3.4 ^C	1.2

1979, Hendrix 1984) but low in the field (Thompson and Baster 1992). Thompson and Baster found Apiaceae establishment in tall unmanaged grass to be low (2-39% of sown seeds), and most of the ten species that did germinate failed to establish. What is most interesting as it relates to this study is that based on their results, Thompson and Baster (1992) produced a short list of species that they predicted most likely to establish in unmanaged grass. Their list included sickleweed and several other umbellifers that are polycarpic and found in open dry habitats.

Effect of Sickleweed on Rangeland Vegetation

The biomass of rangeland vegetation was negatively affected by the presence of sickleweed (Figure 5). The total non-sickleweed biomass of all rangeland vegetation was significantly affected by treatments ($P=0.019$). Non-sickleweed biomass in plots where sickleweed was not clipped was 23% lower than in control plots. The biomass of plots in which sickleweed was clipped was lower than in the controls, but not significantly different than the other two treatments. These results suggest that sickleweed's effect on total biomass may be based on competition for limited resources such as water, light, or nutrients. This is consistent with what Myras and Juntilla (1981) found with giant hogweed (*Heracleum mantegazzianum*, Apiaceae) and its interaction with other vegetation in Norway. They found that competition for light caused the major loss in biomass of other vegetation and that competition for nitrogen was higher in dense stands. Total biomass was then separated into native grass, introduced grass, and forbs and re-analyzed. Native grass was significantly affected by treatments ($P=0.012$) but introduced grasses and forbs were not ($P=0.898$ and $P=0.237$ respectively, data not shown). Average

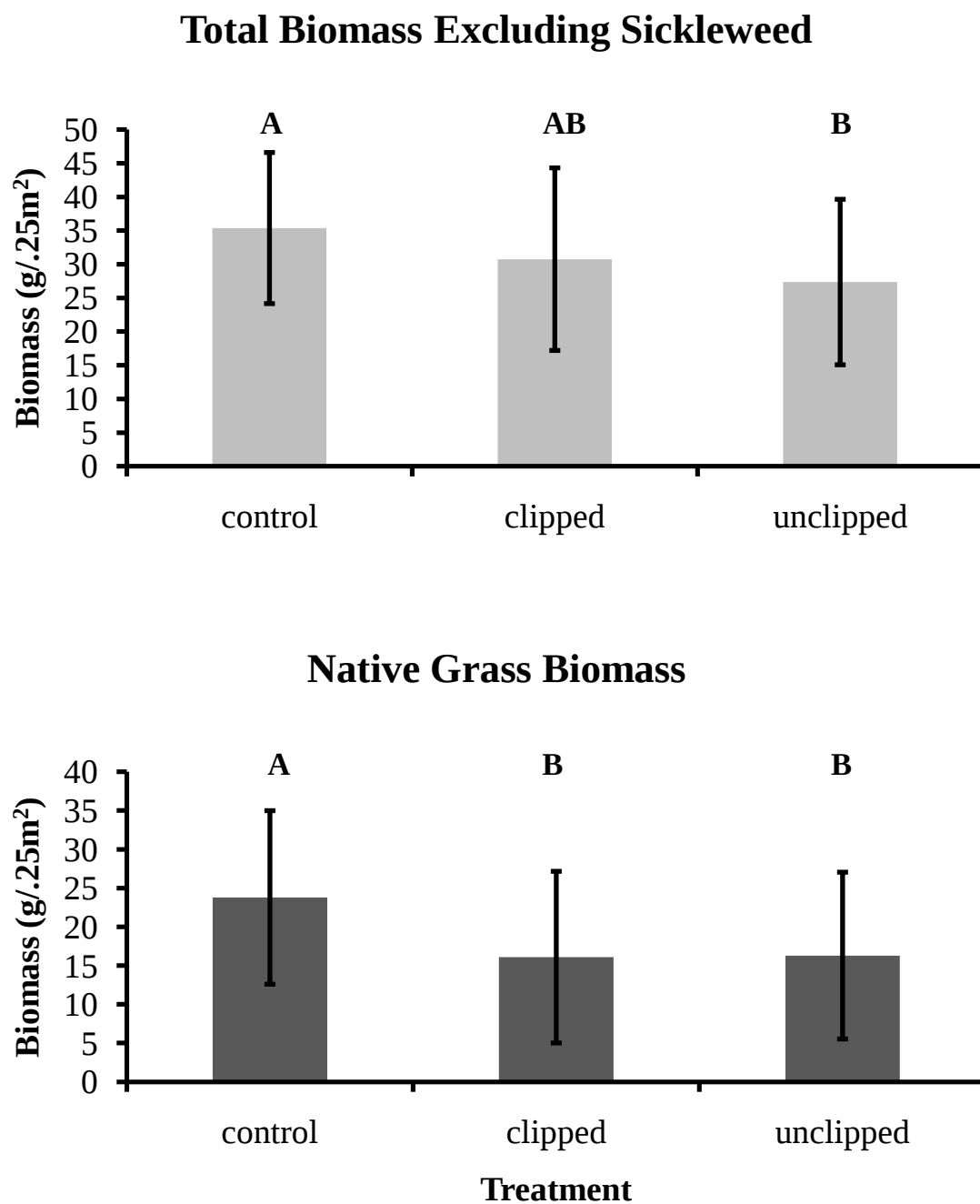


Figure 5. Mean aboveground biomass ($\pm$ standard deviation) of all vegetation excluding sickleweed (top) and native grass (bottom) by treatment. Letters indicate significant differences between treatments at $P \leq 0.05$.

biomass of introduced grasses and forbs was less than 2 g among treatments, which most likely produced the inconclusive results.

The negative effect of sickleweed on native grass biomass was stronger than its effect on total biomass. Native grass was significantly impacted in both unclipped and clipped sickleweed plots (31.5% and 32.3% less than control, respectively, Figure 5). Considering that the reduction in biomass of both treatments was similar, this may indicate a more complex interaction than solely competition for resources. This interaction follows a trend similar to that of negative allelopathic interactions found in other weeds; however, this is only speculation at this time.

The effect of sickleweed on native grasses at FPNG parallels the findings of Ortega and Pearson (2005) showing that a strong exotic invader can negatively affect perennial native grasses. In their study, the strong exotic invader was one that showed the ability to dominate invaded areas, *Centaurea maculosa* in their case. They also found a negative impact on perennial forbs, but not on annual forbs. I did not distinguish between the two, which may have led to my inconclusive findings in regard to forbs, although it should be noted that the presence of forbs was minimal (averaging 0.6 to 2.2 g/plot), which may have also led to the inconclusive findings.

The interaction between sickleweed and native grasses may be similar to what was found by Callaway and Aushehaug (2000) who found that *Centaurea maculosa*, a native of Eurasia, differentially impacted North American species over Eurasian species via root exudates. This was regardless of whether the Eurasian species were from their native range in Eurasia or their introduced range in North America. Although the

chemical they reported as causing the impact has since been discounted as inhibiting growth of other plants (Vivanco et al. 2004), the interaction still exists with a different exude. My results also follow the novel weapons and evolved increased competitive ability hypotheses discussed by Callaway and Ridenour (2004). Their observations and experiments with several species in their native and introduced ranges showed that weedy species generally are small components of plant communities in their native range. But in areas of introduction they come to dominate communities because of 'novel' biochemical interactions. Sickleweed follows this model because it has a wide geographic range in Eurasia where it is considered a common plant but with no accounts of it occurring in large dominating stands.

Allelopathic interactions in other Apiaceae have been reported. Junttila (1975) found that when giant hogweed seeds were placed with lettuce (*Lactuca sativa*) seeds, they caused reduced germination rates and a high level of abnormal germination due to chemicals in the hogweed seeds. However, that study was unable to produce similar effects on lettuce germination using other parts of the giant hogweed plant. The conclusions of Myras and Juntilla (1981) with giant hogweed were that allelopathic chemicals contributed to a reduction in biomass of other plants, although the reduction was not statistically significant. Hillman (1997) reported that poison hemlock (*Conium maculatum*) gained dominance in California grasslands due to allelopathic inhibition of other species. My results follow what other field studies have found in allelopathic inhibition of native North American species, and given that other members of Apiaceae

are reported to possess inhibitory compounds, the dominance of sickleweed at FPNG may be promoted by allelopathic properties.

Species diversity was significantly affected by treatments ($P=0.022$). Diversity was significantly lower in control plots than clipped plots ($P=0.19$), with unclipped plots not significantly different than either clipped or control ($P=0.092$ unclipped vs. clipped and $P=0.447$ unclipped vs. control, Table 5). Native and exotic species richness was not significantly affected by treatments ($P=0.303$ and $P=0.087$, respectively). Species richness and diversity of all plants was highest in 2008 (Table 5), which was most likely due to above average rainfall in the spring of 2008. Average native species richness was higher than exotic species richness in all but one treatment; clipped plots in 2008 had equal native and exotic species richness. The control plots had the largest difference between average native and exotic species richness. The control plots also had the lowest diversity in both years. The lack of sickleweed in the control plots had resulted in higher biomass of native grasses, which may have led to a disproportional abundance of one or more of the native grasses over other species, resulting in the lower diversity. The highest diversity was found in the clipped plots, which may have been a product of the mild disturbance associated with repeated sickleweed clipping, as disturbance has been associated with increased diversity (Connell 1978) and the success of introduced species (Hierro et al. 2006). Also, the presence of sickleweed may have facilitated an increase of exotic species cover and richness since exotic species have been found to compete better against coevolved species (Callaway and Aschehoug 2000). Sickleweed may be

Table 5. Average species richness by year, treatment, and species nativity and average Shannon diversity (H') by year and treatment. Statistical analysis showed diversity was significantly affected by treatment whereas species richness was not significantly affected by treatment. Differences in diversity at $P \leq 0.05$ indicated by different letters.

Year	Treatment	Species Richness		Diversity
		Native	Exotic	
2008	Clipped	6.0	6.0	1.8
	Unclipped	6.3	5.8	1.7
	Control	7.3	4.8	1.5
2009	Clipped	5.3	5.0	1.5
	Unclipped	5.5	4.8	1.1
	Control	6.3	4.0	1.2
Mean	Clipped	5.6	5.5	1.6 ^A
	Unclipped	5.9	5.3	1.4 ^{AB}
	Control	6.8	4.4	1.3 ^B

negatively affecting native species, as discussed above in the context of the biomass data, but not other introduced species.

Vegetative Reproduction and Regeneration from Roots

The number of taproot segments that survived cutting to generate a new shoots was high in all treatments (68-82%). While the percentage of shoot generation decreased with distance of the root segment from the crown, there was no significant difference in root segment survival or shoot production based on origin of root segment (Figure 6). This suggests that sickleweed tap roots can survive subsurface excision or fragmentation and may help explain observations of dense sickleweed growth in parts of Alkali West Allotment where pocket gophers were foraging heavily. Injury or fragmentation of sickleweed taproots by foraging and burrowing pocket gophers may have actually promoted vegetative reproduction.

Some generation of new shoots from roots of sickleweed was observed in the root boxes, especially where sickleweed roots were exposed to faint light through holes in the covering of the root boxes (Figure 7). These new shoots were mostly generated at the tips of branch roots rather than from main taproots. Enlarged root segments that terminated at small lateral branch roots were also observed in the root boxes (Figure 8). This had not been detected in the field, and at the time was thought to simply be a response to confined conditions in the root boxes. Lateral vegetative growth by sickleweed under field conditions was confirmed when, at the beginning of the second growing season, young sickleweed shoots began sprouting around plants transplanted from FPNG into a local garden in Brookings, SD. The plants had been transplanted during late August 2008, and

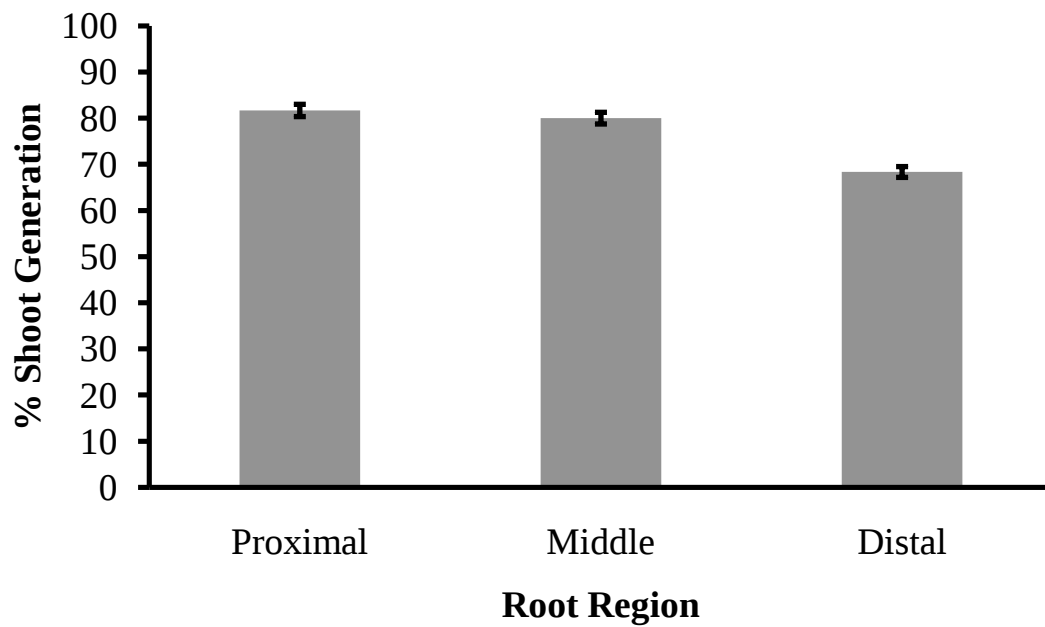


Figure 6. Mean shoot generation (root segments producing shoots after excision, +/- standard deviation) by 4-cm taproot region of origin. No significant differences in generative capacity by region were found at $P \leq 0.05$.



Figure 7. Photographs of sickleweed plants with roots that have produced new shoots below the soil surface where light came through tears in the aluminum foil covering the root box. Photograph at right is close up of the same plants in the picture at left.



Figure 8. Photograph at left is of a plant taken from greenhouse root boxes showing enlarged root segments at distal ends of branch roots. Photograph at right is of new sickleweed shoots produced early in the second growing season after transplanting three pots containing sickleweed plants from FPNG into a garden in Brookings, SD. Plants were top clipped to remove inflorescences. The new shoots were all produced outside of the pots and up to 1 m away at the tips of rather fleshy branch roots, similar to those produced by the plants grown in the root boxes.

were top-clipped during 2009 to remove inflorescences. In March 2010, juvenile shoots were noted up to 1 m away from the original plants. The new shoots were dug up to reveal their connection to fleshy roots ascending from deep in the soil, much like the shoot-producing branch roots observed in the root boxes (Figure 8). Propagation via root-sprouting clearly explains the circular patches of sickleweed that typify early infestations in FPNG (Figure 1).

The degree of lateral spread via the root system observed in sickleweed has been noted in no other Apiaceae in the region. While root sprouting has been ascribed to sickleweed in earlier accounts (Gress 1923, South Dakota Dept. of Ag. 1967), it is not mentioned in later descriptions of the plant in North America. On the other hand, Larina (2008), described sickleweed in Russia and stated that “*F. vulgaris* reproduces vegetatively by rootstocks from the second year of its life.” Larina also describes sickleweed as “a biannual plant that is able to develop as a perennial.” Again, the sickleweed creating problems in FPNG appears strictly perennial.

Barney et al. (2009) found that *Artemisia vulgaris* evolved a more competitive phenotype by producing a more extensive rhizome system, in its introduced range. Richards et al. (2006) also implicated phenotypic plasticity as an important trait of invading species in general. Sickleweed shows a large range of phenotypic plasticity, from acting as an annual in parts of its native range to behaving as a perennial that spreads vegetatively in other areas including its introduced range.

The ability of sickleweed plants to propagate by root sprouting and root fragments to produce new shoots is similar to the capabilities of some of the most invasive, difficult

to control weeds of the FPNG area. Canada thistle (Hamdoun 1972), leafy spurge (Raju et al. 1964, Selleck et al. 1962), Russian knapweed (*Centaurea repens* L.; Koloren 2005), and hoary cress (*Lepidium draba* Desv.; Harradine 1995) are all South Dakota listed noxious weeds that spread vegetatively via rootstocks and have shown the ability to resprout after root fragmentation. The ability of sickleweed to spread and regenerate by roots undoubtedly contributes to its persistence and spread at FPNG.

Conclusion and Recommendations

Sickleweed at FPNG is a perennial plant with high level of germination, reproducing and spreading by sprouting from rootstocks, that negatively impacts the growth and production of native grasses. This study confirms the need for aggressive control measures.

Much research is needed on sickleweed given how much of the published information on sickleweed is either wrong or based on its behavior in a specific region. For example, in Eurasia sickleweed seems to act as annual or biennial in southern areas and a perennial in northern areas. Genetic studies may be able to resolve the origin of introduced populations based on specific genetic markers, and possibly predict if specific populations are more invasive than others.

Future descriptions of sickleweed must acknowledge the capacity of the plant, in its perennial form, to spread laterally via root sprouting. This has important management implications (discussed below) but it also is important for our current knowledge of

Apiaceae as no other members of Apiaceae in the region spread laterally from rootstocks and I was unable to find any other reports of Apiaceae spreading like this in the literature.

Research should be conducted on biological and mechanical means to control sickleweed. At this time herbicide treatments are the only effective option at FPNG, but repeated herbicide treatments have high cost, incomplete control, and negative impacts on native species. My germination work, showing that seed begins losing viability after 1 year, indicates that in areas where sickleweed behaves as an annual or biennial, a few successive years of mowing or grazing would be enough to control the plant. However, in places where sickleweed spreads by root sprouts, mowing and/or grazing may not affect its persistence and spread. Matthew Sleigh (B and T world Seed, personal communication) reported that sickleweed is the host plant for the beetle *Cardoria scutellata*. Research should be conducted to determine if this beetle has potential as a biological control of sickleweed in North America.

Studies should also be conducted on how livestock grazing may influence sickleweed abundance and spread. My personal observations at FPNG (specifically the County Line #18 allotment) are that early spring grazing limits sickleweed's ability to produce seed and possibly its ability to spread vegetatively. I believe this may help control the spread of this weed in conjunction with other control methods.

In personal communications I have become aware of sickleweed infestations in several South Dakota counties (Brookings, Charles Mix, and Pennington) and one Nebraska county (Boyd) that were not found in published literature or in the USDA Plants Database. This indicates that sickleweed may be more prevalent than represented

in available sources or that sickleweed is still actively spreading in the US. This highlights the need of increased vigilance to detect new infestations of sickleweed and other exotic species that may pose a problem in rangelands. Early detection of, and rapid response to, infestations is much more productive than assessing and treating the problem years after establishment.

Vascular Flora of the Fort Pierre National Grassland

The botanical survey of FPNG documented 246 vascular plant species (Appendix A). Of the documented families there was one gymnosperm family, Cupressaceae, with one species represented. Of the angiosperms there were 44 dicot families represented by 171 species and 13 monocot families comprised of 75 species. The largest family was Asteraceae with 49 species, followed by Poaceae (46 species) and Fabaceae (19 species) for the three most specious families. Exotic species (based on USDA Plants database, species non-native to the continental US) constituted 24% of the documented flora with 58 species not native to the continental United States.

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Appendix A

Annotated Checklist of the Vascular Plants of the Fort Pierre National Grassland.

Nomenclature follows Kaul et al. (2006).

*indicates county records from the Great Plains Flora Association (1997).

AGAVACEAE, the century-plant family

***Yucca glauca* Nutt.**, yucca. Common on xeric slopes, Jones.

ALISMATACEAE, the Water Plantain Family

***Alisma triviale* Pursh**, large flowered water-plantain. Common along stock-ponds and wetlands, Lyman and Stanley.

***Sagittaria calycina* Engelm.**, arrowhead. Occasional muddy shores, flats, and flooded areas, Lyman and Stanley*.

***Sagittaria cuneata* E. Sheld.**, duck-potato. Occasional, muddy shores and margins of stock ponds Lyman and Stanley*.

AMARANTHACEAE, the amaranth family

***Amaranthus retroflexus* L.**, redroot pigweed. Occasional along roadsides and disturbed areas, Lyman and Stanley*.

***Amaranthus tuberculatus* (Moq.) J. D. Sauer**, water-hemp. Occasional, shores and banks of streams and stock ponds, Lyman and Stanley*.

APIACEAE, the parsley family

***Daucus carota* L.**, wild carrot. Locally common in parking area of Richland Wildlife Area, Jones.

***Falcaria vulgaris* Bernh.**, sickleweed. Common, generally uplands but in all terrestrial habitats in heavily infested areas, Jones, Lyman, and Stanley.

***Lomatium foeniculaceum* (Nutt.) J. M. Coult. & Rose**, yellow wild-parsley. Common in uplands, Lyman and Stanley.

***Musineon divaricatum* (Pursh.) Nutt. ex Torr. & A. Gray**, plains musineon. Common in uplands, Lyman and Stanley.

APOCYNACEAE, the dogbane or milkweed family

***Apocynum cannabinum* L.**, dogbane. Occasional along wet roadsides, Lyman and Stanley.

***Asclepias pumila* (A. Gray) Vail**, dwarf milkweed. Common in uplands and prairie dog towns, Jones, Lyman, and Stanley*.

***Asclepias speciosa* Torr.**, showy milkweed. Occasional in uplands, Jones*, Lyman*, and Stanley.

***Asclepias syriaca* L.**, Common milkweed. Occasional in uplands, Lyman and Stanley.

***Asclepias verticillata* L.**, whorled milkweed. Common in uplands and prairie dog towns, Jones, Lyman, and Stanley.

ASTERACEAE, the sunflower family

***Achillea millefolium* Piper**, yarrow. Common in uplands, Jones*, Lyman, and Stanley*.

***Ambrosia psilostachya* DC.**, Western ragweed. Occasional in uplands, Stanley.

***Ambrosia trifida* L.**, Giant ragweed. Common in disturbed sites, Lyman and Stanley.

***Antennaria microphylla* Rydb.**, pussytoes. One patch in Alkali (Middle) Allotment, Stanley.

***Antennaria parvifolia* Nutt.**, pussytoes. One patch in Grass Creek (#1) Allotment, Stanley.

***Artemisia absinthium* L.**, wormwood. One infestation near gate in Dry Hole (Chester) Allotment, Lyman.

***Artemisia biennis* Willd.**, Biennial wormwood. One patch in dry stock-pond in Dry Hole (Chester) Allotment, Lyman.

***Artemisia frigida* Willd.**, fringed sage. Occasional in uplands, Stanley.

***Artemisia ludoviciana* Nutt.**, white sage. Occasional in uplands, Jones* and Stanley.

***Aster ericoides* L.**, heath aster. Common in uplands, Lyman and Stanley.

***Aster lanceolatus* Willd. Subsp. *hesperius* (A. Gray) Semple & Cheiel.**, panicked aster. Occasional along margins of stock ponds, Jones.

***Aster oblongifolius* Nutt.**, aromatic aster. Common in uplands, Jones, Lyman, and Stanley*.

***Bidens comosa* (A. Gray) Wiegand**, beggar-ticks. Common along margins of stock ponds, Jones, Lyman, and Stanley*.

***Bidens frondosa* L.**, beggar-ticks. Occasional, along margins of stock dams, Jones and Stanley*.

***Brickellia eupatorioides* L. Shinnery**, false boneset. Common in uplands, Jones, Lyman, and Stanley.

***Carduus nutans* L.**, musk thistle. Occasional in uplands, Lyman.

***Centaurea repens* L.**, Russian knapweed. One infestation on dam in Nels (Home) Allotment, Lyman.

***Cirsium arvense* (L.) Scop.**, Canada thistle. Common in low areas and along stock ponds, Lyman and Stanley.

***Cirsium undulatum* (Nutt.) Spreng.**, wavy-leaf thistle. Common in uplands, Jones, Lyman*, and Stanley.

***Conyza canadensis* (L.) Cronquist**, horseweed. Common in uplands, Stanley.

***Coreopsis tinctoria* Nutt.**, plains coreopsis. One patch in temporarily flooded roadside, Jones* and Lyman.

***Dyssodia papposa* (Vent.) A. S. Hitchc.**, fetid-marigold. Occasional, along roadway and prairie dog towns, Jones* and Stanley.

***Echinacea angustifolia* DC.**, narrow-leaf purple cone flower. Common in uplands, Jones, Lyman, and Stanley.

***Erigeron strigosus* Muhl. ex Willd.**, daisy fleabane. Common in uplands, Jones, Lyman, and Stanley.

***Filago arvensis* L.**, fluffweed. Occasional in uplands, Stanley.

***Grindelia squarrosa* (Pursh) Dunal**, curly-cup gumweed. Common in uplands, Jones, Lyman, and Stanley.

***Gutierrezia sarothrae* (Pursh) Britton & Rusby**, broom snakeweed. Common on hilltops, Jones and Stanley.

***Helianthus annuus* L.**, common sunflower. Common on disturbed sites and uplands, Jones, Lyman*, and Stanley.

***Helianthus maximiliani* Shrad.**, Maximillian's sunflower. Common in low areas and along stock ponds, Jones* Lyman and Stanley.

***Helianthus pauciflorus* Nutt.**, stiff sunflower. Common in uplands, Jones, Lyman, and Stanley.

***Lactuca pulchella* (Pursh) DC.**, pretty blue lettuce. Common in uplands, Jones*, Lyman, and Stanley.

***Lactuca serriola* L.**, prickley lettuce. Common in uplands, Jones, Lyman, and Stanley.

***Liatris punctata* Hook.**, dotted gayfeather. Common in uplands, Jones, Lyman, and Stanley.

***Lygodesmia juncea* (Pursh) D. Don ex Hook**, skeletonweed. Common in uplands, Jones, Lyman, and Stanley.

***Picradeniopsis oppositifolia* (Nutt.) Rydb. ex Britton**, oppositeleaf bahia. Common along roadsides, Lyman and Stanley*.

***Ratibida columnifera* (Nutt.) Wooten & Standl.**, prairie cone flower. Common in uplands, Jones, Lyman, and Stanley.

***Senecio integerrimus* Nutt.**, groundsel. Occasional in uplands, Stanley.

***Senecio plattensis* Nutt.**, prairie ragwort. Common in uplands and roadsides, Lyman* and Stanley.

***Solidago canadensis* L.**, Canada goldenrod. Occasional in uplands, Stanley.

***Solidago gigantea* Aiton**, late goldenrod. One patch in small depression in 50-50 West Allotment, Lyman and Jones*.

***Solidago missouriensis* Nutt.**, prairie goldenrod. Common in uplands, Jones, Lyman, and Stanley.

***Solidago mollis* Bartl.**, soft goldenrod. One patch in Mallard (North) Allotment, Lyman, Jones*, and Stanley*.

***Solidago rigida* L.**, stiff goldenrod. Common in uplands, Jones, Lyman, and Stanley.

***Sonchus arvensis* L.**, field sow-thistle. Common in low areas and wetlands, Jones and Stanley.

***Taraxacum officinale* G. H. Weber ex F. H. Wigg.**, dandelion. Common in uplands, Stanley.

***Tragopogon dubius* Scop.**, goat's-beard. Common in uplands, Jones*, Lyman, and Stanley*.

***Vernonia fasciculata* Michx.**, ironweed. Wetland area in Mallard (North) Allotment, Lyman.

***Xanthisma spinulosum* (Pursh) D Morgan & R. L. Hartm.**, cutleaf ironplant. Common in prairie dog towns, Jones*, Lyman, and Stanley.

***Xanthium strumarium* L.**, cocklebur. Common in disturbed areas, Lyman and Stanley.

BORAGINACEAE, the borage family

***Lappula squarrosa* (Retz.) Dumort.**, stickseed. Common in uplands, Lyman and Stanley.

***Mertensia lanceolata* (Pursh) A. DC.**, bluebells. Occasional in uplands, Lyman and Stanley.

***Plagiobothrys scouleri* (Hook. & Arn.) I. M. Johns.**, popcorn flower. One patch from low area in Mustang West Allotment, Stanley.

BRASSICACEAE, the mustard family

***Camelina microcarpa* DC.**, small seeded false flax. Occasional in uplands, Jones.

***Chorispora tenella* (Pallas) DC.**, blue mustard. Common in disturbed areas, Jones and Stanley.

***Conringia orientalis* (L.) Dumortier**, hare's-ear mustard. Common in disturbed areas and uplands, Lyman and Stanley.

***Descurainia sophia* (L.) Webb ex Prantl**, tansy mustard. Occasional in disturbed areas, Jones and Stanley*.

***Draba reptans* (Lam.) Fernald**, whitlow-grass. Common in disturbed areas, Lyman and Stanley*.

***Erysimum asperum* (Nutt.) DC.**, western wallflower. Common in uplands, Jones, Lyman, and Stanley.

***Erysimum repandum* L.**, bushy wallflower. Occasional in uplands, Stanley.

***Hesperis matronalis* L.**, dame's-rocket. One patch near shelterbelt in Alkali (Middle) Allotment, Stanley.

***Lepidium densiflorum* Schrader**, pepper-grass. Occasional in disturbed areas, Lyman* and Stanley.

***Lepidium chalepensis* L.**, lens-podded hoary cress. Large infestation on private land and ditch by Dobe Flat (East) Allotment, Lyman.

***Lepidium perfoliatum* L.**, clasping pepper-grass. Occasional in disturbed areas, Stanley.

***Rorippa palustris* (L.) Besser. var *hispida* (Desv.) Rydb.**, bog yellow cress. Common along stock ponds, Jones, Lyman, and Stanley*.

***Sinapis arvensis* L.**, charlock. Occasional in disturbed area, Stanley.

***Sisymbrium altissimum* L.**, tumbling mustard. Common in disturbed areas and uplands, Jones and Stanley.

Stanleya pinnata* (Nutt.) Britton var. *pinnata, princes-plume. Occasional in Alkali (Home) Allotment, Lyman and Stanley*.

***Thlaspi arvense* L.**, field penny-cress. Common in disturbed areas, Lyman and Stanley.

CACTACEAE, the cactus family

***Coryphantha missouriensis* (Sweet) Britton & Rose**, Missouri pincushion. Occasional in xeric uplands, Lyman* and Stanley.

***Opuntia polyacantha* Haw.**, plains prickly-pear. Common in xeric uplands and clay pans, Stanley.

CALLITRICHACEAE, the water-starwort family

***Callitriche hermaphrodita* L.**, water-starwort. Occasional, in wetland near roadside, Lyman.

CAMPANULACEAE, the bellflower family

***Triodanis leptocarpa* (Nutt.) Nieuwl.**, slender-fruited venus's-looking-glass. Common in disturbed uplands, Lyman and Stanley.

CAPRIFOLIACEAE, the honeysuckle family

***Symphoricarpos occidentalis* Hook.**, western snowberry. Common in uplands, Jones*, Lyman, and Stanley.

CARYOPHYLLACEAE, the pink family

***Silene latifolia* Poiret**, white campion. Occasional in uplands in Dry Hole (Chester) Allotment, Lyman.

CERATOPHYLLACEAE, the hornwort family

***Ceratophyllum demersum* L.**, coontail. Occasional in stock-ponds and wetlands, Lyman.

CHENOPODIACEAE, the goosefoot family

***Chenopodium album* L.**, lamb's quarters. Occasional in disturbed areas, Stanley.

***Kochia scoparia* (L.) Schrad.**, kochia. Common in disturbed areas, Lyman and Stanley.

***Monolepis nuttalliana* (Roem. & Schult.) Greene**, poverty-weed. Occasional in disturbed areas, Lyman* and Stanley.

COMMELINACEAE, the spiderwort family

***Tradescantia bracteata* Small ex Britton & Brown**, spiderwort. Common in uplands, Jones, Lyman, and Stanley.

CONVOLVULACEAE, the morning-glory family.

***Calystegia macounii* (Greene) Brummit**, Macoun's bindweed. Occasional in low areas, Lyman and Stanley.

***Convolvulus arvensis* L.**, field bindweed. Occasional along roadside, Jones and Lyman*.

CUPRESSACEAE, the cypress family

***Juniperus virginiana* L.**, eastern red-cedar. Infrequent along roadsides and drainages, Stanley.

CYPERACEAE, the sedge family

***Bulboschoenus fluviatilis* (Torr.) M. T. Strong**, river burush. Occasional in stock ponds and low areas, Jones*, Lyman, and Stanley.

***Bulboschoenus maritimus* (L.) Palla**, alkali bulrush. Occasional in stock ponds and wetlands, Jones, Lyman, and Stanley.

***Carex eleocharis* L. H. Bailey**. Occasional in uplands, Lyman and Stanley*.

***Carex heliophila* Mack.**, sun sedge. Occasional in uplands, Stanley.

***Carex molesta* Mack. ex Bright**. Occasional around stock ponds, Stanley.

***Cyperus acuminatus* Torr. & Hook. ex Torr.**, flat sedge. In drainage in Reservation Road #18 Allotment, Stanley.

***Cyperus odoratus* L.**, rusty flatsedge. Common, muddy shores and flats around stock ponds and wetlands, Jones and Stanley.

***Eleocharis acicularis* (L.) Roem. & Schultes**, hair-grass. Occasional in drainages, Lyman and Stanley*.

***Eleocharis erythropoda* Steud.**, redstem spike-rush. Common in wetland in Cactus Flats Allotment, Stanley.

***Eleocharis palustris* (L.) Roem. & Schultes**, spike-rush. Common in stock ponds, Jones and Lyman.

Schoenoplectus acutus* (Muhl. ex Bigelow) A. Love & D. Love var. *acutus, hardstem bulrush. Common in stock-pond in Alkali (Home) Allotment, Lyman and Stanley*.

***Schoenoplectus pungens* (Vahl.) Palla**, common threesquare. Occasional in drainage in Ditch Creek (West/North) Allotment, Lyman and Stanley*.

***Schoenoplectus tabernaemontani* (C. C. Gmel.) Palla**, softstem bulrush. Common in stock pond in Reed Ranch (West) Allotment, Lyman.

ELAEAGNACEAE, the oleaster family

***Elaeagnus angustifolia* L.**, Russian-olive. Occasional along stock ponds, Jones.

EUPHORBIACEAE, the spurge family

***Euphorbia marginata* Pursh**, snow-on-the-mountain. Common in disturbed areas and prairie dog towns, Jones, Lyman, and Stanley*.

***Euphorbia spathulata* Lam.** Common in uplands, Jones, Lyman, and Stanley*.

***Euphorbia stictospora* Englem.**, mat spurge. Occasional along roadsides, Jones*, Lyman*, and Stanley.

FABACEAE, the pea family

***Amorpha canescens* Nutt. ex Pursh**, leadplant. Common in uplands, Lyman and Stanley.

***Amorpha fruticosa* L.**, wild indigo. Infrequent along drainage in Antelope Creek (North Big) Allotment, Lyman* and Stanley.

***Amorpha nana* Nutt.**, Dwarf wild indigo. Common in uplands, Jones*, Lyman, and Stanley.

***Astragalus agrestis* Douglas ex G. Don**, field milk-vetch. Occasional in uplands, Jones.

***Astragalus flexuosus* (Dougl. ex Hook.) G. Don**, pliant milk-vetch. Common in uplands, Jones and Lyman.

***Astragalus racemosus* Pursh**, alkali milk-vetch. Common in low areas, Jones, Lyman, and Stanley.

***Caragana arborescens* (L.) Lam.**, pea shrub. Planted in shelterbelt in Grass Creek Allotment, Stanley.

***Dalea purpurea* Venten.**, purple prairie-clover. Common in uplands, Jones, Lyman, and Stanley.

***Glycyrrhiza lepidota* Pursh**, wild licorice. Occasional in low areas, Jones*, Lyman, and Stanley*.

***Lotus purshianus* (Benth.) F. Clements & E. Clements ex Otley**, prairie trefoil. Occasional in uplands, Jones* Lyman, and Stanley*.

***Medicago sativa* L. subsp *falcata* (L.) Archangeli**, yellow flowered alfalfa. Common in uplands, Lyman and Stanley.

Medicago sativa* L. subsp *sativa, purple flowered alfalfa. Common in uplands, Jones, Lyman, and Stanley.

***Melilotus albus* Medikus**, white sweet-clover. Common in uplands, Jones and Lyman.

***Melilotus officinalis* (L.) Pallas**, yellow sweet-clover. Common in uplands, Stanley.

***Oxytropis lambertii* Pursh**, purple locoweed. Common in uplands, Jones, Lyman, and Stanley*.

***Pediomelum argophyllum* (Pursh) Grimes**, silver-leaf scurfpea. Common in uplands, Lyman and Stanley.

***Pediomelum cuspidatum* (Pursh) Rydb.**, tall breadroot scurfpea. Common in uplands, Jones, Lyman, and Stanley*.

***Pedimelum esculentum* (Pursh) Rydb.**, Indian-breadroot. Common in uplands, Jones, Lyman*, and Stanley.

***Vicia americana* Muhl. ex Willd.**, American vetch. Common in uplands, Lyman* and Stanley.

GERANIACEAE, the geranium family

***Geranium carolinianum* L.**, carolina crane's-bill. Occasional in uplands, Jones*, Lyman*, and Stanley.

GROSSULARIACEAE, the gooseberry family

***Ribes odoratum* H. L. Wendl.**, golden current. Occasional in low areas, Jones, Lyman*, and Stanley.

HYDROPHYLLACEAE, the waterleaf family

***Ellisia nyctelea* (L.) L.**, waterpod. Occasional in disturbed areas, Jones, Lyman, and Stanley*.

IRIDACEAE, the iris family

***Sisyrinchium montanum* Greene**, blue-eyed grass. One patch in Alkali (Middle) Allotment, Stanley.

JUNCACEAE, the rush family

***Juncus arcticus* Willd. var. *balticus* (Willd.) Trautv.**, baltic rush. Occasional along stock ponds, Jones.

***Juncus torreyi* Coville**, Torrey's rush. Occasional along stock ponds, Jones.

LAMIACEAE, the mint family

***Hedeoma hispida* Pursh**, rough false pennyroyal. Common in disturbed areas, Jones and Stanley.

***Lycopus americanus* Muhl. ex W. Barton**, American water-horehound. Common in over flow area of stock tank in Grass Creek Allotment, Stanley.

***Mentha arvensis* L.**, field mint. Common along stock ponds and wetlands, Jones, Lyman*, and Staley.

***Nepeta cataria* L.**, catnip. Common in low areas, Jones and Lyman.

***Salvia reflexa* Hornem.**, rocky mountain sage. Infrequent in low areas, Lyman and Stanley*.

***Sideritis montana* L.**, mountain ironwort. One infestation in Alkali Allotment, Lyman.

LEMNACEAE, the duckweed family

***Lemna turionifera* Landolt.**, duckweed. Occasional in stock ponds and wetlands, Lyman.

LILIACEAE, the lily family

***Allium textile* A. Nels. & J. F. Macbr.**, prairie onion. Common in uplands, Lyman and Stanley.

***Zigadenus venenosus* S. Watson**, death camas. Common in uplands, Jones and Lyman.

LINACEAE, the flax family

***Linum rigidum* Pursh**, stiff flax. Common in uplands, Lyman and Stanley.

LYTHRACEAE, the loosestrife family

***Ammannia robusta* Heer & Regel**, toothcup. Occasional in low areas, Stanley.

MALVACEAE, the mallow family

***Sphaeralcea coccinea* (Nutt.) Rydb.**, red false mallow. Common in uplands, Jones, Lyman*, and Stanley.

MIMOSACEAE, the mimosa family

***Mimosa quadrivalvis* L. var. *nuttallii* (DC.) Beard ex Barneby**, sensitive briar.
Common in uplands, Jones, Lyman, and Stanley.

NYCTAGINACEAE, the four-o'clock family

***Mirabilis hirsuta* (Pursh) MacMill.**, hairy four-o'clock. Common in uplands, Jones, Lyman, and Stanley.

***Mirabilis nyctaginea* (Michx.) MacMill.**, wild four-o'clock. Infrequent along roadsides, Stanley.

OLEACEAE, the olive family

***Fraxinus pennsylvanica* Marshall**, green ash. Infrequent along drainages, also planted in shelterbelts, Lyman* and Stanley.

ONOGRACEAE, the evening-primrose family

***Epilobium ciliatum* Raf.**, fringed willow-herb. Common in over flow area of stock tank in Grass Creek Allotment, Stanley.

***Gaura coccinea* Pursh**, scarlet gaura. Common in uplands, Jones, Lyman, and Stanley.

***Oenothera biennis* L.**, common evening-primrose. Common along stock ponds and wetlands, Jones, Lyman, and Stanley.

OXALIDACEAE, the wood-sorrel family

***Oxalis dillenii* Jacq.**, gray oxalis. Common in uplands, Jones, Lyman*, and Stanley.

PLANTAGINACEAE, the plantain family

***Plantago elongata* Pursh**, slender plantain. Common in disturbed areas, Lyman and Stanley.

***Plantago patagonica* Jacq.**, wooly plantain. Common in disturbed areas and prairie dog towns, Lyman* and Stanley.

POACEAE, the grass family

***Agropyron cristatum* (L.) Gaertn.**, crested wheatgrass. Common in uplands, Jones, Lyman, and Stanley.

***Agrostis stolonifera* L.**, redtop. Wet roadside by Timber Creek (Home) Allotment, Stanley.

***Alopecurus carolinianus* Walter**, Carolina foxtail. Common in wetlands, Jones and Stanley.

***Andropogon gerardii* Vitman**, big bluestem. Common on shallow clay hillsides, Jones, Lyman, and Stanley.

***Aristida purpurea* Nutt.**, red three-awn. Common in uplands and prairie dog towns, Jones, Lyman, and Stanley.

***Avena fatua* L. var. *sativa* (L.) Hausskn.**, cultivated oats. Occasional along roadsides, Stanley.

***Beckmannia syzigachne* (Steud.) Fernald.**, American slough grass. Occasional in wetlands, Lyman and Stanley.

***Bouteloua curtipendula* (Michx.) Torr.**, side outs grama. Common on shallow clay hillsides, Jones, Lyman, and Stanley.

***Bouteloua gracilis* (Willd. ex Kunth) Lag. ex Griffiths**, blue grama. Common in uplands and prairie dog towns, Jones, Lyman*, and Stanley.

***Bromus inermis* Leyss.**, smooth brome. Common in uplands and road sides, Jones, Lyman, and Stanley.

***Bromus japonicus* Thunb. ex Murr.**, Japanese brome. Common in uplands and disturbed areas, Jones, Lyman*, and Stanley.

***Bromus tectorum* L.**, downy brome. Occasional in uplands and disturbed areas, Stanley.

***Buchloe dactyloides* (Nutt.) Englem.**, buffalo grass. Common in uplands and prairie dog towns, Jones and Stanley.

***Calamovilfa longifolia* (Hook.) Scribn.**, prairie sandreed. Occasional in uplands, Jones*, Lyman*, and Stanley.

***Cenchrus longispinus* (Hack.) Fernald**, sandbur. Common in disturbed areas, Stanley.

***Echinochloa muricata* (P. Beauv.) Fernald.**, barnyard grass. Occasional along roadsides, Lyman and Stanley.

***Elymus canadensis* L.**, Canada wildrye. Occasional in low areas, Jones and Stanley*.

***Elymus hispidus* (Opiz.) Meld.**, intermediate wheatgrass. Common in uplands, Jones, Lyman, and Stanley.

***Elymus lanceolatus* (Scribn. & J. G. Smith) Gould**, thickspike wheatgrass. Occasional in drainage, Stanley.

***Elymus smithii* (Rydb.) Gould**, western wheatgrass. Common in all terrestrial habitats, Jones, Lyman, and Stanley.

***Eragrostis cilianensis* (All.) Vignolo ex Janch.**, stinkgrass. Common in disturbed areas, Jones*, Lyman, and Stanley.

***Eragrostis pectinacea* (Michx.) Nees ex Steud.**, Carolina lovegrass. Infrequent along roadside, Stanley.

***Hordeum jubatum* L.**, foxtail barley. Common in low areas, Jones*, Lyman, and Stanley.

***Hordeum pusillum* Nutt.**, Little barley. Common in disturbed areas, Lyman and Stanley.

***Koeleria macrantha* (Ledeb.) Schult.**, june grass. Common in uplands, Lyman, and Stanley.

***Leptochloa fusca* (L.) Kunth subsp. *fascicularis* (Lam.) N. Snow**, strangle-top.
Common along drainage in Ditch Creek (West/North) Allotment, Lyman.

***Panicum capillare* L.**, witchgrass. Common in disturbed areas, Jones*, Lyman, and Stanley.

***Panicum virgatum* L.**, switchgrass. Occasional in uplands, Jones, Lyman*, and Stanley.

***Phalaris arundinacea* L.**, reed canary grass. Occasional along wetlands and stock ponds, Stanley.

***Phleum pratense* L.**, timothy. One small patch in Dry Hole (Chester) Allotment, Lyman and Stanley*.

***Phragmites australis* (Cav.) Trin ex Steud.**, common reed. One large patch near Sheriff Dam, Lyman.

***Poa compressa* L.**, Canada bluegrass. Common in low areas, Stanley.

***Poa pratensis* L.**, Kentucky bluegrass. Common in all terrestrial habitats, Lyman and Stanley.

***Schedonnardus paniculatus* (Nutt.) Trel.**, tumblegrass. Common in uplands and prairie dog towns, Lyman and Stanley.

***Schizachyrium scoparium* (Michx.) Nash**, little bluestem. Common on shallow clay hillsides, Jones and Stanley.

***Setaria pumila* (Poir.) Roem & Schult.**, yellow foxtail. Common in disturbed areas, Stanley.

***Setaria viridis* (L.) P. Beauv.**, green foxtail. Common in disturbed areas, Jones, Lyman, and Stanley.

***Spartina pectinata* Link**, prairie cordgrass. Common along stock ponds and drainages, Jones, Lyman, and Stanley*.

***Sporobolus compositus* (Poir.) Merr.**, rough dropseed. Occasional in uplands, Stanley.

***Sporobolus cryptandrus* (Torr.) A. Gray**, sand dropseed. Common in uplands, Jones* and Stanley.

***Stipa comata* Trin. & Rupr.**, needle-and-thread. Common in uplands, Jones, Lyman, and Stanley*.

***Stipa spartea* Trin.**, porcupine grass. Common in uplands, Jones, Lyman, and Stanley.

***Stipa viridula* Trin.**, green needle grass. Common in uplands, Jones, Lyman*, and Stanley.

***Triticum aestivum* L.**, wheat. Occasional along roadsides, Stanley.

***Vulpia octoflora* (Walter) Rydb.**, sixweeks-fescue. Common in uplands and disturbed areas, Lyman and Stanley.

***xElyhordeum macounii* (Vasey) Barkworth & D. R. Dewey**, Macoun's barley. Occasional in low areas, Jones and Stanley.

POLEMONIACEAE, the phlox family

***Collomia linearis* Nutt.**, collomia. Common in uplands, Lyman and Stanley.

***Phlox hoodii* Rich.**, Hood's phlox. Occasional on xeric hilltops, Jones.

POLYGALACEAE, the milkwort family

***Polygala verticillata* L.**, whorled milkwort. Infrequent in Antelope Creek (North Big) Allotment, Stanley.

POLYGONACEAE, the buckwheat family

***Polygonum aviculare* L.**, knotweed. Common in disturbed areas, Stanley.

***Polygonum coccineum* Muhl. ex Willd.**, scarlet smartweed. Common in wetlands and low areas, Lyman and Stanley.

***Polygonum convolvulus* L.**, black-bindweed. Occasional along road side, Lyman* and Stanley.

***Polygonum lapathifolium* L.**, nodding smartweed. Occasional along stockpond, Jones.

***Rumex crispus* L.**, curly dock. Common in low areas and wetland, Jones, Lyman, and Stanley.

***Rumex fueginus* Philippi**, golden dock. Occasional in wetlands, Jones and Stanley.

***Rumex salicifolius* Weinm.**, willow dock. Occasional in wetlands, Stanley.

***Rumex stenophyllus* Ledeb.**, Toothed dock. Occasional in wetlands, Stanley.

PONTEDERIACEAE, the pickerel-weed family

***Zosterella dubia* (Jacq.) J. K. Small**, water star-grass. Occasional in stock pond, Jones.

POTAMOGETONACEAE, the pondweed family.

***Potamogeton nodosus* Poir.**, floating-leaf pondweed. Occasional in stock ponds and wetlands Jones, Lyman, and Stanley*.

***Potamogeton pusillus* L.**, small pondweed. Occasional in stock ponds and wetlands, Lyman.

PRIMULACEAE, the primrose family

***Androsace occidentalis* Pursh**, western rock-jasmine. Occasional in uplands, Stanley.

RANUNCULACEAE, the buttercup family

***Delphinium virescens* Nutt.**, prairie larkspur. Occasional in uplands, Stanley.

***Ranunculus subrigidus* W. Drew**, white water-crowfoot. Common in stock ponds, Lyman and Stanley*.

ROSACEAE, the rose family

***Potentilla recta* L.**, sulfur cinquefoil. Common in Richland Wildlife Area and Number Two (North West) Allotments, Jones.

***Prunus americana* Marsh.**, American plum. Occasional in drainages, Lyman and Stanley*.

***Prunus virginiana* L.**, chokecherry. Occasional in drainages, Lyman and Stanley.

***Rosa acicularis* Lindl.**, prickly wild-rose. Occasional in uplands, Lyman.

***Rosa arkansana* Porter ex Porter & J. M. Coult.**, Arkansas rose. Common in uplands, Lyman and Stanley.

RUBIACEAE, the coffee family

***Galium aparine* L.**, cleavers. Occasional near Sheriff Dam, Jones.

SALICACEAE, the willow family

***Populus deltoides* Bartr. ex Marsh.**, plains cottonwood. Common along stock ponds and drainages, Jones*, Lyman, and Stanley.

***Salix amygdaloides* N. J. Andersson**, peach-leaf willow. Common along stock ponds and drainages, Jones, Lyman*, and Stanley.

SCROPHULARIACEAE, the figwort family

***Penstemon albidus* Nutt.**, white beardtongue. Common in uplands, Jones, Lyman, and Stanley.

***Penstemon gracilis* Nutt.**, slender beardtongue. Occasional in uplands, Lyman* and Stanley.

***Penstemon grandiflorus* Nutt.**, large beardtongue. Occasional in uplands, Jones and Stanley.

SOLANACEAE, the nightshade family

***Physalis longifolia* Nutt.**, common ground-cherry. Occasional in uplands, Lyman.

***Solanum rostratum* Dunal**, buffalo-bur. Common along roadsides and in prairie dog towns, Lyman and Stanley.

***Solanum triflorum* Nutt.**, cut-leaf nightshade. Occasional in prairie dog towns, Lyman and Stanley*.

TYPHACEAE, the cattail family

***Typha angustifolia* L.**, narrow-leaf cattail. Common along stock ponds and drainages, Jones, Lyman*, and Stanley.

ULMACEAE, the elm family

***Ulmus pumila* L.**, Siberian elm. Lone tree on hill in Missouri Breaks (Road) Allotment, Stanley.

VERBENACEAE, vervain family

***Lippia cuneifolia* (Torr.) Stued.**, wedgeleaf fogfruit. Common in prairie dog towns, Jones and Stanley.

***Verbena bipinnatifida* Nutt.**, Dakota vervain. Common in prairie dog towns, Jones, Lyman*, and Stanley.

***Verbena stricta* Vent.**, hoary vervain. Occasional in uplands, Jones*, Lyman, and Stanley.

VIOLACEAE, the violet family

***Viola nuttallii* Pursh**, Nuttall's violet. Common in uplands, Jones, Lyman, and Stanley*.

ZANNICHELLIACEAE, the horned-pondweed family

Zannichellia palustris L., horned-pondweed. Occasional in stock ponds and wetlands, Lyman.

ZYGOPHYLLACEAE, the caltrop family

Tribulus terrestris L., goat-head. Occasional along roadside in West Bad River Allotment, Stanley.