THE EFFECTS OF TARGETED GRAZING OF YELLOW STARTHISTLE BY DOMESTIC

GOATS IN NORTHERN IDAHO

AND

AN EXAMINATION OF SEED SURVIVAL IN THE RUMINANT DIGESTIVE TRACT

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Abstract

Yellow starthistle (*Centaurea solstitialis* L.) is an invasive winter annual that is one of the most problematic and widespread plants in the western United States. Biological control has been shown to decrease seed production of yellow starthistle but not yet to the level where management is achieved. A variety of control measures (mainly chemical) have been successfully applied to manage infestations of yellow starthistle. However, yellow starthistle offers a particular challenge in infested canyon rangelands where accessibility limits options for control. Targeted grazing by domestic goats may provide an additional option for control of yellow starthistle at landscape scales.

A 3-year grazing study was initiated in 2006 to determine if late-season grazing by goats could provide an ecologically beneficial and effective tool for yellow starthistle control at the landscape scale. Twenty-four paired plots were established on 380 hectares of land managed by the United States Forest Service near White Bird, Idaho (lat 45°45'N, long 116°17'W). Each paired plot consisted of a fenced subplot to exclude grazing and a similar sized adjacent subplot that was grazed. Over1000 Boer goats managed by a grazing service provider grazed the study area during late summer and early fall each year. Density of yellow starthistle plants and seedheads was assessed before and after grazing of each plot. Canopy cover of grasses, forbs, and yellow starthistle was also examined.

After grazing treatment in every year, there were fewer yellow starthistle plants (P<0.001) and seedheads (P<0.001) in grazed areas compared to ungrazed controls. The effect of grazing on yellow starthistle canopy cover depended on the year it was examined. Cover of yellow starthistle did not differ between grazed and ungrazed subplots after grazing in 2006 (P=0.072), while grazing decreased yellow starthistle cover in 2007 and 2008 (P<0.001 for both years). Grazing by goats appeared to have little impact on canopy cover of grasses and other forbs with the exception of after grazing in 2007 when there was less forb cover in grazed areas compared to ungrazed areas (P<0.001).

Goats will graze yellow starthistle throughout most of its life cycle, and late-season grazing by goats may be especially effective for managing yellow starthistle. During late-season grazing, however, goats are consuming mature seedheads with viable seeds. A pen-feeding study was conducted to determine the passage rate and viability of yellow starthistle seeds after ingestion by goats. Five female goats were each fed 2000 yellow starthistle seeds. Feces were then collected from

each goat for eight days and examined for seeds. Recovered seeds were tested for viability. About 3% of ingested seeds were recovered, and about 3% of those recovered seeds were viable. Most seeds were passed in feces within the first three days after ingestion.

Keywords: *Centaurea solstitialis*, invasive plants, targeted grazing, seed passage, seed recovery, digestion

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Introduction

Invasive plants cause tremendous ecological and economic loss on rangelands (Duncan et al. 2004). Yellow starthistle (*Centaurea solstitialis* L.) is a particularly challenging invasive winter annual that is one of the most problematic and widespread invasive plants in the United States (DiTomaso 2000), infesting almost 6,000,000 hectares in the western states (Duncan et al. 2004). The only invasive plant species that infests more area is cheatgrass (*Bromus tectorum* L.; Duncan et al. 2004). Major infestations exist in California, Idaho, Washington, and Oregon. However, yellow starthistle has been increasing in abundance since 1970 (Maddox et al. 1985) and is spreading at an annual rate of 13 to 17% (Duncan et al. 2004).

Yellow starthistle can form thick stands that are avoided by livestock and wildlife (Thomsen et al. 1993; Hovde 2006). Yellow starthistle adds to fuel loads of wildfires (Thomsen et al. 1997) and degrades recreational value and biodiversity (Balciunas and Villegas 1999; Benefield et al. 1999). Yellow starthistle has the ability to impact community structure and function by displacing native and sometimes rare plant species, by altering wildlife populations, and by modifying fuel characteristics and soil moisture levels (Duncan et al. 2004). Yellow starthistle also invades grain fields, orchards, vineyards, cultivated crops, pastures, roadsides, and wastelands and can contaminate alfalfa, cereal grains, hay, and commercial seed (Maddox and Mayfield 1985).

The impact of all invasive plants to rangelands and pastures is estimated to be $6 billion annually (Pimentel et al. 2000; Pimentel et al. 2005). A recent study calculates that Idaho alone suffers about $8.2 million in direct losses relating to yellow starthistle's negative impacts on forage value for livestock, agricultural crops, watershed quality, and wildlife habitat in addition to secondary losses of $4.5 million (Juliá et al. 2007). Yellow starthistle is estimated to cost California ranchers about $17 million annually (Eagle et al. 2007).

A variety of control measures (mainly chemical) have successfully controlled infestations of yellow starthistle. However, yellow starthistle offers a particular challenge in Idaho where the plant presently infests vast areas of rugged, canyon rangelands that limit options for control. Targeted grazing by domestic goats may provide an additional option for control of yellow starthistle at landscape scales.

Yellow Starthistle Natural History and Growth

Yellow starthistle likely originated in the Middle East and became naturalized to the Mediterranean basin (Maddox and Mayfield 1985). Today, yellow starthistle is found in most temperate regions of the world (Maddox et al. 1985; Maddox and Mayfield 1985). Yellow starthistle was first introduced to the United States in California via contaminated alfalfa seed in the mid-1800s (Maddox and Mayfield 1985; Gerlach et al. 1998). The rapid spread of yellow starthistle coincided with extensive ranching, road-building, and suburban development (Gerlach et al. 1998). Yellow starthistle steadily spread after its introduction, and it now infests about 1.2 million hectares in Idaho (Wilson et al. 2003). Yellow starthistle in Idaho occurs predominantly along the banks and canyons of the Snake and Clearwater Rivers in the north central part of the state (Callihan et al. 1989). Yellow starthistle is often found on warm, south-facing slopes amid bluebunch wheatgrass/Idaho fescue and bluebunch wheatgrass/Sandberg bluegrass habitat types (Roché 1989) and can invade these grassland communities in the absence of disturbance (Roché et al. 1994).

Several factors contribute to yellow starthistle's capacity as an invasive plant. Tremendous seed production, flexible germination requirements, and efficient water use give yellow starthistle a competitive ability over many native plants. Yellow starthistle quickly recovers from the effects of defoliation, especially early in the season (Thomsen et al. 1993; Benefield et al. 1999), and human activities such as hiking, hunting, and agricultural practices aid its spread (Gerlach et al. 1998). Yellow starthistle furthermore has great genetic variability across populations within the United States (Sun 1997). This likely reflects the genetic plasticity of yellow starthistle and contributes to its ability to exist in a broad array of ecological conditions and its potential to become resistant to herbicides or adapted to biological control agents (Sun 1997).

Seed production

Yellow starthistle has vast seed production that allows it to rapidly build up its seedbank. Yellow starthistle can produce over 10,000 seeds/m^2 (DiTomaso et al. 1999b) with individual seedheads that can generate 65 to 83 seeds (Benefield et al. 2001). This high seedhead production capacity creates the potential for rapid population expansion. For example, three years of prescribed burning of yellow starthistle infestations in California reduced the yellow starthistle seedbank density from about 10,000 to 50 seeds/m^2 but once treatment was halted, seed counts returned to control levels within four years (Kyser and DiTomaso 2002). Even 91% control by herbicide application

would not be sufficient for long-term management (DiTomaso et al. 1999a). Self-thinning of yellow starthistle can also improve seed production (Sheley and Larson 1994).

Germination and growth conditions

Yellow starthistle is well-adapted to infest the semi-arid lands of western North America (Callihan et al. 1982). Yellow starthistle germinates in winter, spring, or early summer whenever moisture is available (Callihan et al. 1989; DiTomaso 1999a), but most germination occurs in the fall (Joley et al. 2003). In California, yellow starthistle can begin germination after as little as 11 mm of fall precipitation (Joley et al. 2003).

Moisture requirements and water use

Yellow starthistle requires little moisture to germinate although subsequent precipitation events affect seedling survival (Joley et al. 2003). Yellow starthistle seed production is higher in wet years than comparatively dry years (Thomsen et al. 1997). In a study in Washington, moist springs resulted in seed outputs of $21,600/m^2$ whereas a dry spring produced only $5,200$ seeds/m^2 even though plant densities remained constant across years (Sheley and Larson 1994). Rain events can significantly affect outcomes of treatment for yellow starthistle control (Thomsen et al. 1989; Thomsen et al. 1997). For example, effectiveness of chemical control was reduced when yellow starthistle plants exhibited signs of drought-stress (DiTomaso et al. 1999a).

Yellow starthistle develops a deep tap root that extracts moisture lower in the soil profile than either annual or perennial grasses (Enloe et al. 2004), allowing for more efficient moisture use. Yellow starthistle utilizes more moisture than other annual or perennial species resulting in simulated drought conditions for neighboring vegetation (DiTomaso et al. 2000**,** DiTomaso et al. 2003). Plants germinating in late October can have roots over 1.2 meters deep by mid-March (Roché 1989). Heavily infested control plots had significantly greater soil moisture depletion than similar plots treated with herbicides (DiTomaso et al. 2000). This suggests that controlling yellow starthistle at both the site and landscape scales may increase soil moisture levels and may impact plant community dynamics (Enloe et al. 2004).

Allelopathy

Yellow starthistle has long been speculated to possess allelopathic properties (see Fletcher and Renney 1963, Maddox and Mayfield 1985). Other species in the Asteraceae family (i.e., Russian knapweed, *Acroptilon repens* {L.} DC.; spotted knapweed, *Centaurea stoebe* L.; diffuse knapweed,

Centaurea diffusa Lam.) have been shown to be allelopathic (Fletcher and Renney 1963), but recent research found no indication that yellow starthistle suppressed other plants through release of phytochemicals from root exudates (Qin et al. 2007). While this study focused on the roots, other studies showed the shoot tissues of other Asteraceae species have allelopathic properties (Hierro and Callaway 2003).

Competition

Most of the suppression that occurs from yellow starthistle is likely a result of its superior ability to capture resources for growth. Yellow starthistle may inhibit both germination and seedling development of neighboring plants by reducing sunlight penetration in dense stands (Callihan et al. 1989). Dense stands of yellow starthistle may experience self-thinning (Sheley and Larson 1994) but can persist at high population densities (Callihan et al. 1989).

Yellow starthistle apparently avoids competition with native and introduced perennial grasses by germinating late in the fall after grasses have become dormant and early in the spring before perennial grasses initiate growth (Callihan et al. 1989). This gives a competitive advantage to yellow starthistle in native grasslands and during rehabilitation efforts. As an introduced species, yellow starthistle lacks its native predators and pathogens in North America, and this further enhances its competitive edge against native species (DiTomaso et al. 2003).

Seed dispersal

Wind is not considered a major vector for seed dispersal (Roché 1989; Roché 1991) although yellow starthistle produces both plumed and unplumed seeds. Unplumed seeds remain in the seedhead while plumed seeds are extruded at maturity (Roché 1989). Plumed seeds are not usually windborne because the pappus is too small in relation to seed size (Roché 1991; Callihan et al. 1993). Most plumed seeds fall within a meter of the parent plant (Roché 1991), but gusts of wind can disperse plumed seeds downwind as far as 9.8 meters (Roché 1989). Wildlife tend to avoid thick stands of yellow starthistle (Thomsen et al. 1993) and most seeds consumed by birds are not germinable following digestion (Roché 1991). Spread of yellow starthistle through natural processes (i.e., wind, wildlife) thus actually appears to be quite slow. Preliminary studies have shown that post-dispersal seed predation by insects occurs in Idaho canyonlands (L. Wilson, pers. comm.).

The pappus of plumed seeds can allow attachment to clothing, animal fur, or feathers (Callihan et al. 1993). Recreational and agricultural activities facilitate dispersion (Gerlach et al.

1998). The few seeds that are widely dispersed by whirlwinds, birds, other animals, or vehicles can initiate new infestations by the large numbers of seeds that will later be generated from those few establishing seeds (Roché 1991; DiTomaso et al. 1999a; Kyser and DiTomaso 2002; Joley et al. 2003). Scattered satellite populations can result in tremendous yellow starthistle seed production (Gerlach et al. 1998).

Seedbank longevity

Several studies have demonstrated that yellow starthistle seedbanks can be rapidly depleted (Joley et al. 1992; Sheley and Larson 1994; DiTomaso et al. 1999b; Benefield et al. 2001; Joley et al. 2003). For example, when recruitment was prevented on a site in California, the yellow starthistle seedbank was reduced by more than 80% the first year and by more than 96% the third year (Joley et al. 1992). Greater than 90% of yellow starthistle seeds are germinable after dispersal (Benefield et al. 2001), and seeds germinate as soon as sufficient moisture is present (Gerlach et al. 1998). Near complete loss of germinable seed can be attributed to germination during burial (Joley et al. 1992). The seedbank may be significantly reduced by microbial degradation and invertebrate predation in California conditions (Joley et al. 1992; Benefield et al. 2001). Control thus depends on preventing yellow starthistle seed recruitment or replenishment (Benefield et al. 2001).

Seedbank characteristics and conditions in Idaho differ from those in California. For example, in Idaho, plumed seeds are more viable than unplumed seeds (Callihan et al. 1993) while no differences in viability were found between the two seed types in California conditions (Joley et al. 1992). Unplumed and plumed yellow starthistle seeds in Idaho have average longevities of six and ten years, respectively (Callihan et al. 1993). Joley and colleagues (1992) estimated that yellow starthistle seeds in California can remain viable beyond three years. Seeds collected from California, Idaho, Oregon, and Washington exhibit a broad variation in ecology (plant and rosette size, growth dates, flowering rate, seed production) that is independent of rain, elevation, or latitude (Roché 1989).

Control Methods

Biological control agents, herbicide use, mowing, and prescribed burning have all been used with variable success in controlling yellow starthistle, but each of these options is most effective for a given scenario and each also has limitations. It needs to be acknowledged that yellow starthistle is not going to be eradicated in the United States. The most appropriate treatment for yellow starthistle may thus follow an Integrated Pest Management (IPM) approach in which multiple treatment methods

coupled with knowledge of yellow starthistle's life cycle are used to assert selection pressure on yellow starthistle in a way that is both economical and ecological. The components of an IPM are conditional upon land management objectives. Is the objective to add forage value to a yellow starthistle-infested landscape or to restore an infested site by decreasing yellow starthistle abundance and increasing native species abundance? Is the goal to control or to manage yellow starthistle? Control refers to reducing or eliminating weed populations while managing weed populations emphasizes minimizing the competitive ability of weeds with desirable vegetation (Buhler et al. 2000).

A window of opportunity exists when yellow starthistle is sensitive to treatment. Several studies demonstrate that timing and frequency of treatment application to invasive plants is the critical factor that determines the success of any particular method of control (Thomsen et al. 1989; Thomsen et al. 1993; Lym et al. 1997; Olson et al. 1997a; Thomsen et al. 1997; Benefield et al. 1999; DiTomaso et al. 1999b; Benefield et al. 2001; Hovde 2006). For example, Benefield and colleagues (1999) showed that mowing at the late-flowering stage of yellow starthistle phenology resulted in the greatest reduction of yellow starthistle seedheads and biomass compared to mowing at earlier stages, and Thomsen and colleagues (1993) found that targeted livestock grazing had to be applied at least twice within a season in order to prevent re-growth of yellow starthistle.

Biological control

Biological control is a sustained method to reduce yellow starthistle seed production, especially in the canyon grasslands of the inland Northwest. However, biological control approaches are not sufficient in themselves to contain yellow starthistle (DiTomaso et al. 2000). Five insect species and one rust species have been released as approved biological control agents for yellow starthistle in the United States since 1984 (Balciunas and Villegas 1999, Woods and Villegas 2004)**.** These include three weevils: *Bangasternus orientalis*, *Eustenopus villosus*, and *Larinus curtus*; and two flies: *Chaetorellia australis* and *Urophora sirunaseva*. *Urophora jaculata* was also released but never established viable populations. *Puccinia jaceae* var. *solstitialis*, commonly called yellow starthistle rust, was released in California in 2003. This rust is highly specific to yellow starthistle and affects it by attacking the leaves and stems of yellow starthistle.

Another fly species, *Chaetorellia succinea*, was accidentally introduced in the late 1980s and was predicted to be a viable biological control agent of yellow starthistle in California (Balciunas and

Villegas 1999). *Chaetorellia succinea* is presently widespread throughout California and has spread to the Northwest (Balciunas and Villegas 2007). *Chaetorellia succinea* destroys the seeds of yellow starthistle, but recent work found that it will also feed on two other introduced *Centaurea* species (Maltese starthistle, *Centaurea melitensis* L.; Sicilian starthistle, *Centaurea sulphurea* Willd.) and the native American starthistle (*Centaurea americana* Nutt.; Balciunas and Villegas 2007). American starthistle, found in the southwestern United States, could be at risk if *Chaetorellia succinea* also spreads to that region (Balciunas and Villegas 2007). *Chaetorellia succinea* likely displaces *Chaetorellia australis* where the two species occur together (L. Wilson, pers. comm.). Competitive interactions may also exist between *Chaetorellia spp.* and *Eustenopus villosus* because both genera attack yellow starthistle seedheads (Tonkel and Piper 2009).

At a study site in California, the attack on yellow starthistle seedheads by one or more of the biological control insect species increased from 22 to 83% over four years while seed production decreased from 13,839 to 3,802 seeds/m^2, and seedling density dropped from 897 to 234 seedlings/m^2 (Pitcairn et al. 2000). Despite these declines, no decrease was observed in adult plant densities for that year in this study; however, the authors predicted a decline in adult plant density in future years if the decline in seed production and seedling recruitment continue while biological control agent densities continue to increase.

In Idaho, *Eustenopus villosus* is the most common biological control agent and has had the greatest impact on yellow starthistle (Wallace et al. 2008). The life cycle of *Eustenopus villosus* coincides with that of yellow starthistle so that the insects are active while seedheads are developing (Connett et al. 2001). Feeding by adult *Eustenopus villosus* damaged up to 93% of flower heads per yellow starthistle plant (Wallace et al. 2008). The damage resulting from adult and larval feeding and oviposition can reduce seed production by 26 to 70% compared to undamaged seedheads (Wallace 2005). Prescribed grazing and biological control insects, specifically *Eustenopus villosus*, are potentially compatible for an integrated control plan as attack rates were equal between grazing treatments and the ungrazed control in an Idaho study (Wallace et al. 2008).

Herbicide

Chemical control is the most commonly utilized method of treating yellow starthistle in California (Eagle et al. 2007) and likely the rest of the United States. Herbicide can be used to control or eradicate small patches and is probably the most effective method for new infestations. However,

few nonselective pre-emergence herbicides that can effectively control yellow starthistle can be used in rangelands and pastures because of the grasses present (DiTomaso et al. 1999a). Chlorsulfuron combined with 2,4-D or triclopyr has resulted in 91% control of yellow starthistle but this rate is not sufficient for long-term management due to the high seed production of yellow starthistle (DiTomaso et a. 1999a). Clopyralid, a selective pre- and post-emergence herbicide, is safe for grasses and biological controls and has no grazing restrictions and has provided 100% control consistently throughout the entire season when applied at low rates (DiTomaso et al. 1999a). One concern with clopyralid use is that it may lead to selection of resistant biotypes of yellow starthistle or even to selection of other undesirable species such as cheatgrass, medusahead (*Taeniatherum caput-medusae* {L.} Nevski)*,* ripgut brome (*Bromus diandrus* Roth)*,* or barbed goatgrass (*Aegilops triuncialis* L.; DiTomaso et al. 1999a; DiTomaso et al. 2000). The widespread use of picloram, clopyralid, triclopyr, dicamba, and 2,4-D on yellow starthistle infestations has resulted in developed herbicidal resistance in some yellow starthistle populations (Fuerst et al. 1996; Sterling et al. 2001). Aminopyralid, a new pyridine carboxylic acid herbicide, effectively treats Russian knapweed (*Acroptilon repens* {L.} DC.) and Canada thistle (*Cirsium arvense* {L.} Scop.) at lower use levels than other herbicides such as clopyralid (Enloe et al. 2008; Bekun et al. 2009). Preliminary research shows that aminopyralid is also highly effective on yellow starthistle (T. Prather, pers. comm.).

Mowing

With proper timing, mowing can successfully control yellow starthistle (Thomsen et al. 1997; Benefield et al. 1999). Yellow starthistle often recovers rapidly with re-growth and flowering when mowed at an early growth stage, and viable seedheads can still be produced even when plants are mowed at late flowering stages (Benefield et al. 1999). Biomass is greatly reduced when plants are mowed twice in the spiny stage and once or twice in the early flowering stage (Benefield et al. 1999). Mowing after the formation of viable seeds can hinder efforts to reduce yellow starthistle plant densities because viable seeds will still reach the seedbank (Thomsen et al. 1997). Benefield and colleagues (1999) determined that the most effective time to decrease seedhead numbers was to mow in the early flowering stage, but these results were not always consistent. Effects of mowing vary from season to season in relation to precipitation (Thomsen et al. 1997).

Desirable perennial grasses gain a competitive advantage when mowing reduces or prevents seed production and carbohydrate reserves of yellow starthistle (DiTomaso 2000). Mowing is

commonly used in California (Thomsen et al. 1997), but this method of control is limited by topography and the accessibility of the mowing equipment to infested areas.

Prescribed burning

Control of yellow starthistle can be accomplished through prescribed fire. Three years of prescribed burning of yellow starthistle resulted in a 99% decrease in the seedbank in a California study (DiTomaso et al. 1999b). Treatment should occur after desirable vegetation completes its reproductive cycle but before yellow starthistle begins seed production (DiTomaso et al. 1999). The use of fire can create wildfire and health risks and requires appropriate weather conditions and sufficient fuel loads (DiTomaso et al. 2000). In Idaho, yellow starthistle produces seeds when wildfire hazard is high. Additionally, application of prescribed burning to treat yellow starthistle across the landscape in the West may be limited because niches formerly occupied by yellow starthistle will most likely be filled by cheatgrass at sites where cheatgrass is present. Cheatgrass is a highly invasive annual plant that is often co-dominant with yellow starthistle. The increased nitrogen availability that occurs after fire increases cheatgrass's ability to compete with perennial grasses (Pellant 1996).

Targeted grazing

Yellow starthistle readily recovers from defoliation throughout most of its life cycle and can recover faster than other plants species even in drought (Thomsen et al. 1993). Early season defoliation of yellow starthistle and other species in the community results in greater plant density of yellow starthistle by the end of the season (Thomsen et al. 1993; Hovde 2006).

Livestock readily graze yellow starthistle in certain stages of its growth (Thomsen et al. 1993; Hovde 2006), but most livestock avoid grazing yellow starthistle that has fully developed spines (Thomsen et al. 1993). In Thomsen et al. (1993), cattle, sheep, and goats grazed yellow starthistle-infested paddocks in the rosette, bolting, and pre-spiny stage. Goats grazed upright bolting plants while passing over rosettes (Thomsen et al. 1993), and this likely relates to their characteristic browsing behavior (Hofmann 1989). Goats also avoided plants with spines (Thomsen et al. 1993). This study suggests that familiarity with yellow starthistle may be an important factor associated with targeted grazing.

Thomsen and colleagues (1993) found that effectiveness of grazing in their study was more dependent on timing and repeated grazing than the type of animal used because cattle, sheep, and goats each grazed yellow starthistle; however, yellow starthistle use differed among the three species

(Thomsen et al. 1993). Single grazing treatments by cattle and sheep to yellow starthistle in the rosette and bolting stages resulted in increased yellow starthistle density and seed production (Hovde 2006). Thomsen and colleagues (1993) experienced similar results with initial grazing treatments, but yellow starthistle density and cover was reduced when grazing treatments were applied multiple times in a growing season. Management of yellow starthistle will be accomplished only if grazing is repeated at the most vulnerable growth stage of yellow starthistle (Thomsen et al. 1993).

Targeted Grazing Considerations

Targeted grazing is defined as the application of a specific kind of livestock at a specified season, duration, and intensity to accomplish defined management goals (Frost and Launchbaugh 2003). Targeted grazing becomes a viable option for invasive plant management when plants have forage value, when livestock will selectively consume the target plants, and when livestock utilization results in suppressed or reduced growth of target plants with an acceptable level of impact on desired vegetation.

Consideration must be given to the type of vegetation being managed and to the foraging abilities of the livestock used for a targeted grazing project. Those foraging abilities depend on the species of livestock and the foraging behavior of the individual. Foraging behavior develops in response to food preferences, and food preferences result from post-ingestive feedback, experience, and social models (Provenza and Launchbaugh 1999).

The use of livestock for target grazing invasive plants is a worldwide concept and has been applied in many weed scenarios (Popay and Field 1996). Targeted grazing finds many different uses in pastures, croplands, orchards, and rangelands. Advantages of using livestock to manage plants include decreased chemical residue, more effective control than herbicides in some cases, improved pasture and range quality, sustainable control, and decreased impact on non-target species. Additional benefits relate to animal production, including conversion of plants to animal protein and animal weight gains (Popay and Field 1996). Disadvantages of using livestock include spread of weed seed by feces or hair, damage to non-target species, and negative impact on soil and cover (Popay and Field 1996).

Targeted grazing may be valuable in the control of yellow starthistle. Goats specifically may be the most appropriate species of livestock in a grazing project that targets yellow starthistle because of their dietary habits. Thomsen and colleagues (1993) conducted early season grazing by

inexperienced dairy goats in small infested paddocks, demonstrating that goats will consume yellow starthistle. Goats have since become quite popular for grazing small infestations of yellow starthistle (DiTomaso et al. 2000) though they also offer application for landscape-scale infestations.

Grazing of yellow starthistle can cause disturbance, reduce intraspecific competition, and increase sunlight penetration by removing canopy cover. Removing litter can increase germination and seedling survival of yellow starthistle (Roché and Thill 2001; Gelbard and Harrison 2005), and yellow starthistle's ruderal traits of rapid growth and high seed production suggest that disturbance should favor yellow starthistle germination and growth (Roché and Thill 2001). At the same time, seed recruitment is potentially being prevented by the ingestion of seedheads. We might thus anticipate that increased germination from the seedbank coupled with decreased seed recruitment would result in more rapid depletion of the yellow starthistle seedbank. Yellow starthistle responses following initial treatment are often unnoticeable because of the established seedbank (Joley et al. 1992; Thomsen et al. 1997). Several years of treatment are consequently required before the yellow starthistle seedbank is significantly reduced.

Effects of Ruminant Digestion on Seed Survival

Targeted livestock grazing with goats, sheep or cattle as a vegetation management tool has been gaining popularity and has application for the control of many weedy species. For example, goat grazing can reduce yellow starthistle plant densities (see Chapter 2) and sheep grazing can reduce spotted knapweed plant densities (Olson et al. 1997a). Goat grazing can be a viable tool for managing kudzu (*Pueraria* DC.) and multiflora rose (*Rosa multiflora* Thunb.; Luginbuhl et al. 1996). Sheep have been used successfully to control leafy spurge (*Euphorbia esula* L.; Lacey et al. 1984). One concern that arises with targeted grazing is the potential spread of weed seed by attachment to hair and wool or passage in feces and thus aggravating the very problem that targeted grazing is trying to address. Knowledge of seed survival and dispersal can be used to guide weed management decisions.

Seed attachment to hair and wool is likely an important mode of dispersal for some plant species, but that topic has not been greatly explored. Seeds may be picked up in sheep fleeces, but wool character causes seeds to become easily embedded so that seeds will not likely fall out except perhaps during shearing (Olson et al. 1997b).

Research and field observations clearly show that livestock do pass viable seeds in feces. However, this same research demonstrates that passage through the digestive tract significantly

reduces the germination and viability of the seeds of many invasive plant species (e.g., Lehrer and Tisdale 1956; Thill et al. 1986; Blackshaw and Rode 1991; Lacey et al. 1992; Wallander et al. 1995; Olson et al. 1997b; Cosyns et al. 2005). A relatively small body of research has explored the fate of seeds traveling through the digestive systems of herbivores and omnivores, and research focusing specifically on livestock and dry-seeded plant species is scarce. Traveset (1998) provides a review of how vertebrate ingestion of seeds of nearly 200 plant species affects germination. The majority of studies in that review examined fleshy-fruited species in the tropics, and only two studies involved domestic ruminants. It can be anticipated that digestion will destroy a minimum of half of ingested seeds (Janzen 1984). Surviving seeds must then have sufficient seedling vigor to establish in feces, and feces must also be deposited in such a way that seedling establishment is not inhibited (Gökbulak and Call 2004).

Passage of an ingested seed through a ruminant digestive tract

The purpose of digestion is to convert complex plant material into a usable, absorbable form. Thus, it is not surprising that passage through a ruminant's digestive tract and subsequent excretion is a costly mechanism for seed dispersal. Loss of viability occurs throughout most of the digestive tract due to both physical and chemical processes (Figure 1.1). The loss of viability can be a result of either total destruction and absorption by digestive processes (i.e., the seed has nutritive value) or sufficient damage to cause seed death (i.e., the seed is excreted but not viable). The possibility also exists at any point along the way for a seed to become caught in a fold of tissue, thus increasing retention time. Increased retention time can result in greater loss of seed viability (e.g., Thill et al. 1986; Simao Neto et al. 1987; Blackshaw and Rode 1991).

The digestion process initiates with prehension where ruminants use lips, teeth, and tongue to move food (and seeds) into their mouths. Following prehension, ruminants generally only chew enough to get the proper mixture of food and saliva to form a bolus and facilitate swallowing (Church 1976). Damage to the seed coats can occur during this initial, although cursory, mastication. Seeds are swallowed and enter the mixing vat of the reticulorumen where they are subjected to microbial fermentation and ruminal contractions. Microbial fermentation has been shown to kill seeds (Blackshaw and Rode 1991; Carpinelli et al. 2005).

Ingesta in the rumen stratify into layers after initial mastication and swallowing. Size and specific gravity of the seeds likely influence which layer ingested seeds sort into. Small, dense

particles can settle into the lower, liquid layer of the reticulorumen and get flushed through the reticulo-omasal orifice into the omasum (Van Soest 1982). Katoh and colleagues (1988) found that the percentage of excreted particles increased and the percentage of ruminated particles decreased as specific gravity of those particles increased. This results in particles with higher specific gravity having faster passage rates (Katoh et al. 1988). In a seed-feeding study, retention time of seeds increased as specific gravity decreased (Gardener et al. 1993a).

The reticulo-omasal orifice prevents passage of large particles and triggers rumination (Welch and Hooper 1988). Consequently, "small" seeds of a range of sizes can still pass through the orifice and avoid rumination. However, a 1-mm particle has a 10% chance of passing into the omasum without being ruminated (Smith 1968 as cited by Van Soest 1982).

Larger or lighter particles may settle into the middle stratum of ingesta, which is mostly comprised of recently ingested forage (Van Soest 1982). Rumination occurs when a bolus from this middle layer is regurgitated to be re-masticated. This provides another opportunity for seeds to be damaged by mastication. The bolus is swallowed and enters the reticulorumen where seeds are again subjected to microbial fermentation. Liquid digesta are flushed through the reticulo-omasal orifice into the omasum where water is absorbed. At this point, retention time of seeds could be increased if they were to get caught in the omasal leaves. Digesta in the omasum pass into the abomasum where gastric digestion occurs. Seeds are exposed to an acidic environment (pH 2.5) in the abomasum due to the presence of hydrochloric acid. Enzymatic secretions by the pancreas and the liver raise the acidic pH of digesta as it enters the small intestine where nutrients are digested and absorbed. Digesta that have escaped digestion are subjected once more to microbial fermentation in the large intestine (cecum, colon, and rectum). Water is absorbed, and the remaining matter is finally excreted from the rectum as feces.

Factors affecting the passage and survival of an ingested seed

Digestion by ruminants generally destroys some, if not a substantial portion, of ingested seeds. The effects of digestion on seeds that get passed in feces can vary. To interpret results of such studies, it is important to understand the context of each study (e.g., pen feeding versus *in sacco* studies) and to note how data were collected (e.g., percent of recovered seeds that tested viable versus percent of fed seeds that germinated from feces). Digestion can cause some seeds to lose viability and others to increase germination percentage or rate. Still other passed seeds have no change in

germinability. These differences of effects of digestion on seeds may depend on the characteristics of the seed ingested or the herbivore doing the ingesting.

Published research reveals that no single factor or combination of factors can consistently predict the effects of digestion on seeds across plant species. For example, Russi and colleagues (1992) found that seed recovery from sheep feces was inversely related to seed size while Manzano and colleagues (2005) found no correlation between seed size and seed recovery. Lehrer and Tisdale (1956) found that six range plant species had similar germination percentages following passage through sheep even though seeds had very different sizes, shapes, and seed coats. The interactions between plant and animal attributes likely determine the survivability of an ingested seed.

Plant attributes

Seed size. Effects of size and shape on a seed's ability to survive digestion are ambiguous although most research reports that small seeds have greater survivability (e.g., Russi et al. 1992; Edward et al. 1998; Pakeman et al. 2002; Mouissie et al. 2005). Small seeds may be more likely to survive digestion than large seeds because small seeds are more likely to escape mastication (Russi et al. 1992; Gökbulak 2006). Seed size limits seed coat thickness so that small seeds are more likely to be affected by digestive fluids (Manzano et al. 2005). In one seed recovery study, the largest-seeded (arrowleaf balsamroot; *Balsamorhiza sagittata* {Pursh} Nutt.) and the smallest-seeded species (scarlet globemallow; *Sphaeralcea coccinea* {Nutt.} Rydb.) both had low recovery from bison feces relative to the intermediate-sized seeds of Indian ricegrass (*Achnatherum hymenoides* {Roem. & Schult.} Barkworth; Gökbulak 2002). The low recovery of scarlet globemallow was attributed to a soft seed coat. Intermediate-sized shrub seeds common to central Spain also had greater recovery than small- or large-seeded shrubs from sheep feces (Manzano et al. 2005).

Seed size may affect seed passage rate. Smaller seeds typically passed faster than larger seeds in research by Burton and Andrews (1948) and Blackshaw and Rode (1991), but this relationship was not observed by Simao Neto et al. (1987) or Gökbulak (2006). Seed size may not affect passage rate when seeds are smaller than a threshold size (Russi et al. 1992). Particles below a certain size experience less resistance to passage as they can easily pass through the reticulo-rumen orifice (Poppi et al. 1985). Greatest differences in passage rates relative to seed sizes might then be observed when comparing seeds that fall on different sides of this threshold size.

Seed shape. Seeds that can survive digestion by ruminants vary in shape, and consistent relationship between seed shape and recovery or germination success has seldom been observed (e.g., sheep, Lehrer and Tisdale 1956; fallow deer, Mouissie et al. 2005). Round seeds had faster passage and greater recovery from bison (Gökbulak 2002). In another study, the elongated seeds of graminoid species had greater germination after being passed by rabbits, cattle, sheep, horses, and donkeys (Cosyns et al. 2005).

Seed coats and dormancy. Some seeds that have hard seed coats may be more likely to survive digestion (Atkeson et al. 1934; Burton and Andrews 1948; Simao Neto et al. 1987; Blackshaw and Rode 1991; Russi et al. 1992; Gardener et al. 1993a; Doucette et al. 2001; Gökbulak 2002). Soft seed coats may allow seeds to imbibe in the gut and thus rupture (Gardener et al. 1993a; Edward et al. 1998). Lambsquarters (*Chenopodium album* L.) and redroot amaranth (*Amaranthus retroflexus* L.) maintained relatively high viability after twenty-four hours in the rumen of fistulated cows possibly due to hard seed coats (Blackshaw and Rode 1991). Mediterranean legumes in particular are noted for having seed coats of varying hardness within a single species, and hard seeds survive digestion more frequently than soft seeds (Simao Neto et al. 1987; Russi et al. 1992). Simao Neto and colleagues (1987) found viability greater than 95% in hard-seeded legumes recovered from the feces of cattle, sheep, and goats; however, digestion still reduced total viability of the four legume species fed to livestock. Scarlet globemallow, although small-seeded, has a soft seed coat and thus had low recovery from bison feces (Gökbulak 2002). This relationship between seed coat characteristics and seed survival during digestion is not found in all species. Soft-seeded Spanish lavender (*Lavandula stoechas* ssp. *pedunculata*) had an estimated retrieval of about 16% of fed seeds which compared to 12 to 23% recovery of four other hard-seeded shrub species common in Spain (Manzano et al. 2005).

Digestion may enhance germination of some hard-seeded species by scarifying seed coats or breaking dormancy (Lehrer and Tisdale 1956; Blackshaw and Rode 1991). Seeds of field pennycress (*Thlaspi arvense* L.) that were placed directly in the rumens of fistulated cows for twenty-four hours had 98% germination compared to 54% germination of control seeds in one trial, but a separate experiment showed that field pennycress rapidly lost viability with continued time in the rumen (Blackshaw and Rode 1991). Passage through sheep greatly improved the germination percentage of legume seeds by two- to six-fold of seeds recovered twenty-four and forty-eight hours after ingestion

(Russi et al. 1992). Digestion by sheep improved germination for *Halimium umbellatum* ssp. *viscosum*, a shrub found in central Spain (Manzano et al. 2005).

Digestion can break dormancy of some species and in others seems to have little effect. Several tropical grass species noted for high dormancy had greater germination after being placed directly in the rumens of fistulated cattle for up to forty-eight hours (Gardener et al. 1993b). In contrast, Gökbulak (2002) found that passage through bison did not break dormancy of Indian ricegrass, needle-and-thread grass (*Hesperostipa comata* {Trin. & Rupr.} Barkworth), arrowleaf balsamroot, or scarlet globemallow. Seeds can imbibe in the gut and then fracture apart; this occurrence becomes more likely as time in the gut increases (Gardener et al. 1993a). Seeds can even germinate *in vivo* as germinated seeds have been recovered from fresh feces (Doucette et al. 2001).

Other seed factors. Physical characteristics of seeds (i.e., size, shape, and seed coat) are not reliable indicators of seed response to ruminant digestion. A plant species' longevity index, or the ability of a species to have a persistent seedbank (Thompson et al. 1998), may have some merit for predicting seed survival. Two recent studies that examined the survivability of a wide range of plant species with differing traits reported that seeds of species with a higher longevity index tended to be more likely to germinate after ingestion and passage by ruminants, rabbits, horses, and donkeys than those with a lower longevity index (Pakeman et al. 2002; Cosyns et al. 2005). Pakeman and colleagues (2002) further reported that species that had both low mass and a high longevity index were more likely to germinate in feces while other plant traits like life history, life form, canopy structure and height, agency of dispersal, and regenerative strategies had relatively low correlation with seed germination in feces.

Animal attributes

Published research is relatively consistent regarding how the species of ruminant ingesting seeds affects seed passage when examining domestic livestock, (i.e., cattle, sheep, goats). Cattle in general pass more viable seed than either sheep or goats (Thill et al. 1986; Simao Neto et al. 1987; Shayo and Uden 1998; Razanamandranto et al. 2004; Cosyns et al. 2005). The difference of seed recovery between small ruminants (sheep and goats) and cattle may be because sheep and goats more thoroughly masticate their food than cattle (Church 1976; Thill et al. 1986; Cosyns et al. 2005). The relatively larger tongue, teeth, and mouth of a bovine likely increase the chance for seeds to escape damage from mastication. Differences in digestive tracts may also affect this difference between

species. Razanamandranto and colleagues (2004) hypothesized that passage through a smaller digestive tract would increase the chance of seeds being in contact with the gut wall and subsequently increase damage due to abrasion or digestion.

Cosyns and colleagues (2005) found that passage rates between the three classes of livestock were similar, but Razanamandranto and colleagues (2004) reported that seeds were passed more quickly by sheep than cattle. Leafy spurge seeds were recovered from goats within five days versus nine days from sheep (Lacey et al. 1992). Viable spotted knapweed seeds were ingested by sheep were recovered within seven days while viable spotted knapweed seeds were recovered from every day of the 10-day period that feces were collected from mule deer (Wallander et al. 1995). Despite these differences, most seeds are passed within the first four days after ingestion (cattle, Burton and Andrews 1948; sheep, Piggin 1978; cattle, sheep, goats; Simao Neto et al. 1987; sheep, Lacey et al. 1992; bison, Gökbulak 2002).

Seed germinability after ingestion can be affected by retention time (Janzen 1984). Germination percentages of seeds of tropical pasture species fed to cattle, sheep, and goats tended to decrease the longer the seeds resided in the digestive tract (Simao Neto et al. 1987). However, long retention times have improved germination of some legume seeds (Russi et al. 1992). Gökbulak (2006) found that germinability of bluebunch wheatgrass (*Pseudoroegneria spicata* {Pursh} A. Löve) decreased as time spent in the digestive tract increased while germinability of Sandberg bluegrass (*Poa secunda* J. Presl) remained constant or increased over time, but digestion reduced seed viability of both species compared to unfed seeds.

The type of diet seeds are ingested with may affect the microbial population and ruminating time; ruminating periods are longer for high-grain diets than high-forage diets (Blackshaw and Rode 1991). Blackshaw and Rode (1991) predicted that 68 to 71% of wild oat and cheatgrass seed would remain viable with a high-forage diet compared to an estimated 38 to 41% viability in a high-grain diet.

Seeds germinating in feces

Seeds can germinate directly from feces, but different feces types could influence germination and establishment of seeds. Dungpats of cattle can create a safe site for seedling germination while this would likely not occur in fecal pellets (Wicklow and Zak 1983). Seedling vigor contributes to the ability of a species to germinate in dungpats (Gökbulak and Call 2004).

Environmental conditions are important for seeds germinating in cattle dungpats because a crust layer can rapidly develop and inhibit seedling emergence (Akbar et al. 1995). Furthermore, seedlings may not survive in dungpats that are located on dry soil (Akbar et al. 1995). Seedling emergence of perennial grass species most often occurred in dungpats less than 2 cm thick or on the periphery of dungpats (Akbar et al. 1995; Gökbulak and Call 2004). Even if conditions within a dungpat or fecal pellet are not ideal for germination, seeds can remain viable within feces.

Implications of seed survival

Solid guiding principles of seed survival after ruminant ingestion are lacking. However, at best, fewer than half of ingested seeds survive digestion to germinate and establish (Janzen 1984). If the hard-seeded legumes that have often been studied in the Mediterranean region are excluded, substantially less than half of ingested seeds tend to survive. Still, attempts to predict the survivability of seeds through the digestive tract of a ruminant have met with limited success. It may then be wise to test the passage of ingested seeds of all species that are of major ecological importance, such as invasive plants or plants that are valuable for revegetation purposes. This knowledge will help determine the chance of plant spread through feces and is important from a livestock management standpoint.

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Summary

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The economic and ecological impacts of yellow starthistle in addition to its ability to invade and then dominate in both disturbed and undisturbed sites should make management and control of yellow starthistle a priority. Yellow starthistle has a persistent seedbank; consequently, equally persistent efforts will be required for suppressing yellow starthistle. Management goals for yellow starthistle infestations must take into consideration site conditions. For instance, after sixty-four years, natural revegetation of a disturbed site in southeastern Washington resulted in a predominantly perennial grass community on the north-facing aspect of the site while the south-facing aspect remained dominated by cheatgrass and yellow starthistle (Dillon 1967). This observation could have important implications for management of yellow starthistle; restoration of natural communities may be more feasible where yellow starthistle is limited by ecological conditions. Many control methods can successfully reduce or contain yellow starthistle. The most practical control methods for yellow starthistle will be site-specific and will require an integration of methods. When targeted grazing is used as a management tool, consideration must be given to viable seeds that may be ingested and passed in feces.

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Figure 1.1. A ruminant digestive tract. Ingested seeds can be subjected to either physical or chemical damage throughout most of the tract. Seeds may also become caught in folds of tissue and retained.

Chapter 2:   Targeted Grazing of Yellow Starthistle by Domestic Goats in Northern Idaho

Introduction

Invasive plants cause tremendous ecological and economic loss on rangelands (Duncan et al. 2004). Yellow starthistle (*Centaurea solstitialis* L.) is a particularly challenging invasive winter annual that is one of the most problematic and widespread plants in the United States (DiTomaso 2000), infesting almost 6,000,000 hectares in the western states (Duncan et al. 2004). The only invasive plant species that infests more area is cheatgrass (*Bromus tectorum* L.; Duncan et al. 2004). Major infestations exist in California, Idaho, Oregon, and Washington. Yellow starthistle has been increasing in abundance since 1970 (Maddox et al. 1985) and is spreading at an annual rate of 13 to 17% (Duncan et al. 2004). Yellow starthistle was first introduced to the United States in California via contaminated alfalfa seed in the mid1800s (Maddox and Mayfield 1985; Gerlach et al. 1998). The rapid spread of yellow starthistle paralleled the expansion of ranching, road-building, and suburban development (Gerlach et al. 1998).

Yellow starthistle can form dense, mono-specific stands that are avoided by livestock and wildlife (Thomsen et al. 1993). Yellow starthistle adds to fuel loads of wildfires (Thomsen et al. 1997) and degrades recreational value and biodiversity (Balciunas and Villegas 1999; Benefield et al. 1999). Yellow starthistle has the ability to impact community structure and function by displacing native and sometimes rare plant species, by altering wildlife populations, and by modifying fuel characteristics and soil moisture levels (Duncan et al. 2004). Yellow starthistle also invades grain fields, orchards, vineyards, cultivated crops, pastures, roadsides, and wastelands and can contaminate alfalfa, cereal grains, hay, and commercial seed (Maddox and Mayfield 1985).

The impact of all invasive plants to rangelands and pastures is estimated to be $6 billion annually (Pimentel et al. 2000; Pimentel et al. 2005). A recent study calculates that Idaho alone suffers about $8.2 million in direct losses relating to yellow starthistle's negative impacts on forage value for livestock, agricultural crops, watershed quality, and wildlife habitat in addition to secondary losses of $4.5 million (Juliá et al. 2007). Yellow starthistle is estimated to cost California ranchers about $17 million annually (Eagle et al. 2007).

Yellow starthistle spreads primarily with disturbance but can also spread to undisturbed areas (Roché et al.1994), and this accentuates the need for treatment options. A variety of control measures (mainly herbicide) have successfully controlled small infestations. However, yellow starthistle

presently infests vast areas of rugged canyon rangelands where feasibility of some control methods are limited by accessible terrain. Targeted grazing by domestic goats may be an effective means for managing yellow starthistle in steep terrain and at landscape scales.

Preliminary data and observations indicated that goats readily consumed yellow starthistle seedheads that had fully developed spines. Other livestock have not been observed to consume yellow starthistle in its mature stage. Recovery from grazing decreases as grazing occurs later in the growing season (Maschinski and Whitham 1989). Benefield and colleagues (2001) recommended that late-season control occur before 2% of the spiny heads begin flowering because as flowering progresses, seed production increases exponentially. Yellow starthistle seeds are viable as early as five days following the onset of flowering (Benefield et al. 2001). Viable seeds were produced during the time that late-season grazing occurred in this study.

Our objective of this research was to examine if grazing by goats is a viable tool for yellow starthistle management. Specifically, we evaluated the effect of late-season grazing by goats on: 1) plant and seedhead density of yellow starthistle, and 2) canopy cover of yellow starthistle and associated plants in the community.

Materials and Methods

A field study was conducted from 2006 to 2008. We compared vegetation parameters (plant and seedhead density of yellow starthistle and canopy cover of yellow starthistle, grasses, and forbs) in grazed areas to ungrazed exclosures. Vegetation assessments were conducted in each year of the study.

Study area

The study was conducted on Bentz Ridge located just northeast of White Bird, Idaho and 16 km south of Grangeville, Idaho (lat 45°45'N, long 116°17'W; elevation 496 m to 745 m; Appendix A). The aspect of the site is predominantly southeasterly, and the slope ranges from 20 to 35° for the study plots. Soils are predominantly Tannahill-Lickskillet complex (USDA 2007). Tannahill series soils are deep, well-drained soils that occur on south-facing slopes and form from igneous rock (predominantly basalt) and can be over 150 cm deep (USDA 2007). Lickskillet soils are shallow, well-drained colluviums soils that are composed of loess and weathered material from basalt and rhyolite (USDA 2007).

The research site encompassed about 380 hectares and was located in a semi-arid grassland that received annual precipitation of 54.4 to 58.8 cm with a 60-year average of 59 cm, and most of this occurred in the spring and the fall (Western Regional Climate Center 2008). Mean summer temperatures ranged from 13.3°C to 22.5°C, and mean winter temperatures ranged from -5.7°C to 2.4°C (WRCC 2008).

Winter precipitation was slightly below the long term average for all years of the study (Fig. 2.1). The initial study year experienced slightly below average fall precipitation and then slightly above average spring precipitation while summer months of all three study years were drier than normal. Fall 2005 had average temperatures while winter months of 2006 had temperatures that varied slightly from the average. This was followed by spring temperatures that were similar to average and a summer that was slightly warmer than typical (Fig. 2.2). The second study year had above average precipitation in fall 2006 while spring and summer 2007 were substantially drier than average for all the growing season. Temperatures during the fall months of the second study year varied somewhat from the long term average, and the winter was slightly cooler than average. Study year 2 was then generally warmer for most of the spring and summer months. Fall temperatures of study year 3 were close to average, and this was followed by cold weather during the winter months. Temperatures remained cool into the spring through the beginning of summer. The spring of 2008 was both wetter and cooler than the other study years and the long term average.

The study area is characterized by steep, open terrain. The land is managed by the United States Forest Service and was historically used for grazing by cattle. It is currently an active cattle allotment that receives alternating seasons of use. This rangeland is in fairly degraded condition as indicated by the dominance of exotic annual plant species. The site was selected for its relatively high density of yellow starthistle (37%). Other vegetation present was perennial and annual grasses (22%) and forbs (13%; Table 2.1). Annual grasses were more abundant than perennial grasses. Annual grass present at the site was predominantly cheatgrass (*Bromus tectorum* L.), with lesser coverage of medusahead (*Taeniatherum caput-medusae* {L.} Nevski), wild oat (*Avena fatua* L.), field brome (*Bromus arvensis* L.), and rattlesnake brome (*Bromus briziformis* Fisch. & C.A. Mey.). Perennial grasses comprised less than 4% of the grasses present and mainly included bluebunch wheatgrass (*Pseudoroegneria spicata* {Pursh.} A. Löve) and Sandberg bluegrass (*Poa secunda* J. Presl). The forbs present on the study area were a mixture of native and introduced, perennial and annual species. Weedy exotic annual forbs besides yellow starthistle were fiddleneck (*Amsinckia* Lehm.), field

pennycress (*Thlaspi arvense* L.), redstem stork's bill (*Erodium cicutarium* {L.} L'Hér Ex Aiton), tall tumblemustard (*Sisymbrium altissimum* L.), and yellow salsify (*Tragopogon dubius* Scop.). Perennial forbs present included arrowleaf balsamroot (*Balsamorhiza sagittata* {Pursh} Nutt.), biscuitroot (*Lomatium spp.*), lupine (*Lupinus spp.*), field bindweed (*Convolvulus arvensis* L.), penstemon (*Penstemon spp.*), and common yarrow (*Achillea millefolium* L.). Rose (*Rosa spp.*) was also common throughout the study area.

Twenty-four plots were established on Bentz Ridge in the summer of 2006 (study year 1). Plots were not randomly established because of the ruggedness of the terrain—some areas were not feasible locations for plots. We established plots in six locations with four plots in each location (Appendix A). At each plot, we marked two subplots of similar vegetation and topography in close proximity to each other. One subplot was then randomly selected as the control (fenced) while the other remained unfenced to be the grazed treatment subplot (Fig. 2.3). Each fenced subplot was a 15 x 7 m exclosure constructed from ElectroNet™ temporary fencing (Premier 1 Supplies, Washington, Iowa). This fencing was 88.9 cm tall and had 9 horizontal twines, white plastic vertical struts every 30.5 cm, and PVC posts every 3.65 m. The fencing was not electrified for this study, but goats avoided the fences because they were familiar with these fences in an electrified state. Aspect, slope, bearing, and location determined by Global Positioning System (GPS) units were recorded for all plots.

Treatment application

Grazing service provider Ray Holes of Prescriptive Grazing Services (White Bird, Idaho) managed the goat grazing for the duration of this study. In the first year of the study (2006), we applied goat grazing to the study area twice within a season because Thomsen et al. (1993) demonstrated that follow-up treatments within a season were necessary to treat re-growth of yellow starthistle. Also, it was widely believed that livestock avoid yellow starthistle when it is in full spine. However, after observing in study year 1 that goats consumed fully developed yellow starthistle seedheads, we adjusted our methods so that goat grazing was only applied once late-season in study years 2 and 3 (2007, 2008). In study year 1 (2006), 1233 yearling goats grazed about 405 ha in 68 days (June 25 to Aug. 31). Grazing occurred a second time Oct. 10 to Nov. 10. In study year 2 (2007), 1657 dry nannies grazed about 648 ha in 42 days (July 27-Sept. 10). For study year 3 (2008), 1706 dry nannies grazed about 809 ha in 52 days (Aug. 8 to Sept. 28). The area grazed increased each year and expanded beyond the study area. This was a management decision determined by the grazing

service provider and the United States Forest Service. The goats used were of predominantly Boer and Boer-cross breeds that had experience grazing yellow starthistle and foraging on rugged terrain. Two herders remained with the goats each year to ensure that even grazing of the ridge occurred, that bedding sites were changed every few days, and that animals had minimal impact on riparian areas.

Vegetation assessment

Study year 1 (2006). Vegetation assessments were not conducted before grazing in study year 1. Data were collected after grazing was applied for the second time for that year and goats were removed from the site. Vegetation sampling consisted of recording density and estimating canopy cover in quadrats located along a pace transect within each subplot. After goat grazing was applied in 2006, the number of yellow starthistle plants and mature seedheads and percent canopy cover of yellow starthistle, grass, and forbs were assessed for five quadrats in each subplot. Quadrats were 50 x 25 cm and placed on alternating sides of the pace transect so that each quadrat fell about halfway between the transect and the closest side of the exclosure.

Study year 2 (2007). A permanent transect was staked for each subplot in 2007 (Fig. 2.3). For the control, the lower stake was placed three paces up from the lowest edge of the exclosure, and the transect line was run eight meters uphill toward the top of the exclosure where another stake was placed. For the treatment subplot, the transect line was run parallel to the control transect and likewise staked. Starting at the lower stake with quadrat 1, eight quadrats were located 1.5 m from the transect at 1-m intervals on alternating sides (Fig. 2.3). Vegetation assessments were avoided 1 m along the borders of the exclosures because we predicted that these areas might receive additional impact from the goats.

Vegetation assessments for study year 2 were conducted July 24 to Aug. 2 before goats grazed the area and again Sept. 8 to Sept. 23 after goat grazing was complete. Vegetation assessments consisted of densities of yellow starthistle plants and seedheads and percent canopy cover of yellow starthistle, grass and forbs as it did in study year 1. Anytime a quadrat fell over an area that was predominantly rock (greater than 40 %), that quadrat was adjusted 25 cm out from the transect line until a less rocky area was reached. Cover was estimated to the nearest 1% up to 15%. Cover over 15% was estimated to the nearest 5%. Cover of 1% indicated the presence of only one or two plants.

Study year 3 (2008). Vegetation assessments for the final study year were consistent with data collected previously except the number of quadrats examined at each subplot was increased from

eight to ten. Twenty-four additional subplots were established in the study area that matched the terrain of the existing grazed subplots. Each of these additional subplots was placed in the vicinity of an existing study plot. Vegetation assessments were also conducted at these vicinity subplots to ascertain if grazing effects were occurring consistently across the landscape and not just concentrating around the exclosures due to either animal attraction or herder bias.

Statistical analysis

Vegetation assessments were conducted after goat grazing in 2006, 2007, and 2008, and before grazing in 2007 and 2008 and described as "before" and "after" grazing. Variables in each set were densities of yellow starthistle plants and yellow starthistle seedheads, as well as canopy cover of yellow starthistle, grass, and forbs. Vegetation assessments were conducted immediately preceding and following grazing treatment for 2007 and 2008. The vegetation assessment for 2006 occurred in November 2006 after grazing treatment concluded in early November.

Variables were examined for normality using the capability procedure of SAS (SAS 2004). Normality of variables, including plant and seedhead density data collected before grazing and seedhead density collected after grazing, was improved by square root transforming (Steel and Torrie 1980). Normality of seedhead density collected after grazing was improved by logarithmic transformation (Steel and Torrie 1980). Canopy cover variables that were arc sine transformed (Steel and Torrie 1980) to obtain normal distributions were forb cover collected both before and after grazing and yellow starthistle cover for after grazing. Transformed variables and untransformed variables with acceptable normality were analyzed with a completely randomized mixed model analysis of variance using the mixed model procedure of SAS (SAS 2004). Different covariant structures were examined, and the most appropriate was selected for each variable. Each model was analyzed for differences between years, between grazed and ungrazed subplots, and for a treatment by year interaction. Variables that produced treatment by year interactions were further analyzed separately by year. Differences were considered significant at the α level of $P<0.05$.

Results

Grazing effects on yellow starthistle

Data collected at the additional subplots established in the vicinity of paired study plots in 2008 supported the assumption that grazing treatment occurred evenly across the landscape and was not focused on the areas immediately surrounding each exclosure as after grazing was applied, these

newer subplots had fewer yellow starthistle plants than the ungrazed areas inside of all exclosures (P<0.001) and were similar to the grazed plots paired with the exclosures (P=0.306).

There was a treatment by year interaction for yellow starthistle plant density after grazing (P=0.003); therefore, treatment effects were examined by year, and we found that grazing resulted in decreased yellow starthistle plant density outside compared to inside the exclosures (P<0.001 for each year; Fig. 2.4; Appendix B). The interaction was likely because of the greater difference in plant number between the grazed and ungrazed plots found in 2007 compared to 2006 or 2008. Even before the grazing treatment was applied in 2007 and 2008, fewer yellow starthistle plants were found in grazed plots compared to inside the exclosures (P<0.001; Fig. 2.4; Appendix B). More plants were present in the study area before grazing in 2007 than 2008 (P<0.001), but there was no treatment by year interaction (P=0.104).

Grazing reduced yellow starthistle seedhead density compared to the ungrazed control (P<0.001; Fig. 2.5; Appendix B) with no treatment by year interaction (P=0.254). The seedhead density was highest in 2006 (P<0.001) with lower density observed in 2007 or 2008, which were equivalent to each other. Seedhead density before grazing was similar inside and outside the exclosures (P=0.331; Fig. 2.5; Appendix B). More seedheads were present in the study area before grazing in 2007 than 2008 (P=0.003) but there was no treatment by year interaction (P=0.556).

The effect of grazing on yellow starthistle canopy cover depended on the year it was examined (treatment by year interaction; P=0.032 after grazing; P=0.042 before grazing; Fig. 2.6; Appendix C). Cover of yellow starthistle did not differ between grazed and ungrazed subplots after grazing in 2006 (P=0.072), while grazing decreased yellow starthistle cover in 2007 and 2008 (P<0.001 for both years). There was less yellow starthistle cover in grazed areas compared to the ungrazed control before grazing occurred in 2007 (P=0.005), but grazed and ungrazed subplots had similar yellow starthistle cover before grazing in 2008 (P=0.212; Fig. 2.6; Appendix C).

Grass and forb cover response

Cover of perennial grasses accounted for less than 4% of the total grass composition; thus, annual and perennial grasses were combined for analysis. Grass canopy cover after grazing was similar for grazed and ungrazed areas (P=0.063) and over the study years (P=0.142; Fig. 2.7; Appendix C) with no treatment by year interaction (P=0.457). We found a treatment by year interaction for grass canopy cover before the grazing treatment was applied (P=0.029), but when

analyzed by year, we found that grazed and ungrazed subplots had similar grass cover for both 2007 (P=0.118) and 2008 (P=0.986; Fig. 2.7; Appendix C).

With the exclusion of yellow starthistle, forbs represented only 13% of the average cover of the study site, so perennial and annual forbs were combined. There was a treatment by year interaction for forb canopy cover after grazing (P<0.001). Forb cover for grazed subplots was similar to the ungrazed exclosures in 2006 (P=0.961) and 2008 (P=0.179; Fig. 2.8; Appendix C). In 2007, however, grazing reduced forb cover compared to the ungrazed exclosures (P<0.001). Forb canopy cover was similar for grazed and ungrazed subplots before grazing (P=0.497; Fig. 2.8; Appendix C). There was less total forb cover in 2007 than in 2008 (P<0.001), but there was no treatment by year interaction (P=0.547).

Discussion

Impacts of targeted goat grazing to yellow starthistle

Goats uniformly and thoroughly grazed yellow starthistle as indicated by the prominent lack of seedheads or foliage throughout most of the study area after grazing. In this study, goats grazed yellow starthistle late-season when plants were in full spine and when resources might not be available for the plants to compensate for herbivory. As anticipated, we observed no evidence of re-growth when we conducted vegetation assessments after the grazing treatment. Other studies have demonstrated that timing of grazing is critical for affecting yellow starthistle (Thomsen et al. 1989; Thomsen et al. 1993; Hovde 2006). Hovde (2006) observed that when yellow starthistle was grazed in early stages of its growth (i.e., rosette, bolting, and pre-spiny), the plant responded with sufficient re-growth so that plant density, seedhead production, and yellow starthistle cover were greater in grazed pastures compared to the ungrazed control at the end of the season. Early season grazing of yellow starthistle required a second grazing treatment in the same season to target re-growth (Thomsen et al. 1993).

Cattle, sheep, and goats readily graze yellow starthistle, but use differs among animal species. Thomsen and colleagues (1993) observed that sheep grazed yellow starthistle when it was in the rosette stage but goats passed over rosettes and grazed yellow starthistle when it was in the bolting stage. Hovde (2006) noted that both cattle and sheep grazed yellow starthistle in the rosette and bolting stage though sheep grazed yellow starthistle more readily than cattle. In the flower stage, sheep stripped leaves and avoided seedheads, but cattle avoided yellow starthistle plants completely

in this stage (Hovde 2006). This knowledge suggests that the phenological stages being grazed may dictate what class of livestock should be used for grazing yellow starthistle.

Thomsen and colleagues (1993) observed that goats avoided yellow starthistle with spines, but in our study, goats readily grazed yellow starthistle in full spine. This may indicate that animal experience is important. For example, Walker and colleagues (1992) found that lambs with previous exposure to leafy spurge generally had greater preference for this plant later in life. Goats in the Thomsen et al. study (1993) had no previous experience with yellow starthistle while the goats used in our research were familiar with grazing yellow starthistle. Flores and colleagues (1989) demonstrated that lambs that grew up eating shrubs were more efficient at harvesting shrubs and had higher intake rates than sheep that had no experience grazing shrubs. Thus, goats in our study may also have developed the necessary foraging skills to efficiently harvest yellow starthistle.

After just one year of treatment, we found fewer yellow starthistle plants where goats had grazed the previous year. This was unexpected because yellow starthistle responses following initial treatment are often unnoticeable because of the established seedbank (Joley et al. 1992; Thomsen et al. 1997). Furthermore, grazing of yellow starthistle causes disturbance and increases sunlight penetration by removing canopy cover. Removing litter can increase germination and seedling survival (Roché and Thill 2001; Gelbard and Harrison 2005). Yellow starthistle's ruderal traits of rapid growth and high seed production suggest that disturbance should favor yellow starthistle germination and growth (Roché and Thill 2001).

The reduced yellow starthistle density that we observed in grazed sites before grazing was applied in 2007 and 2008 may indicate that seed rain was effectively prevented by late-season goat grazing. Seed and seedling densities were decreased to 3.9 and 1.1% of initial densities, respectively, when seed rain was prevented for three years in a California study (Joley et al. 1992). This suggests that the yellow starthistle seedbank can be rapidly depleted, especially because yellow starthistle exhibits very little dormancy (Roché 1965; Joley et al. 1992; Benefield et al. 2001) and germinates as soon as moisture is available (Callihan et al. 1989; DiTomaso et al. 1999a). Seedheads removed by goats in our study may have reduced seed abundance to a level that resulted in reduced seedling establishment the subsequent year.

Research and field observations clearly demonstrate that livestock pass viable seeds in feces (e.g., Burton and Andrews 1948; Heady 1954; Blackshaw and Rode 1991; Manzano et al. 2005).

Goats in our study were ingesting viable yellow starthistle seeds, but less than 3% of recovered seeds were viable following passage through the digestive tract of a goat (see Chapter 3). Other weed seed species have also experienced low survival and substantial reductions in viability following ingestion (Thill et al. 1986; Lacey et al. 1992; Wallander et al. 1995). For example, 2 to 7% of common crupina (*Crupina vulgaris* Cass) seeds fed to cattle, horses, and deer were recovered from feces and viable, and no seeds were recovered from sheep feces (Thill et al. 1986). Seed destruction in the goat digestive tract may have also contributed to decreased seed rain and might explain why we saw decreased yellow starthistle densities before grazing was applied in 2007 and 2008 despite conditions that seemed to promote germination and seedling recruitment (e.g., disturbance, reduced cover, sufficient moisture availability).

Other treatment methods that remove viable yellow starthistle seeds, such as fire, may have similar affects as grazing on seedbank dynamics. DiTomaso and colleagues (1999b) found no change in yellow starthistle cover after one year of prescribed fire in California, but seedbank and seedling density were still decreased by about 74% and 83%, respectively, following the initial summer burn. Two more summers of prescribed burning reduced yellow starthistle cover and further reduced the seedbank and seedling density. The decrease in yellow starthistle cover and plant density may have been caused by a decline of seeds that was observed in the seedbank (DiTomaso et al. 1999b). The seedbank declined because of germination and seed mortality and the prevention of seed rain (Kyser and DiTomaso 2002). While we lack similar knowledge of yellow starthistle seedbank dynamics in our study area, if goat grazing is reducing seed rain of yellow starthistle, continued seasons of goat grazing should result in further declines of yellow starthistle plant density.

We observed no difference in seedhead density between the control and grazed areas before grazing was applied in 2007 and 2008 despite reduced plant densities. This agrees with DiTomaso and colleagues (1999b) where prescribed fire resulted in increased seedhead production per plant in the growing seasons following the burn treatment. A possible reason for this is that while plant densities may be reduced, seedhead production per plant may have increased (Uygur et al. 2004), resulting in relatively constant seedhead production. This may also explain why we observed no difference in yellow starthistle cover before grazing in 2008.

Within four years after the cessation of prescribed burning treatment, the seedbank density in burned areas increased from <1 to 80% of the seedbank density of the control site, and seedling

recruitment increased from <1 to 56% (Kyser and DiTomaso 2002). This was attributed to the failure to eradicate yellow starthistle (Kyser and DiTomaso 2002). Under California conditions, a small number of viable seeds can persist in the seedbank although most seeds are removed from the seedbank (i.e., germinated, preyed upon, died) within three years (Joley et al. 1992). Unplumed and plumed yellow starthistle seeds in Idaho have average longevities of six and ten years, respectively (Callihan et al. 1993).

Impacts of targeted goat grazing to non-target vegetation

Goat grazing had few effects on canopy cover of either grasses or forbs in our study. One exception was in 2007 where forb cover was lower in the grazed areas compared to the control after grazing was applied. Natural forb recruitment may have been low due to the combined effects of the dry year and grazing and trampling by goats. Alternatively, forbs may have experienced increased use by goats for that study year. However, goat grazing of yellow starthistle occurred when most other forbs and grasses were dormant or senesced. Forbs and grasses may be avoided during summer grazing because of low palatability and nutritive value compared to the yellow starthistle that was abundantly available. Yellow starthistle offers nutritive value throughout its entire life cycle (Frost et al. 2008). At seed set, the phenological stage when grazing occurred in our study, yellow starthistle leaves and seedheads have 6.7 and 4.6% crude protein content, respectively and neutral detergent fiber (NDF) content of 43.8 and 47.5% (Frost et al. 2008).

The lack of difference in non-target vegetation cover in grazed areas compared to the control indicates that they were not readily consumed by goats. The dominant plants on Bentz Ridge besides yellow starthistle were annual grasses, notably cheatgrass. Cheatgrass received very little use by goats when it was actively growing and no use by goats after it had cured in a grazing study in a sagebrush-bunchgrass community in Oregon (Fajemisin et al. 1996). It must be noted that cheatgrass made up a small amount of the available forage in this Oregon study compared to its abundance on Bentz Ridge in our study.

Perennial grasses were not abundant enough on our study site for us to determine how targeted goat grazing of yellow starthistle may have affected these plants. Bluebunch wheatgrass was a substantial part of the diets of goats grazing sagebrush-bunchgrass habitat types in Oregon when bluebunch wheatgrass was actively growing and after it had cured (Fajemisin et al. 1996). Research is

needed to determine perennial grass response to goat grazing of yellow starthistle-infested sites that have a greater abundance of perennial grasses than our study area.

Variations of yellow starthistle over time

There appeared to be a trend for decreased cover of yellow starthistle over the three study years that was independent of the goat grazing, and seedhead densities were markedly lower in 2007 and 2008 than in 2006. Yellow starthistle growth is highly dependent on rainfall patterns, specifically, the timing and amount of initial rainfall and the following dry period (Joley et al. 2003). Recruitment and germination from the seedbank and subsequent seedling survival are likewise dependent on precipitation (Joley et al. 2003). Precipitations patterns during our study could possibly explain the decreased yellow starthistle cover and seedhead production. Precipitation in fall 2006 was above average and may have created ideal conditions that encouraged yellow starthistle germination. Most yellow starthistle germination occurs in the fall (Joley et al. 2003). High germination rates may have then occurred in fall 2006 (the beginning of study year 2) when moisture was sufficient for seedling recruitment and establishment, but the winter, spring, and summer of 2007 were much drier than average. These low moisture conditions may have caused the drastic decline in seedheads observed in 2007 because many yellow starthistle seedheads desiccate in drought years (L. Wilson, pers. comm.). Precipitation in fall 2007, spring 2008, and summer 2008 was close to average or above and suggests that moisture should not have been a factor limiting seedhead production or yellow starthistle cover in 2008, so causes of the low amount of seedheads and yellow starthistle cover in this year are uncertain.

Yellow starthistle plant density was higher in 2007 than either 2006 or 2008. This may be due to the above-average precipitation in fall 2006, or this may reflect the oscillating pattern that has been noted for yellow starthistle where a year of high plant density is followed by a year of low plant density (Enloe et al. 2004; Uygur et al. 2004; Hovde 2006). Enloe and colleagues (2004) attributed this to soil water recharge; a year of high water use by large populations one year prevents soil water recharge and thus negatively impacts the following generation of yellow starthistle. Yellow starthistle cover is highly correlated with soil moisture availability (Roché et al. 1994).

Alternatively, Uygur and colleagues (2004) suggested that the cyclic nature sometimes observed in yellow starthistle may be caused by the presence of natural enemies: yellow starthistle can rapidly build up its population density, and over time natural enemies will also increase in density until yellow starthistle reproductive output is suppressed. This may also account for the large decrease

observed in seedhead density from 2006 to 2007 and 2008. Two seedhead-feeding insects of yellow starthistle, the yellow starthistle hairy weevil (*Eustenopus villosus*) and the yellow starthistle flower weevil (*Larinus curtus*), appeared to be abundant throughout the study area on Bentz Ridge, and many seedheads appeared to be damaged; however, data about these weevils and their impact to yellow starthistle at this site were not collected. Precipitation patterns and seedhead-feeding insects may be exerting dual pressure on yellow starthistle resulting in the multiple effects that we observed.

Management Implications

It is difficult to predict the value of targeted grazing for restoration in our study area. After sixty-four years, natural revegetation of a disturbed site in southeastern Washington resulted in a predominantly perennial grass community on the north-facing slope of the site while the south-facing slope was dominated with cheatgrass and yellow starthistle (Dillon 1967). Shifting the balance of competition in favor of perennial species may thus not be the realistic goal for south-facing infested sites like Bentz Ridge in our study. Continuing the present study through several more seasons of goat grazing would better reveal impacts to the plant community. Seeding of perennial grasses species would be a necessary part of restoration for Bentz Ridge, because the low abundance of perennial grasses in the study area suggests that the seedbank of perennial grasses is likely small.

Yellow starthistle can rapidly recover from small populations and reduced seedbanks. For example, Kyser and DiTomaso (2002) found that three years of prescribed burning drastically reduced yellow starthistle cover and the size of the seedbank at an infested study site, but within four years the burned areas had nearly returned to their pre-burn state. A cessation of goat grazing on Bentz Ridge after three years of grazing will likely result in a return to pre-grazing yellow starthistle cover and density.

Eradication of yellow starthistle from North America is likely not a feasible goal. The most appropriate treatment for yellow starthistle may thus follow an Integrated Pest Management (IPM) approach in which multiple treatment methods coupled with knowledge of yellow starthistle's life cycle are used to assert selection pressure on yellow starthistle in a way that is both economical and ecological. The components of an IPM are conditional upon land management objectives. These objectives may vary from adding forage value to a yellow starthistle-infested landscape to restoration by decreasing yellow starthistle abundance and increasing native species abundance.

Infested canyon rangelands with limited accessibility pose a special challenge for management. Targeted grazing by goats offers two distinctive advantages for managing such infested areas: 1) goats can easily navigate the rugged terrain of these canyonlands, and 2) goats will graze yellow starthistle throughout most of its life cycle. Our research clearly shows that goats will even graze yellow starthistle when the plant is in full spine. Multiple grazing treatments or a single late-season grazing treatment can reduce yellow starthistle populations. This creates a large window of opportunity for grazing treatment and flexibility for land and livestock managers.

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Table 2.1. Common plants on a yellow starthistle-dominated site near White Bird, Idaho. Forbs are listed if they appeared at 25% or more of the plots while all grasses found are listed. Common and scientific names are according to the USDA PLANTS Database (http://plants.usda.gov/).

| Common name	Scientific Name
Perennial Grasses	
Bluebunch wheatgrass	*Pseudoroegneria spicata*
Bulbous bluegrass	*Poa bulbosa*
Mountain brome	*Bromus marginatus*
Kentucky bluegrass	*Poa pratensis*
Sandberg bluegrass	*Poa secunda*
Timothy	*Phleum pretense*
Western wheatgrass	*Pascopyrum smithii*
Annual Grasses	
Cheatgrass	*Bromus tectorum*
Field brome	*Bromus arvensis*
Hare barley	*Hordeum murinum*
Medusahead	*Taeniatherum caput-medusa*
Millet	*Panicum miliaceum*
Rattlesnake brome	*Bromus briziformis*
Wild oat	*Avena fatua*
Perennial Forbs	
Arrowleaf balsamroot	*Balsamorhiza sagittata*
Biscuitroot	*Lomatium spp.*
Common yarrow	*Achillea millefolium*
Field bindweed	*Convolvulus arvensis*
Largeflower triteleia	*Triteleia grandiflora*
Lupine	*Lupinus spp.*
Penstemon	*Penstemon spp.*
Varileaf phacelia	*Phacelia heterophylla*
Annual forbs	
Black medick	*Medicago lupulina*
Bristly hawksbeard	*Crepis setosa*
Common sunflower	*Helianthus annuus*
Fiddleneck	*Amsinckia spp.*
Field pennycress	*Thlaspi arvense*
Prickly lettuce	*Lactuca serriola*
Redstem stork's bill	*Erodium cicutarium*
Tall tumblemustard	*Sisymbrium altissimum*
Yellow salsify	*Tragopogon dubius*
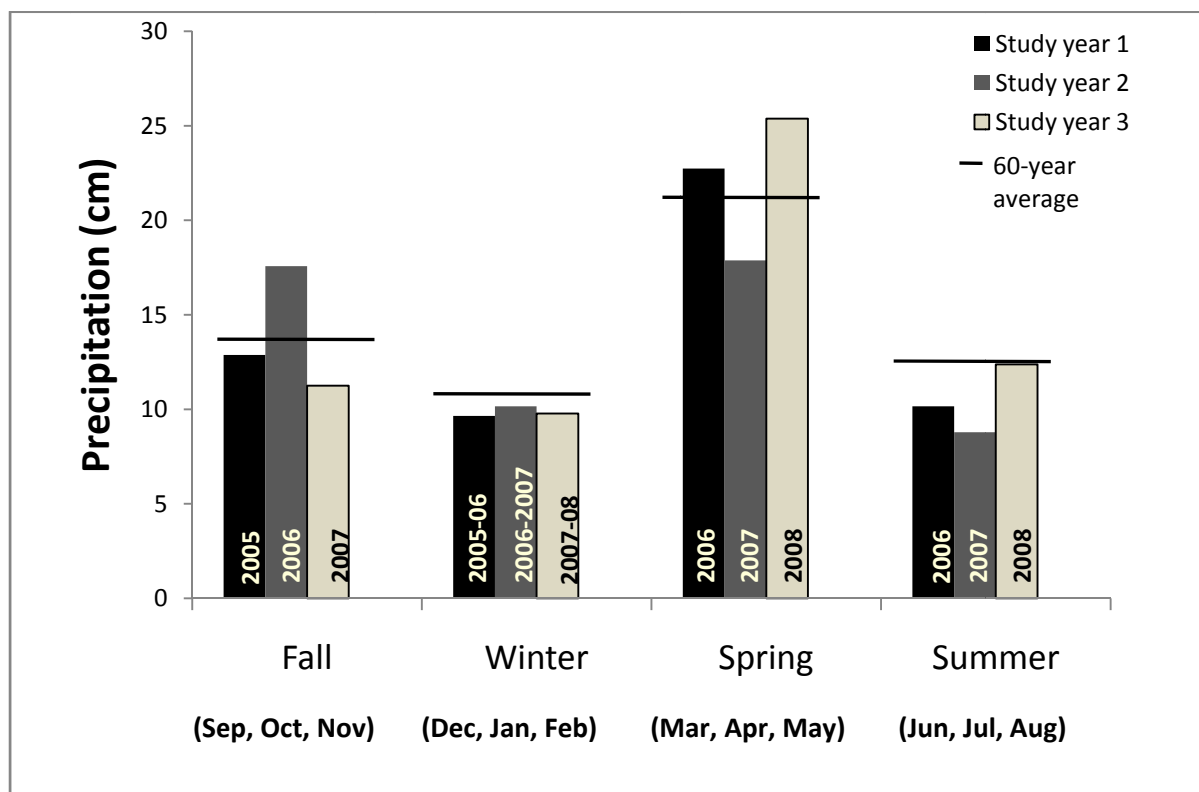
Figure 2.1. Seasonal precipitation from fall 2005 to summer 2008 compared to the 60-year average for Grangeville, Idaho, located near the Bentz Ridge study site in north central Idaho. Data obtained from the Western Regional Climate Center; http://www.wrcc.dri/edu/summary/. Values for July and August 2008 were not available from WRCC, so those values were obtained from http://weather.msn.com/.
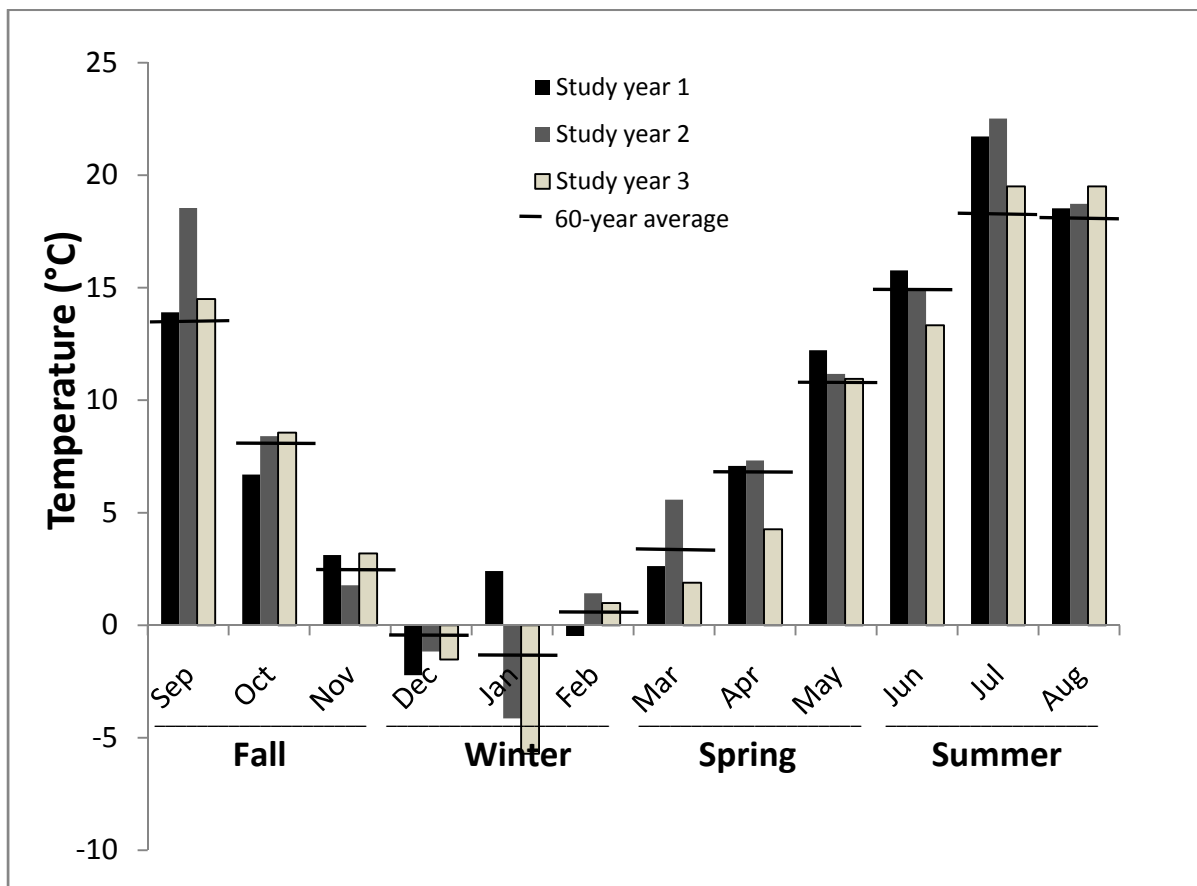
Figure 2.2. Mean monthly temperature from fall 2005 to summer 2008 compared to the 60-year average for Grangeville, Idaho, located near the Bentz Ridge study site in north central Idaho. Study year 1 includes Sep. 2005 to Aug. 2006; study year 2 includes Sep. 2006 to Aug. 2007; study year 3 includes Sep. 2007 to Aug. 2008. Data obtained from the Western Regional Climate Center; http://www.wrcc.dri/edu/summary/. Values for July and August 2008 were not available from WRCC, so those values were obtained from http://weather.msn.com.

Figure 2.3. Exclosures were randomly placed in groups of four throughout the study area. Two similar subplots were selected, and the control and treatment were randomly applied between the subplots. This was randomly decided if both sides were determined to be equally similar to the control. The treatment and control of each plot was staked with a transect line, and ten quadrats were taken along each transect for 2008.
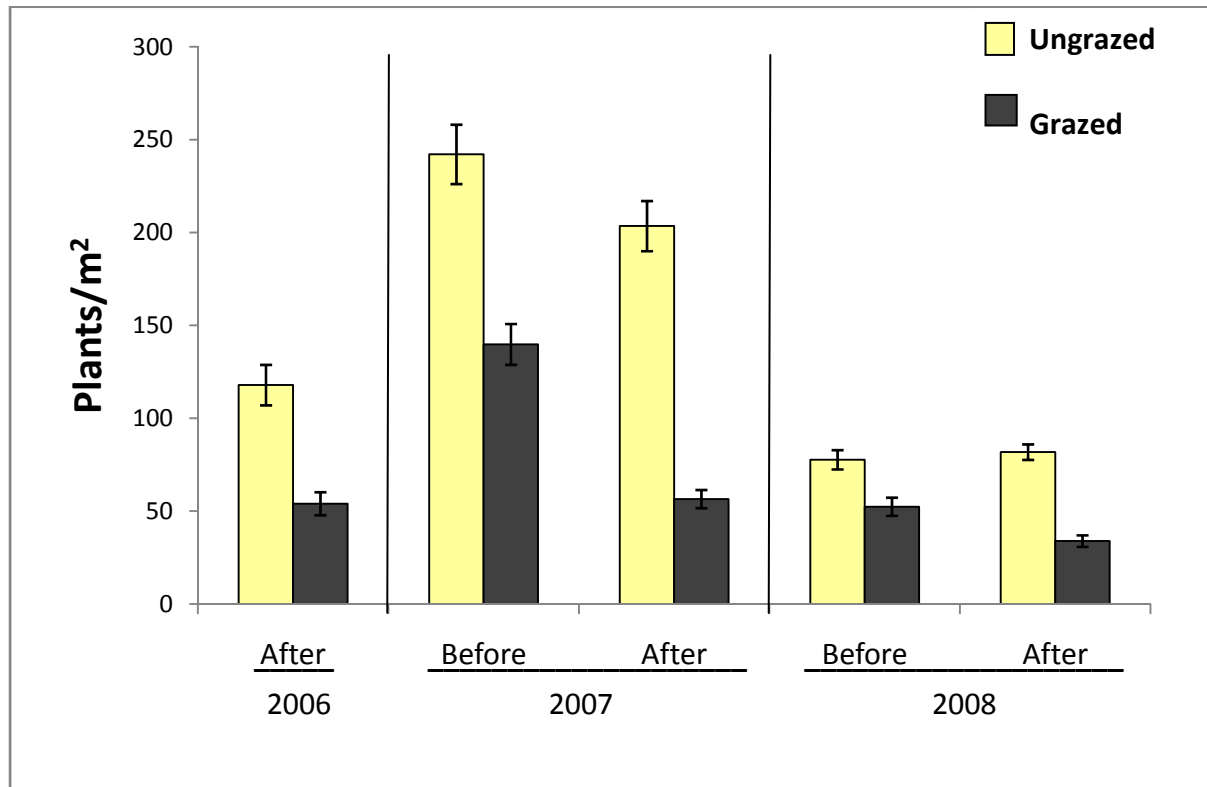
Figure 2.4. Density of yellow starthistle plants with standard error bars in grazed and ungrazed plots after goat grazing in 2006 and before and after grazing in 2007 and 2008 in a yellow starthistle-dominated site in north central Idaho.

Figure 2.5. Density of yellow starthistle seedheads with standard error bars in grazed and ungrazed plots after goat grazing in 2006 and before and after grazing in 2007 and 2008 in a yellow starthistle-dominated site in north central Idaho.
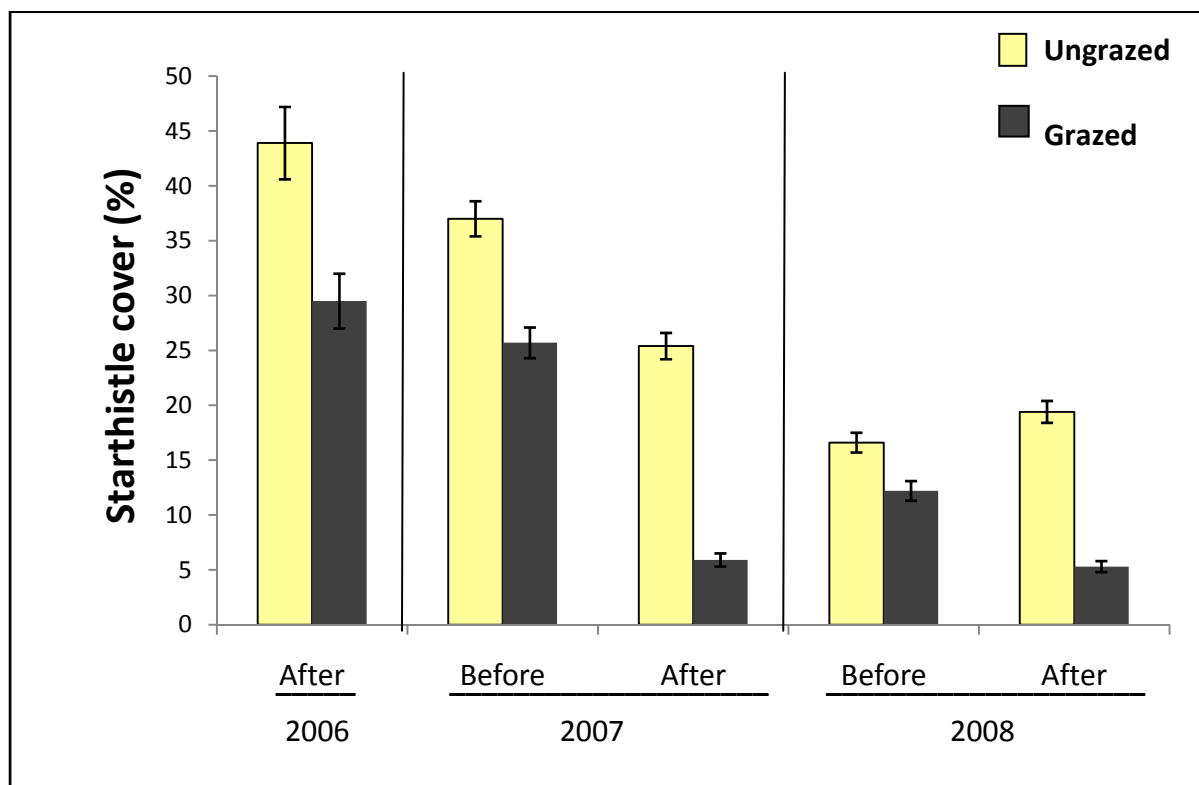
Figure 2.6. Yellow starthistle canopy cover with standard error bars in grazed and ungrazed plots after goat grazing in 2006 and before and after grazing in 2007 and 2008 in a yellow starthistle-dominated site in north central Idaho.
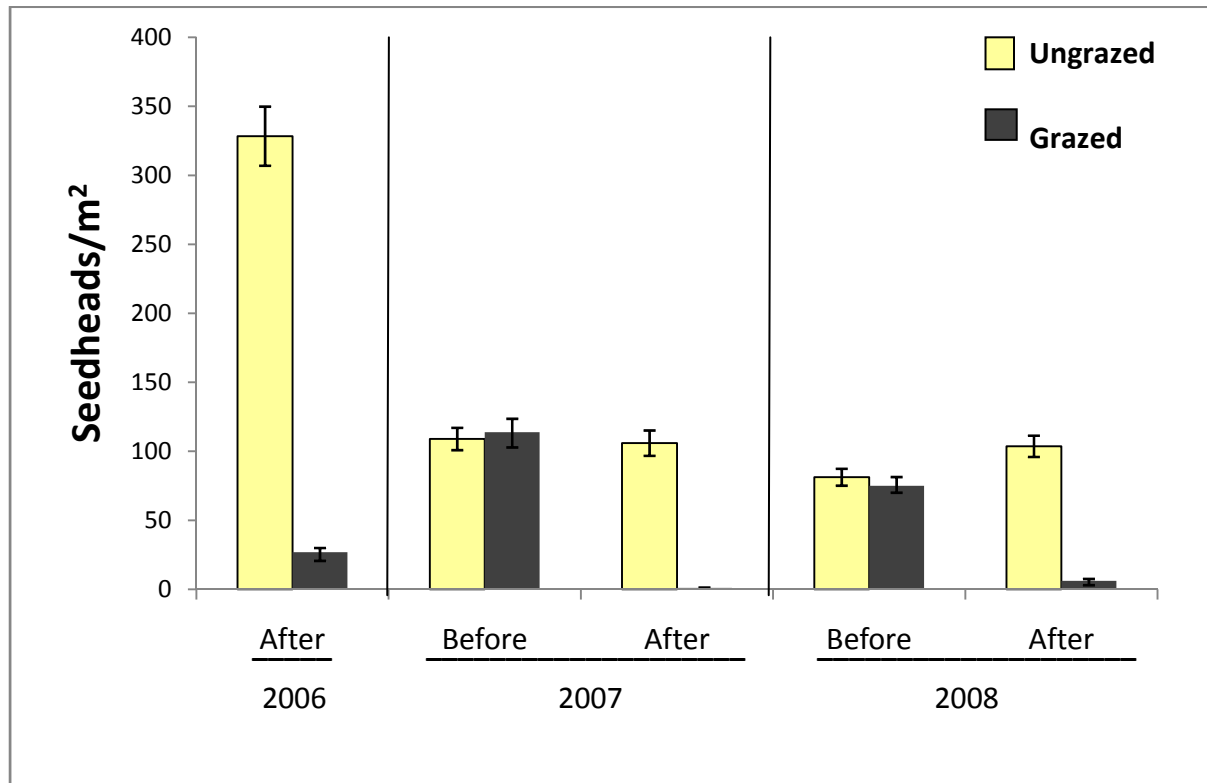
Figure 2.7. Grass canopy cover with standard error bars in grazed and ungrazed plots after goat grazing in 2006 and before and after grazing in 2007 and 2008 in a yellow starthistle-dominated site in north central Idaho.

Figure 2.8. Forb canopy cover with standard error bars in grazed and ungrazed plots after goat grazing in 2006 and before and after grazing in 2007 and 2008 in a yellow starthistle-dominated site in north central Idaho.
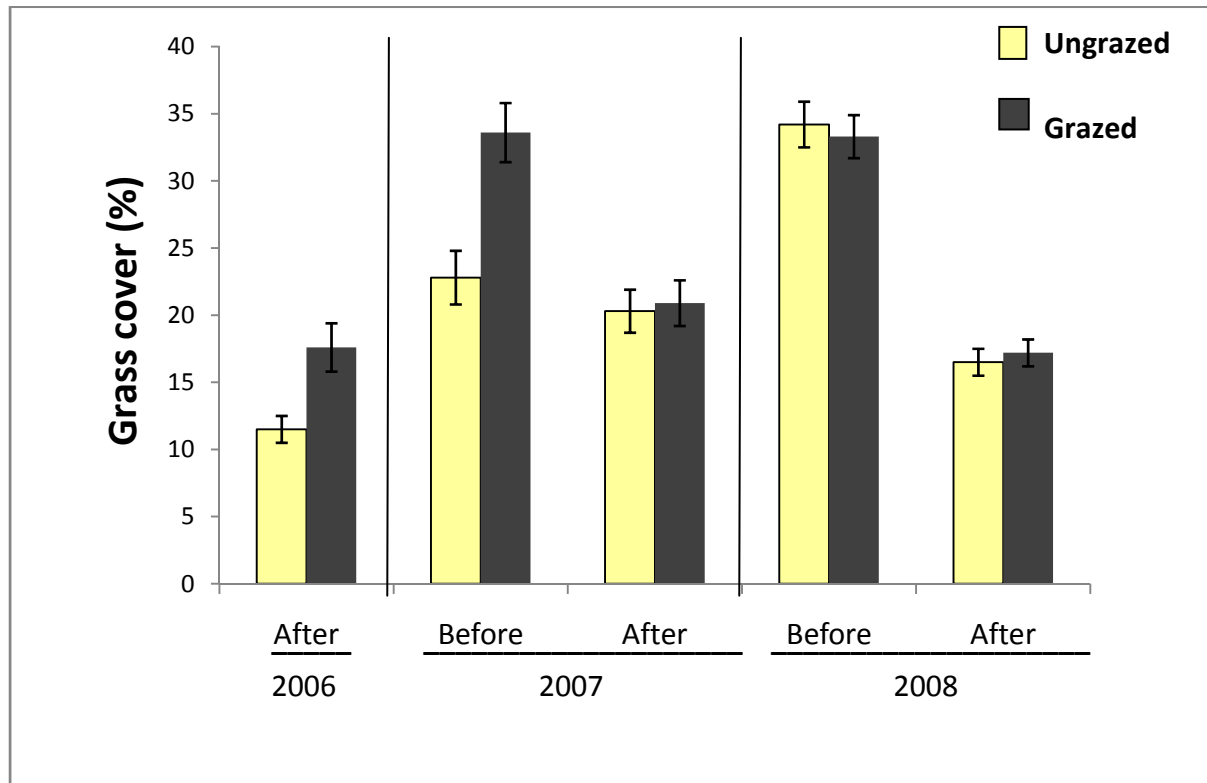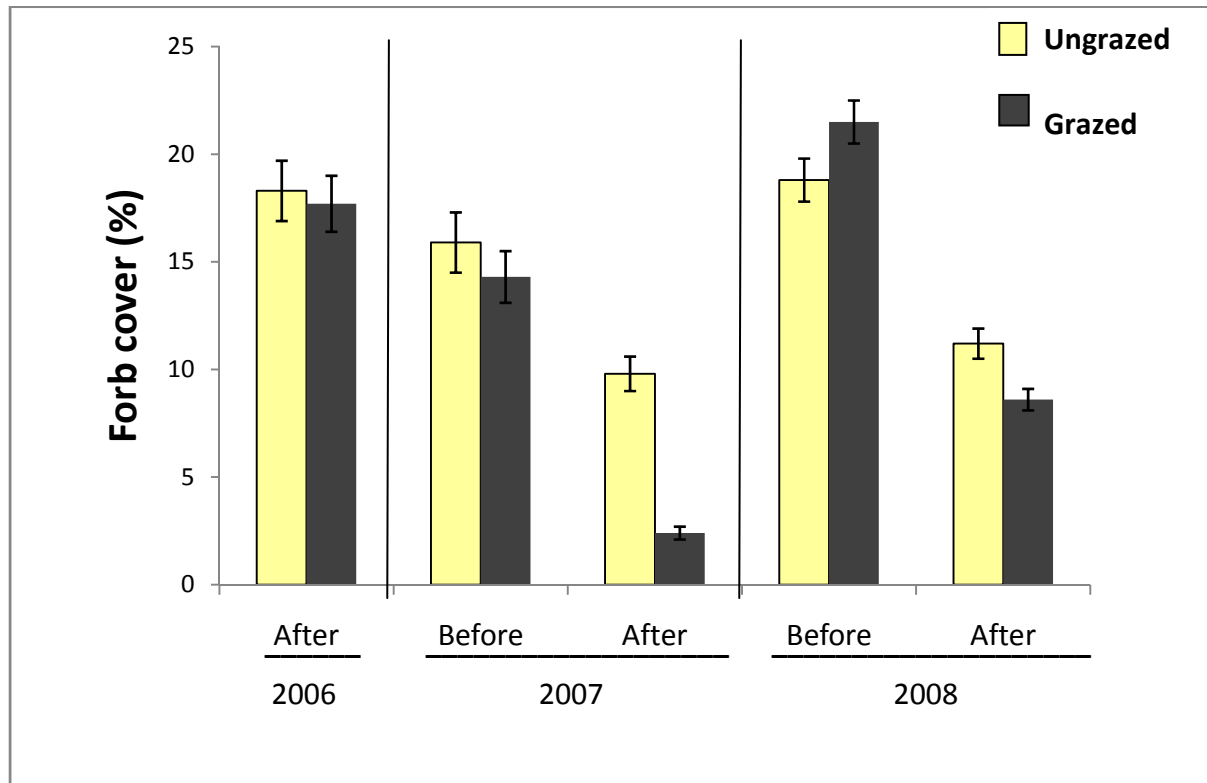
Chapter 3:   Survival of Yellow Starthistle Seeds through the Digestive Tract of a Goat

Introduction

Targeted livestock grazing with goats, sheep or cattle as a weed management tool has been gaining popularity and has application for many weedy species. For example, goat grazing can reduce yellow starthistle (*Centaurea solstitialis* L.) plant densities (see Chapter 2) and sheep grazing can reduce spotted knapweed (*Centaurea stoebe* L.) plant densities (Olson et al. 1997). Sheep have been used successfully to control leafy spurge (*Euphorbia esula* L.; Lacey et al. 1984). One concern that arises with targeted grazing is the potential spread of weed seed through passage in feces thus aggravating the very problem that targeted grazing is trying to address, that of seed dispersal. Information on seed viability of some major weeds in the United States following digestion by livestock is summarized in Table 3.1.

Yellow starthistle is an especially troublesome weed that has had severe economic and ecological impacts in the western United States (Duncan et al. 2004; Eagle et al. 2007; Juliá et al. 2007). Goats will graze yellow starthistle throughout most of its life cycle even when the plant is in full spine, and this creates a large window of opportunity for grazing treatment. Late-season grazing by goats may be especially effective for managing yellow starthistle (see Chapter 2). During late-season grazing, however, goats are consuming mature seedheads fully loaded with viable seeds. This creates a potential for spreading yellow starthistle as goats are herded or transported to un-infested areas. Information about the passage and survival of yellow starthistle seeds, as well as other weed seeds, is important information for both livestock managers and weed managers. Our objective in this study was to determine the passage rate and viability of yellow starthistle seeds after ingestion by goats.

Materials and Methods

A study was conducted at the University of Idaho Sheep Research and Teaching Center with five female goats for the recovery of yellow starthistle seeds. All procedures related to the use of animals were approved by the University of Idaho Animal Care and Use Committee as Protocol #2009-11.

Seed collection

Mature yellow starthistle seedheads were clipped from plants in August 2008 from infestations on Bentz Ridge located just northeast of White Bird, Idaho and 16 km south of

Grangeville, Idaho (lat 45°45'N, long 116°17'W; elevation 496 m to 745 m). Seeds were removed from the seedhead by hand threshing. Only plump, viable appearing seeds were retained. Both types of yellow starthistle seeds, plumed and unplumed, were collected. Plumed and unplumed seeds were separated and counted to determine the relative abundance of each. About 15% of all seeds collected were unplumed. Seeds were stored in paper coin envelopes in the dark at room temperature until used in the seed recovery study.

Collection of goat feces in the field

Goat feces collected around goat bedding areas on Bentz Ridge in September 2008 were returned to the lab to retrieve any passed seeds. Feces samples were divided into 10 g subsamples and then crushed and sifted over a 500 micrometer sieve. Recovered yellow starthistle seeds were combined into one set and stored until used for germination and viability tests.

Goat handling

Five female goats were group penned for five days at the University of Idaho Sheep Research and Teaching Center. The number of animals and seeds used in this study compare to numbers used in other similar studies (see Lacey et al. 1992; Wallander et al. 1995). These goats were fed grass pellets in excess with small amounts of rolled barley twice a day (6 AM and 6 PM)**.** The goats were then individually penned in concrete-floored pens (150 x 190 cm) two days before seed dosing. The goats were bedded with sheets of plywood, and water was always present for each animal. Each goat was fed as many grass pellets as it would consume for forty-five minutes at 6:00 AM and 6:00 PM for two days before they were fed the yellow starthistle seeds. Rolled barley (150 g) was mixed with a small amount of molasses and was fed in addition to the grass pellets for two days before seed dosing. At 6 PM on 29 Oct 08, each of the five goats were fed 2000 yellow starthistle seeds (15% of these were unplumed) that were mixed with molasses and barley. Each goat was carefully observed to make sure that the majority of seeds were consumed (greater than 95%). Each goat consumed all of the barley-molasses mix, and it appeared that all seeds were ingested.

Feces collection and seed recovery

Following the seed dosing, feces were collected twice daily (6 AM and 6 PM) for eight days. The 24-hour collection (AM and PM combined for each day) of feces from each animal was weighed fresh and recorded daily. One hundred grams of feces from each goat for each day were stored in cool conditions for subsequent examination.

A seed remover was used to wash feces samples. Fifty grams of feces were washed at a time over a stacked coarse (1.18 mm) sieve and fine (500 micron) sieve. The seed remover spun each sample over the sieves while rinsing with water. After each sample was washed, the seed remover was rinsed with water to prevent contamination of other samples with any seeds that may have become caught inside of the remover (see Kovach et al. 1988 for a description of the seed remover). Washed fecal material was placed in a paper filter pouch labeled for each day from each goat. Each sample was then dried at 30°C overnight in a forced-air oven. This occurred promptly as feces were being collected. Dried samples were sifted lightly over a 500 micron sieve and then examined under a magnifying lens for seeds. Each sample was examined twice. Morphology of each recovered seed (plumed or unplumed) was noted. Recovered seeds were stored in paper packets in the dark at room temperature until tested for germinability and viability.

To examine the efficiency of the seed recovery methods, five test runs, consisting of 20 control seeds mixed with 50 grams of seed-free fecal material, were conducted, beginning with washing through the seed remover and ending with recovery of seeds under a magnifying lens. The average seed recovery rate was 88% with 19, 17, 18, 15, and 19 seeds recovered, respectively for each test run.

Germination and viability tests

Fifty control seeds (42 plumed, 8 unplumed) were rinsed with de-ionized water and placed on moistened blotter paper in covered petri dishes. The blotter paper was kept completely moist so that there was a slight film of moisture on top of the blotter paper. Seeds were allowed 21 days at room temperature for germination (see Roché 1965; Joley et al. 1992). Yellow starthistle has been found to possess no after-ripening requirements (Young et al. 2005). Seeds were considered germinated when the radicle was 2 mm long (see Joley et al. 1992) and were then removed from the petri dishes. After 7 days, 82% of the control seeds germinated; 86% germination was achieved by 21 days. Fungi appeared in the petri dishes after about one week. Other test germinations using several different rates of a bleach solution did not improve germination or eliminate the fungi for this study.

Based on control germinations, seeds recovered from feces were rinsed with de-ionized water and placed on moistened blotter paper in covered petri dishes. After seven days, ungerminated seeds

were tested for viability using a 0.1% tetrazolium solution (see Peters 2000). Seeds with embryos that stained completely red were considered viable. Control seeds tested 100% viable.

Recorded results

Total number of seeds recovered from each goat for each day was estimated by multiplying the number of seeds collected per 100 g of feces by the total fecal output (g). These values were then used to obtain the average number of seeds passed each day and the average total percent recovered. A value that corrected for efficiency of seed recovery methods was calculated by dividing the average total percent recovered by 88% (average seed recovery in the test recovery trials). Due to the low recovery, seeds from all goats and days were combined for the germination and viability tests.

<div align="center">Results and Discussion</div>

We recovered 2.7% ± 2.1of yellow starthistle seeds fed to goats, or 3.1% of fed seeds when correcting for the efficiency of seed recovery methods. Most seeds were recovered within the first three days after ingestion (Fig. 3.1). No seeds were recovered on Day 5, but very small numbers were recovered on Days 6, 7, and 8. One unplumed seed from Day 4, or 2.4% of all recovered seeds, germinated, no other seeds germinated or tested viable.

Relatively greater numbers of yellow starthistle seeds were recovered from the goat feces that were collected from Bentz Ridge. These seeds also had higher viability than the yellow starthistle seeds in the controlled recovery study. An average of 6 yellow starthistle seeds were recovered per 10 g of dried feces, and about 8.5% of these seeds germinated or tested viable.

This low recovery and viability of ingested yellow starthistle seeds in this study is comparable to the recovery and viability of ingested seeds of other plant species used in similar studies. For example, 11 and 4% of spotted knapweed seeds fed to sheep were recovered from feces in two trials (Wallander et al. 1995), and no common crupina (*Crupina vulgaris* Cass) seeds fed to sheep were recovered while 3% of common crupina seeds fed to deer were recovered (Thill et al. 1986). After leafy spurge seeds were ingested by goats and sheep, 18% of all seeds ingested were recovered (Lacey et al. 1992). Common crupina seeds recovered from cattle, horse, and deer feces had viability that ranged from 2 to 7% (Thill et al. 1986). Viability of recovered spotted knapweed seeds from sheep feces ranged between 0 and 26% compared to control seeds that had 98 and 88% viability (Wallander et al. 1995). Viability of leafy spurge seeds recovered from sheep feces was 5% compared to 42% viability of seeds collected directly from plants (Olson et al. 1997b). However, seed

survivability can be as high fifty percent (Janzen 1984) and depends on both plant and animal attributes (see Chapter 1).

Forty-four percent of recovered seeds were unplumed. This relatively high rate of recovery for unplumed seeds compared to plumed seeds in relation to the number of each type fed may be due to the presence of the pappus. The pappus attachment site may be a weak point in the seed coat and thus more plumed seeds may be destroyed by digestion than unplumed seeds although recovery rate for either seed type is still extremely low.

From this study, we can conclude that passage through the goat digestion system substantially reduces yellow starthistle seed germinability and viability as control seeds had germinability and viability greater than 92%. In this seed feeding study, germination only occurred from one seed passed on Day 4, and passage rate cannot be determined of seeds recovered from feces collected in the field, so we cannot determine if increased time spent in the digestive tract influenced viability of seeds. However, several studies have reported that seeds can rapidly lose viability with increased retention time (e.g., Thill et al. 1986; Simao Neto et al. 1987; Blackshaw and Rode 1991). Research is also fairly conclusive that most viable seeds regardless of ruminant species are passed within seven days after being ingested with peak passage typically occurring two to four days after ingestion (e.g., Piggin 1978; Simao Neto et al. 1987). Common crupina seeds fed to sheep, cattle, horses, and deer were recovered within five days of ingestion (Thill et al. 1986). Viable leafy spurge seeds that were ingested by goats and sheep were recovered in feces within five days (Lacey et al. 1992), and viable spotted knapweed seeds that were ingested by sheep were recovered within seven days (Wallander et al. 1995). One exception to this was observed with mule deer where viable spotted knapweed seeds were recovered from every day of the 10-day period that feces were collected (Wallander et al. 1995).

Management Implications

Benefield et al. (2001) recommended that late-season control of yellow starthistle occur before 2% of the spiny heads begin flowering because as flowering progresses, seed maturation and production increase exponentially. Yellow starthistle seeds are viable as early as five days following the onset of flowering (Benefield et al. 2001). Based on our research, we suggest that a four-day holding period for goats that have been grazing yellow starthistle at this stage of yellow starthistle phenology or later should be adequate to eliminate risk of seed spread through feces. Cattle and sheep will also readily graze yellow starthistle, but cattle and sheep avoid plants that have fully developed

spines and thus will only consume yellow starthistle in earlier phenological stages when few or no seeds are viable. This makes risk of yellow starthistle spread through cattle or sheep feces relatively low and consequently no holding period is needed.

Research on the effects of ruminant digestion on seeds of many key weeds is still lacking, but existing research indicates that a seven-day holding period should be the maximum amount of time needed to prevent weed seed spread. However, this holding time may be substantially shorter for livestock that have been grazing some weed species. For instance, viable leafy spurge seeds were found to be passed in sheep feces only on the first two days after ingestion (Lacey et al. 1992), and viable common crupina seeds were only recovered from cattle feces in the first three days after ingestion and none were recovered at all from sheep feces (Thill et al. 1986). Because research has not demonstrated solid guiding principles for predicting seed survival, it may be beneficial from a livestock management standpoint to know accurate passage rates of viable seeds for livestock that have been grazing specific weeds.

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Table 3.1. A list of studies that examined the effects of digestion by cattle, goats, and sheep on seed viability and germinability of some major weeds in the United States.

Animal	Plant species	Results	Authors
Cattle	Cheatgrass (*Bromus tectorum*)	Control[1]: 98% viable[2]. Treatment: 0% viable.	Blackshaw and Rode 1991[3]
	Common crupina (*Crupina vulgaris*)	Control: 95% germinable Treatment[4]: 7% recovered *and* germinable. Last day of viable recovery[5]: 3.	Thill et al. 1986
	Perennial pepperweed (*Lepidium latifolium*)	Control: 3.4% germinated. Treatment: 59%	Carpinelli et al. 2005[6]
Goats	Leafy spurge (*Euphorbia esula*)	Control: 90% viable. Treatment: 31% *of* recovered seeds were viable. Last day of viable recovery: 5.	Lacey et al. 1992
	Sulfur cinquefoil (*Potentilla recta*) mature seeds	Control: 75% Treatment: 31% recovered *and* viable Last day of viable recovery: 3.	Frost, unpubl. data
	Yellow starthistle (*Centaurea solstitialis*)	Control: 92% viable Treatment: 2.7% *of* recovered seeds were viable. Last day of viable recovery: 4.	See this chapter.
Sheep	Cheatgrass (*Bromus tectorum*)	Control: 90% germinable Treatment: 2.2% recovered *and* germinable. Last day of viable recovery: 8.	Lehrer and Tisdale 1956
	Common crupina (*Crupina vulgaris*)	Control: 95% germinable. Treatment: 0% recovered.	Thill et al. 1986
	Leafy spurge (*Euphorbia esula*)	Control: 90% viable. Treatment: 14% *of* recovered seeds were viable. Last day of viable recovery: 2.	Lacey et al. 1992
	Medusahead (*Taeniatherum caput-medusae*)	Control: 99% germinable. Treatment: 2.4% recovered *and* germinable. Last day of viable recovery: 9.	Lehrer and Tisdale 1956
	Spotted knapweed (*Centaurea stoebe*)	Control: 98% germinable. Treatment: 0-26% *of* recovered seeds were viable. Last day of viable recovery: 7. (Trial 1)	Wallander et al. 1995
	Sulfur cinquefoil (*Potentilla recta*; mature seeds)	Control: 75% viable. Treatment: 46% recovered *and* viable. Last day of viable recovery: 3.	Frost et al. unpubl

[1]Seeds tested for germinability or viability before the digestion treatment. [2]Includes germinated seeds and ungerminated seeds that tested positive with a tetrazolium viability test. [3]Seeds were placed directly into the rumens of fistulated cattle for 24 hr and then removed and tested for germinability or viability. [4]Seeds were ingested by ruminants and then recovered from feces. [5]The last day following ingestion that any germinable or viable seeds were recovered from feces. [6] Seeds placed in the rumen were removed after 24 hr.
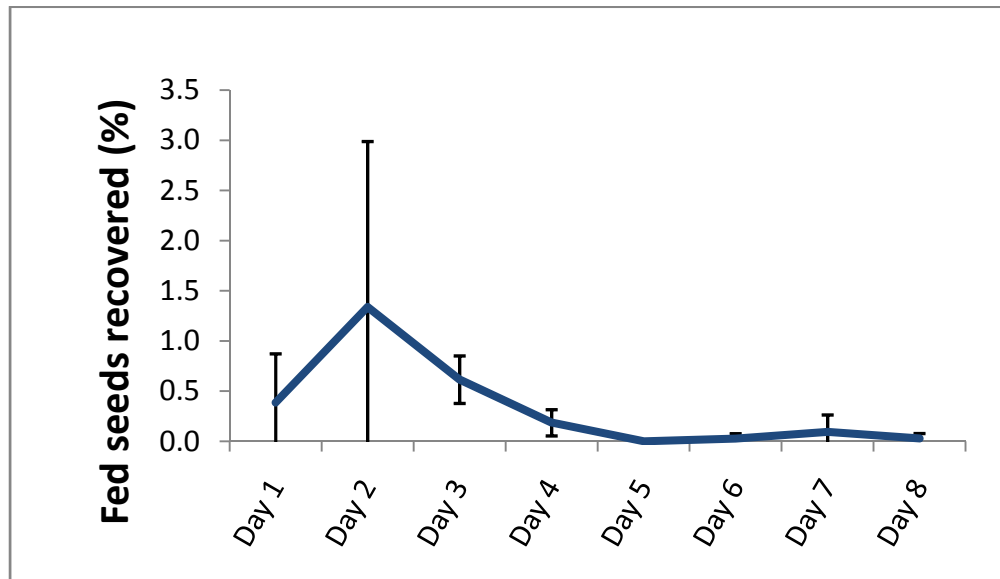
Figure 3.1. Mean percent with standard error bars of yellow starthistle seeds that were recovered from goat feces over an 8-day feces collection period.

Appendix A: Study area for research examining the effects of targeted grazing by goats on yellow starthistle in northern Idaho.

Appendix B: Density (mean ± SE) of yellow starthistle plants and seedheads per m^2 in a three year grazing study on a site near White Bird, Idaho. Grazing treatment was applied across the study site (grazed) while ungrazed exclosures served as controls (ungrazed).

Time	Treatment (Ungrazed or Grazed)	Yellow starthistle plants	Yellow starthistle seedheads
After grazing 2006	Ungrazed	117.9 ± 10.9	328.4 ± 21.4
	Grazed	54.0 ± 6.2	26.9 ± 3.1
After grazing 2007	Ungrazed	203.5 ± 13.5	106.0 ± 9.2
	Grazed	56.5 ± 4.9	1.04 ± 0.26
After grazing 2008	Ungrazed	81.8 ± 4.2	103.7 ± 7.7
	Grazed	33.9 ± 3.1	6.2 ± 1.4
Before grazing 2007	Ungrazed	242.1 ± 16.0	109.0 ± 8.1
	Grazed	139.8 ± 11.0	113.9 ± 9.7
Before grazing 2008	Ungrazed	77.7 ± 5.2	81.3 ± 6.1
	Grazed	52.4 ± 4.9	75.0 ± 6.4

Appendix C: Canopy cover (mean ± SE) of yellow starthistle, grass, and forbs in a three year grazing study on a site near White Bird, Idaho. Grazing treatment was applied across the study site (grazed) while ungrazed exclosures served as controls (ungrazed).

Time	Treatment (Ungrazed or Grazed)	Yellow starthistle	Grass	Forbs
After grazing 2006	Ungrazed	43.9 ± 3.3	11.5 ± 1.0	18.3 ± 1.4
	Grazed	29.5 ± 2.5	17.6 ± 1.8	17.7 ± 1.3
After grazing 2007	Ungrazed	25.4 ± 1.2	20.3 ± 1.6	9.8 ± 0.8
	Grazed	5.9 ± 0.6	20.9 ± 1.7	2.4 ± 0.3
After grazing 2008	Ungrazed	19.4 ± 1.0	16.5 ± 1.0	11.2 ± 0.7
	Grazed	5.3 ± 0.5	17.2 ± 1.0	8.6 ± 0.5
Before grazing 2007	Ungrazed	37.0 ± 1.6	22.8 ± 2.0	15.8 ± 1.4
	Grazed	25.7 ± 1.4	33.6 ± 2.2	14.3 ± 1.2
Before grazing 2008	Ungrazed	16.6 ± 0.9	34.2 ± 1.7	18.8 ± 1.0
	Grazed	12.2 ± 0.9	33.3 ± 1.6	21.5 ± 1.0