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WILIDLIFE RESEARCH REPORT

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EXTENDED ABSTRACT

The Columbian sharp-tailed grouse (CSTG, *Tympanuchus phasianellus columbianus*) is one of six subspecies of sharp-tailed grouse in North America. Historically its distribution ranged from the northwest in British Columbia in the northwest to Colorado in the southwest. Isolated populations exist (or formally existed) in Washington, Idaho, Wyoming, Colorado, Montana (extirpated), Utah, Nevada (reintroduced) and Oregon (reintroduced), with CSTG currently occupying 10% of its former range. Habitat loss and degradation from anthropogenic activities are cited as the primary reasons for the decline in CSTG, with the conversion of native shrub plant communities to agricultural production being the most prevalent habitat impact. The United States Fish and Wildlife Service (USFWS) has been petitioned twice to list the CSTG for protections under the Endangered Species Act and concluded that the CSTG was not warranted for listing following both petitions. The ESA listing decision was, in part, not warranted because of CSTG range expansion facilitated by Conservation Reserve Program (CRP) in 1985 and subsequent reauthorizations. In Colorado a preponderance of plantings were seeded to intermediate wheatgrass (*Agropyron intermedium*), smooth brome (*Bromus inermis*), and occasionally included alfalfa (*Meticago sativa*). These mixes resulted in mature herbaceous stands of grass that provide marginal benefits to CSTG. In contrast, mineland reclamation sites in northwest Colorado have been shown to be beneficial to CSTG and provide high quality spring-summer-fall habitat to CSTG when compared to CRP or native rangeland. Mineland reclamation provides sufficient quality to support favorable demographic rates for females when compared to CRP. Thus, based on past observational research, and that some existing CRP habitats are not occupied by CSTG, there is building evidence that habitat improvements could improve existing or expired CRP. This has resulted in management recommendations to improve CRP for CSTG. Ecological theory supporting habitat improvements (quality) through wildlife habitat enhancement and/or management has been a long established tenet of wildlife management, but the

wildlife-habitat relationship is complex. CSTG provide an opportunity to evaluate demographic rates and population growth in relation to changes in habitat quality. CSTG are a highly productive, generalist species that have centralized breeding locations and have limited movements during the breeding season with relatively small home ranges.

Our overall research objective is to ascertain the demographic and population response of CSTG to improvements in habitat quality by increasing floristic horizontal and vertical structure and species richness in monotypic stands of non-native grasses. The goal of our research is to conduct treatments (habitat improvements) in two lek complexes (T1 and T2). A Before-After Control-Impact (BACI) design with paired controls is employed. Our study area is located in northwestern Colorado, specifically in southwestern Routt and southeast Moffat counties. Our study area is predominantly (70%) privately owned by individuals or mining companies and is interspersed with Bureau of Land Management and State Land Board properties (Fig. 2). Working cooperatively with the Northwest Region Terrestrial Habitat Coordinator (NWRTHC), we identified and finalized treatment areas working cooperatively with private landowners, the Natural Resources Conservation Service (NRCS), and the Farm Services Agency (FSA). Although we initially outlined treatments to be conducted in one year, FSA vegetation manipulation restrictions for mid-contract maintenance of the properties enrolled in CRP will prevent such an approach. Maintenance requirements differ for enrolled fields that are at a 65 ha threshold. For enrolled fields < 65 ha, we can only treat 50% of the field in year 1 and the remainder in year 2. For fields > 65 ha, we can only treat 33% of a field in year 1, 33% in year 2, and 33% must remain untreated. Thus, in Treatment Area 1 (Fig. 2), we will treat 140 ha in 2016 and 140 ha in 2017. In Treatment Area 2 (Fig. 2), we will treat 202 ha in 2016 and 202 ha in 2017. Although there are numerous vegetation manipulation approaches to reduce non-native grass cover and increase plant species richness, we identified the following protocol to implement habitat treatments. First, during late-summer (after nest hatch), we will initiate treatments with mechanical tillage equipment (off-set disc) to reduce viable non-native perennial grass cover and assist with seed-bed preparation. Second, approximately 2 - 4 weeks after mechanical tillage, we will treat sites with a chemical aerial application of Plateau® and glyphosate to reduce non-native perennial grass and limit annual and perennial grass seed germination. We may need to treat with a second application of glyphosate. Lastly, in late-fall, we will drill a seed mixture of native and non-native grasses, forbs, and shrubs (Table 1) with a no-till drill.

We captured female CSTG in the spring using walk-in funnel traps in the morning on dancing grounds. Trapping occurred on dancing grounds in three study sites in Moffat county (T1, T2, C3) and leks ranged in size from 10 – 45 males. We also trapped at dancing grounds at one study site in Routt county (C2) that ranged in size from 6 – 24 males. We fitted females with 15 g elastic necklace-mounted radio transmitter equipped with a 12-hour mortality circuit having an 8.5 month nominal battery life. We monitored movements every 1-3 days with hand-held Yagi antennas attached to a receiver. When monitoring revealed a successful hatch, we attempted to capture all chicks in the brood within 24 hours. We randomly selected 4 chicks/brood and fit a 0.55 g backpack style transmitter using sutures along the dorsal midline between the wings (Fig. A-3). We captured juveniles when they reached 20-23 days-of-age at approximately two hours before sunrise while juveniles are brooding with the female. We removed chick transmitters and replaced them with a 2.4 g back-pack style juvenile transmitter (Fig. A-4). We sampled vegetation at all nest and a sample of brood sites.

In 2016 we captured 105 female CSTG (78 adults: 27 yearlings) during 9 April – 3 May (Fig. 4). We trapped on 8 dancing grounds in 4 study areas (Hayden; Big Elk 1: West Axial; Moffat County Road 53 and Temple; Iles Dome; Iles Dome 2, 3, and 4: Trapper; Trapper Mine 1, and 7). Adult and yearling female mass ($\bar{x} \pm$ SE) was 671.3 ± 5.2 g (n = 78) and 620.8 ± 8.9 g (n = 27), respectively. Female mass appears to vary by study area (Fig. 5), by age, and spatially (Fig. 6). From April through September 2016, we documented 31 and 6 adult and yearling female mortalities resulting in a 6-month adult female survival rate of 0.52 ± 0.05 (n = 100; 95% CI 0.43 - 0.61) and a yearling survival rate of 0.49 ± 0.01 (n = 27; 95% CI 0.33 - 0.65) (Fig. 7). Female survival appeared similar between 2015 (0.62 ± 0.01 (n = 107; 95% CI 0.52 - 0.72) and 2016 (Fig. 10). We documented an overall nest initiation rate of 95% (n =

74/78) and 96% (*n* = 21/22) for adult and yearling females, respectively. We documented an overall 54.4% (*n* = 56/103) and 59% (n = 56/95) apparent nest and female success, respectively. Seven females renested once yielding 42.9% (n = 3/7) nest success and 1 female successfully nested on a second renest attempt. Female movement in 2016 from the lek of capture to nest averaged 2.42 ± 0.48 km (*n* = 94; range 0.17 - 35.0 km) (Fig. 11). Seventy percent (*n* = 66/84) of the nests were located within 2 km of the lek of capture (Fig. 11). We captured 391 chicks from 56 broods with an overall mean mass of 15.5 ± 0.1 g (range 11.8 - 23.1) and the average age of broods at chick capture was 2.4 ± 0.1 days (range 2 - 6 days). A majority of chicks (96%; *n* = 376/391) were captured 1 - 3 post-hatch and included 93% (*n* = 52/56) of the broods. Thus, the mean mass for chicks from 1-3 days-of-age was 15.2 ± 0.1 g (range 11.8 - 20.0). There was a clear shift in chick mass with chicks being heavier in 2016 compared to 2015, with no chicks smaller than 12 g. We radio-marked 211 chicks resulting in an average number of chicks marked/brood of 3.8 chicks. We also PIT tagged 172 chicks. The average brood size at marking was 7.7 chicks (range 3 - 14). We recaptured and/or marked 119 juveniles that averaged 27 days post-hatch (range 18 – 53 days post-hatch). Juvenile mean mass was 112.0 ± 5.0 g (range 36.0 - 404.0 g). Nineteen of the juveniles were not previously marked.

At the writing of this report, data entry and proofing is continuing. Field data collection will continue in 2017.

WILDLIFE RESEARCH REPORT

COLUMBIAN SHARP-TAILED GROUSE DEMOGRAPHIC RESPONSE TO HABITAT IMPROVEMENTS

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INTRODUCTION

The Columbian sharp-tailed grouse (CSTG, *Tympanuchus phasianellus columbianus*) is one of six subspecies of sharp-tailed grouse in North America (Connelly et al. 1998). Historically its distribution ranged from British Columbia in the northwest to Colorado in the southwest (Aldrich 1963, Miller and Graul 1980). Isolated populations exist (or formally existed) in Washington, Idaho, Wyoming, Colorado, Montana (extirpated), Utah, Nevada (reintroduced) and Oregon (reintroduced) (Bart 2000, Hoffman et al. 2015), with the species currently occupying 10% of its former range (U.S. Department of the Interior 2000). Habitat loss and degradation from anthropogenic activities are cited as the primary reasons for the decline of CSTG (Yocom 1952, Giesen and Braun 1993, McDonald and Reese 1998, Schroeder et al. 2000), with the conversion of native shrub plant communities to agricultural production being the most prevalent habitat impact.

The United States Fish and Wildlife Service (USFWS) has been petitioned twice to list the CSTG for protections under the Endangered Species Act and concluded that the CSTG was not warranted for listing following both petitions (U.S. Department of the Interior 2000, 2006). ESA listing was, in part, not warranted because of CSTG range expansion facilitated by Conservation Reserve Program (CRP) in 1985 and subsequent reauthorizations. CSTG have increased in distribution and densities primarily in Idaho, Utah, and Colorado (U.S. Department of the Interior 2000) and the USFWS concluded that these increases secured the larger metapopulations of CSTG and thus, the CSTG was not at risk of extinction. The CSTG (Mountain Sharp-tail) is a game species in Colorado, and is designated as a Tier 1 species of greatest conservation need in the Colorado State Wildlife Action Plan (Colorado Parks and Wildlife 2015). There have been efforts to increase the range of CSTG through reintroductions into vacant habitat in Oregon and Nevada. Additional reintroduction efforts have occurred within Utah and Colorado to expand its range into historic vacant suitable habitat (Colorado: Dolores, Eagle, and Grand counties).

The CSTG historically inhabited, and currently inhabits where available, native big sagebrush (*Artemisia tridentata spp.*) mountain shrub, and shrub-steppe communities in western North America (Connelly et al. 1998). By the mid-1950s to mid-1960s many of the native sagebrush communities on private land were converted to agricultural production (Braun et al. 1976). These practices continued into the mid-1980s until the 1985 Farm Bill provided an opportunity for private landowners to enroll highly erodible lands into the CRP and remove these agricultural lands from production (Negus et al. 2010). Since the goal was to stabilize erodible soils, many CRP planting seed mixes included only two or three plant species (Boisvert 2002, Negas et al. 2010). Generally, CRP fields provide breeding, summer, and fall habitat for CSTG in the western United States (Sirotnak et al. 1991, Apa 1998, Hoffman 2001, Rodgers and Hoffman 2005, Gorman and Hoffman 2010, Stinson and Schroeder 2012, Hoffman et al. 2015), but do not provide substantial winter habitat (Schneider 1994, Ulliman 1995).

In Colorado a preponderance of plantings were seeded to intermediate wheatgrass (*Agropyron intermedium*), smooth brome (*Bromus inermis*), and occasionally included alfalfa (*Meticago sativa*) (Hoffman 2001, Hoffman et al. 2015). These mixes resulted in mature herbaceous stands of grass that provide marginal benefits to CSTG (Hoffman et al. 2015). Some CRP plantings in Idaho were sufficiently diverse to support CSTG (Apa 1998) and facilitate range expansion (Mallett 2000). In Washington, some CRP fields were so small in size, McDonald (1998) hypothesized that these stands could act as ecological traps (Gates and Gysel 1978, Best 1986) for nesting CSTG females. There are concerns that aging CRP fields are of reduced quality and may reduce the production and survival of

CSTG (Boisvert 2002, Gillette 2014, Hoffman et al. 2015). Many CRP fields in Colorado and elsewhere once supported high quality habitat, but more recently have declined in quality (Negus et al. 2010).

In contrast, mineland reclamation sites in northwest Colorado have been shown to be beneficial to CSTG and provide high quality spring, summer, and fall habitat to CSTG when compared to CRP (Boisvert 2002) or native rangeland (Collins 2004). Reclaimed mineland fields provide sufficient quality to support favorable demographic rates for females when compared to CRP. Boisvert (2002) reported that the 282-day post-capture female survival rate in reclaimed mineland habitat was two times higher than survival of females captured in CRP. In addition, females that inhabited CRP had >11 times higher proportional hazards mortality risk than females in reclaimed mineland habitat. Boisvert (2002) also reported higher productivity of CSTG using reclaimed mineland habitat; nest success was nearly five times higher for females nesting in reclaimed mineland habitat when compared to CRP. In addition, Boisvert (2002) reported that chick mortality was higher for females that inhabited native shrubland communities and CRP when compared to females in reclaimed minelands. Boisvert (2002) concluded that CRP and upland shrub habitats likely were deficient in quality brood-rearing resources (e.g. forbs).

Although CRP fields do not provide all the life requisites for CSTG (e.g. winter habitat; Connelly et al 1998, Schneider 1994, Ulliman 1995), and CRP provides only marginal benefits to CSTG in Colorado (Boisvert 2002) and Idaho (Gillette 2014), CRP is substantially better than fields in active agricultural production (Sirotnak et al. 1991, Mallet 2000, Hoffman 2001, Boisvert 2002, Gillette 2014). This is because CRP replaced agricultural crops with perennial grasses and forbs, effectively linking native sagebrush communities between private and public land. These larger functioning landscapes provide generalist species like the CSTG (Apa 1998) suitable habitat (Hoffman 2001, Rodgers and Hoffman 2005) on a large scale.

Thus, based on past observational research, and that some existing CRP habitats are not occupied by CSTG, there is building evidence that habitat improvements could improve existing or expired CRP. This has resulted in management recommendations to improve CRP quality (Hoffman 2001, 2015, Boisvert 2002, Gillette 2014, Hoffman et al. 2015) by improving plant diversity in existing CRP (1-2 grass and < 3 forb species) that currently provides low quality CSTG nesting and brood-rearing habitat. Habitat improvements (adding legumes and bunchgrasses) would enhance CSTG habitat quality and suitability and could improve population productivity and growth (Gillette 2014). Habitat improvements could also counteract losses in CRP due to contract conclusion and an overall reduction of CRP (Gillette 2014, Hoffman et al. 2015) or mitigate other potential threats (energy development; Hoffman et al. 2015).

Ecological theory supporting enhancement and management of wildlife habitat quality has been a long established tenet of wildlife management (Leopold 1933, Dassman 1964), but the wildlife-habitat relationship is complex (Morrison et al. 2006). The understanding of the wildlife-habitat relationship is constantly evolving through defining and assessing habitat quality as it relates to population growth rates, density, and demographic rates (Van Horne 1983, Knutsen et al 2006, Johnson 2007). This is especially true when attempting to couple the intended or unintended changes in habitat quality with the mechanisms inherent with wildlife population change, especially with avian species (Marzluff et al. 2000).

The assessment of habitat quality in relation to avian species is a complex question and an issue of concern for wildlife and habitat managers (Marzluff et al. 2000). Knutson et al. (2006) reviewed approaches to assess habitat quality and suggested that estimates of abundance, food availability, nest survival, annual productivity, and annual survival (see Knutson et al. 2006 for citations) should be included as indicators of habitat quality. Additionally, home range size has been shown to be inversely related to habitat quality (Cody 1985), but Knutson et al. (2006) concluded that there is no single indicator of habitat quality. Johnson (2007) furthered recommendations by Franklin et al. (2000) and suggested that several possible indicators of habitat quality should be assessed because if only one parameter is used it could lead to misrepresentations of habitat quality (e.g. density; Van Horne 1983). Therefore, when attempting to link habitat-specific measurements of quality to the performance or productivity of birds, research should address demography (Johnson 2007) in an effort to hypothesize a causal link between a demographic population response and a change in habitat quality (Block and Brennan 1993, Hall et al. 1997, Knutson et al. 2006, Johnson 2007).

Although it would be desirable to experimentally manipulate as many mechanisms as possible that influence demography, it is financially and logistically impractical. Thus, it could be advantageous to experimentally manipulate a minimal number of mechanisms (e.g. nest sites, food) and gain a thorough understanding of these and then use observation and future research to infer the remaining suite of mechanisms (Marzluff et al. 2000; Fig. 1). Improved understanding of the mechanistic relationship between habitat quality and demography will lead to more accurate predictions of the population-level effectiveness of habitat management programs (Raphael and Maurer 1990, Marzluff et al. 2000; Fig 1.).

We define wildlife habitat quality as "the ability of the environment to provide conditions appropriate for survival, reproduction, and population persistence" (Block and Brennan 1993:38). Johnson (2007) suggests that habitat quality is best described and defined at the perspective of the individual as the per capita rate of population change for a given habitat. Thus, abundance, reproduction and survival are the most efficient measures to assess habitat quality (Virkkala 1990, Homes et al. 1996, Franklin et al. 2000, Murphy 2001, Persson 2003, Knutson et al. 2006, Johnson 2007). Specifically, since survival and reproduction directly influence a population growth rate (λ), Sæther and Bakke (2000) suggest that λ is also an important parameter to assess habitat quality, especially in single species management (Williams et al. 2002, Johnson 2007). Williams et al. (2002) also suggested that nest survival and annual production (chicks/female) could be used to assess habitat quality and are useful tools when evaluating population growth change in prospective or retrospective analyses (Sæther and Bakke 2000).

CSTG provide an opportunity to evaluate demographic rates and population growth to assess changes in habitat quality. CSTG are a highly productive, generalist species (Apa 1998) that have centralized breeding locations and have generally limited movements during the breeding season (Boisvert t al. 2005) with relatively small home ranges that have a median size of 65 - 113 ha and 69 - 75 ha in spring-fall and brood-rearing habitat, respectively (Collins 2004). Boisvert (2002) reported similar home ranges sizes, with smaller median home range size in reclaimed minelands (75 ha) when compared to CRP (112 ha). These life history traits and relatively small movements facilitate a relatively rapid response to habitat management, providing managers and researchers an opportunity to work collaboratively to investigate a mechanistic response to landscape level habitat quality improvements.

To evaluate the demographic response of CSTG to habitat improvements, rigorous estimates of adult female survival and production (Sæther and Bakke 2000) are needed. Although techniques to estimate female survival are well established using VHF radio telemetry (McDonald 1998, Boisvert 2002, Collins 2004, Gillette 2014), elasticity analysis suggests that the population growth rate may be less sensitive to adult survival rate in "highly productive" species (Sæther and Bakke 2000). Thus, obtaining rigorous estimates of the temporal variation in chick and juvenile survival are necessary to support future management recommendations (Sæther and Bakke 2000).

A standard for estimating CSTG chick survival from hatch to 4–7 weeks post-hatch has involved flush counts or observing female behavior. Flush counts to estimate productivity from 35–49 days post-hatch and brood survival (McDonald 1998, Boisvert 2002, Collins 2004, Gillette 2014) have been conducted, but Collins (2004) acknowledged biases (e.g. detectability) associated with flush counts. In an effort to minimize biases associated with flush counts, Collins (2004) attempted to improve detectability by incorporating and pairing flush counts using hunting dogs. Unfortunately, these approaches can lead to imprecise estimates of chick survival because of unknown detection probabilities associated with cryptic chicks combined with a no movement defensive strategy to avoid detection. Chick survival estimates can also be biased by chick exchange between broods, as has been observed with greater sage-grouse (*Centrocercus urophasianus*) (Dahlgren et al. 2010, Thompson 2012).

To obtain a more reliable estimate of chick survival, our field methods will include the use of VHF micro transmitters attached to day-old chicks to obtain survival estimates using techniques established with greater sage-grouse, Gunnison sage-grouse (*C. minimus*) and plains sharp-tailed grouse

(*T. p. jamesi*) (Burkepile et al. 2002, Manzer and Hannon 2007, Dahlgren et al. 2010, and Davis 2012, Thompson et al. 2015), and more recently with CSTG (Apa 2014).

OBJECTIVES

Our overall research objective is to ascertain the short- and long-term demographic and population response of CSTG to improvements in habitat quality by increasing floristic horizontal and vertical structure and species richness in monotypic stands of non-native grasses. Specific objectives are to:

1. Ascertain the current baseline (before impact) demographic (age specific survival, nest success) and spatial (home range and movements) parameters in existing non-native grass dominated communities (controls and treatments sites).
2. Ascertain the short-term (2 year) post-habitat enhancement, demographic (age specific survival, nest success), and spatial (home range and movements) parameters in non-native grass dominated communities and compare with treated sites.

STUDY AREA

The study area is located in northwestern Colorado, specifically in southwestern Routt and southeastern Moffat counties (Fig. 2). The area is further described by Boisvert (2002) and Collins (2004). The study area is predominantly (70%) privately owned by individuals or mining companies and is interspersed with Bureau of Land Management and State Land Board properties (Hoffman 2001).

The landscape cover types that contribute to CSTG breeding and summer habitat were historically sagebrush-grass or mountain shrub communities but currently have a grassland cover type created by the CRP. Elevations range from 2,000-2,600 m with soils ranging from silt and clay loams 8-150 cm deep (Boisvert 2002, Collins 2004). Daily temperatures range from 5-25 ^{o}C and average annual precipitation varies by elevation, but ranges from 50 cm near Steamboat Springs to <25 cm near Craig (Boisvert 2002, Collins 2004).

METHODS

Survival and Productivity

Grouse Spring Capture – We captured female CSTG using modified walk-in funnel traps (Schroeder and Braun 1991) in the morning on dancing grounds. Trapping occurred on dancing grounds in three study sites in Moffat county (T1, T2, C3; Fig. 2) that had leks ranging in size from 10 – 45 males. Trapping also occurred on dancing grounds in two study sites in Routt county (C1, C2; Fig. 2) that had leks ranging in size from 6–24 males. We opened traps one-half hour before sunrise and closed or blocked trap openings at the cessation of trapping each morning. We timed trapping to coincide with the peak of female attendance (Giesen et al. 1982, Giesen 1987, R. Hoffman, personal communication).

We fitted each captured female with 15 g elastic necklace-mounted radio transmitter (Model RI-2BM, Holohil Systems, Ltd., Carp, Ontario) equipped with a 12-hour mortality circuit having an 8.5 month nominal battery life. The transmitter mass is < 3% (range 2.0 – 2.9%) of an adult or yearling female body mass. We bent the 16 cm antenna down the back to lie between the wings and down the back of the grouse. We classified captured grouse by sex (Snyder 1935, Henderson et al. 1967) and age (Ammann 1944). We aged females as yearling or adult by examining the condition of the outer primaries (Ammann 1944) and we measured mass (± 1 g) by placing a restrained individual in a cotton bag and weighing it on an electronic balance.

We fitted all females with individually numbered aluminum leg bands (size 12) attached on the tarsus. When time and logistics allowed, we banded males and released them after capture. We collected two feathers that included the shaft and placed them in a paper envelop before storing them in a freezer.

We processed individuals and released them at the point of capture. When releasing birds, we quickly and quietly backed away until the bird walked, ran, or flushed away.

Nest Monitoring and Chick Capture – We monitored movements every 1-3 days and obtained general locations using triangulation from ≥ 30 m distance (to minimize disturbance) with hand-held Yagi antennas attached to a receiver. We obtained locations between 0800 and 1800 hours to monitor movements and determine nest initiation, location, and incubation. When a female was located in the same location for two consecutive days, we assumed nest initiation or incubation. We attempted to make visual observations of females on nests at 7-10 days post-incubation confirmation, but visibility depended on vegetation density. We monitored incubating females 2-3 times/week to monitor nest fate. We monitored nesting by using telemetry at two points at right angles from one another 30-50 m distance (25-26 day incubation period) from the incubating female.

When monitoring revealed a successful hatch (female movement away from nest), we attempted to capture all chicks in the brood within 24 hours. We located females <2 hours after sunrise in order to capture chicks while they are brooded. We flushed the female and captured chicks by hand. We confined chicks in insulated soft sided coolers equipped with hot water bottles (sufficiently large to handle 10–12 chicks) to maintain the cooler temperature between 35-38 ^{o}C. We did not attempt to capture chicks during inclement weather to reduce thermoregulation issues with chicks.

We weighed (± 0.01 g) all captured chicks using an electronic scale, and randomly selected four chicks per brood and fitted each with a 0.55 g backpack style (model A1015, Advanced Telemetry Systems, Isanti, MN) transmitter using sutures along the dorsal midline between the wings (Burkepile et al. 2002, Dreitz et al. 2011, Manzer and Hannon 2007, Thompson et al. 2015; Fig. A-3). In advance of attaching the transmitter, we swabbed the suture site with isopropyl alcohol, and inserted two sterile, 20-gauge needles subcutaneously and perpendicular to the dorsal mid-line. We threaded the monofilament suture material (Braunamide: polyamide 3/0 thread, pseudo monofilament, non-absorbable, white) through the needle barrels. We then removed the needles and tied off the suture material using a square knot and removing excess suture material. We applied one drop of cyanoacrylate glue on the knot. We monitored the brood female during brood processing to assure that she remained in the near vicinity.

We marked the remaining chicks in each brood with a passive integrated transponder (PIT) tag (Model HPT8, Biomark, Inc., Boise, Idaho). Initially we swabbed the injection site with isopropyl alcohol and then injected the PIT tags subcutaneously along the dorsal midline in the interscapular region. We scanned each PIT tag prior to insertion to confirm the unique identification number and tag functionality. A handler secured the chick, while another person injected the PIT tag (weight 30 ± 6 mg) beneath the skin on the chick's back. We sealed the insertion site using skin glue to help prevent tag loss. Marking procedures in this protocol were approved by the University of Wisconsin-Madison Institutional Animal Care and Use Committee (IACUC) (Protocol #A005282).

We determined chick and brood locations by first locating females and circling at a 25 m radius. We also identified the position (i.e., distance) of radio-marked chicks in relation to the female. We attempted to find all chicks that were separated or missing from broods to determine fate and cause of mortality and we attempted to obtain brood locations equally among four time periods: brooding (<2 hour after sunrise or before sunset), morning (08:00-11:00), mid-day (11:00-14:00), and afternoon (14:00-18:00).

We attempted to re-capture juveniles previously marked as chicks when they reached 20-23 days after hatch at approximately two hours before sunrise while juveniles are brooding with the female (Apa 2014). We circled the female and brood using radio telemetry and spotlights. Once we obtained a visual location, the female and brood were captured using a 1.5 m diameter hoop net. We restrained all captured juveniles and released the female at the point of capture to avoid injury of juveniles.

We removed the chick transmitters and replaced them with 2.4 g back-pack style juvenile transmitters (Model A1050, Advanced Telemetry Systems, Isanti, MN) (Fig. A-4). We used the same attachment method earlier described for day-old chicks (Burkepile et al. 2002, Dreitz et al. 2011, Manzer and Hannon 2007, Thompson et al. 2015, Apa 2014). We selected a new suture site near the previous suture site. We weighed all captured juveniles. We applied sulfadiazine (thermazine) water-based cream before the juvenile was released if there was any sign of irritation or infection (L. Wolfe, personal communication). The juvenile transmitter had a nominal battery life of 68-158 days months and was 3.0-4.6% of chick mass (Apa 2014). We scanned all juveniles for PIT tags. If there was no PIT present, we inserted a PIT using the same procedure previously mentioned.

We obtained aerial locations or detections for survival estimates as needed for missing birds, and locations of active transmitters will be obtained once per month throughout the research.   All trapping and handling protocol (unless otherwise noted) were approved by the CPW Animal Care and Use Committee (Permit # 02-2015).

Habitat Quality

We sampled vegetation at all nest sites and a sample of brood sites. We created a grid layer of 200 m^2 cells centering on the dancing grounds out to 2 km in each study area and then selected individual grid cells based on a spatially balanced random sample. We used this to determine sampling locations for random sites. We did not consider cells with grouse locations as part of the random sample. We used the same vegetation data collection techniques on at least one paired random location for each nest and brood site.

Sampling at nest sites was conducted as soon as logistically possible, within seven days of nesting cessation (successful or unsuccessful). We sampled paired random site vegetation sampling within seven days of its paired sample. We placed four, 10-m transects in the cardinal directions intersecting at the nest bowl. We located females with broods 1-2 times per week. We circled females and broods, and we used the intersection point of flags placed in the cardinal directions to identify the center of the brood location which will determine the intersection point of the transect. We conducted habitat measurements at as many brood locations as possible with equal sampling across individuals to retain sample independence and avoid sampling autocorrelation issues.

When present, we sampled overstory shrub canopy cover (foliar intercept) by lowest possible taxa using line-intercept (Canfield 1941). Gaps greater than 5 cm were not included. Height of the nearest shrub within 1 m of the transect line were measured at 2.5 m, 5 m, and 10 m. We documented the percent of forbs and grass cover (by lowest possible taxa), bare ground, and litter horizontal understory cover using 20 x 50 cm quadrats (Daubenmire 1959). We used the following 11 cover classes: Trace: 0-2%, 1: 3-9%, 2: 10-19%, 3: 20-29%, 4: 30-39%, 5: 40-49%, 6: 50-59%, 7: 60-69%, 8: 70-79%, 9: 80-89%, 10: 90-100%. We placed two quadrats on opposite sides of the nest bowl along the N/S transect line, and placed subsequent plots systematically and perpendicular to the transect at 2.5, 5, and 10 m locations, totaling 2 nest plots and 12 others. We measured grass and forb height along the transect, as well as the nearest plant using the grass/forb part at the point where the edge of the nest bowl and the transects intercept, and within the bottom left quarter each quadrat. We also recoded date, time, UTM coordinates, slope, aspect, and elevation of vegetation sampling sites.

Treatments

We will conduct habitat treatments in two lek complexes (T1 and T2; Figs. 2, 3). The actual location and placement of the habitat treatments depended upon landowner permission, federal restrictions with enrolled CRP, and agency funding. We will place treatments in habitat adjacent to and within 2 km of dancing grounds to elicit the maximum influence on breeding and summer habitat. Several authors report that 80% of the breeding and summer habitat is within 2 km of a dancing ground (Apa 1998, Boisvert 2002, Collins 2004, Apa 2014, Hoffman et al. 2015, this study). NWRTHC finalized treatments in the spring of 2016.

The NWRTHC prescribed and conducted treatments in collaboration with CSTG experts, a possible approach could include a disking with interseeding of bunchgrasses and forbs (Negus et al. 2010). Negus et al. (2010) recommended that 25-50% (314 ha - 628 ha) of the potential treatment area (area of a 2 km radius from a capture lek; 1,256 ha) should be treated per year with all treatments occurring in four years or less. This area of potential treatment could encompass several spring-fall or brood-rearing home ranges (Boisevert 2002, Collins 2004). Negus et al. (2010) found treatment establishment in approximately three years post treatment, but recommended that research should be delayed as much as five years post-treatment to yield more conclusive results of bird response. We began treatments in the fall of 2016 and will continue into the fall of 2017.

Working cooperatively with the NWRTHC, we identified and finalized treatments areas working cooperatively with private landowners, the Natural Resources Conservation Service (NRCS), and the Farm Services Agency (FSA). Although we initially outlined treatments to be conducted in one year, FSA vegetation manipulation restrictions for mid-contract maintenance of the properties enrolled in CRP will prevent such an approach. Maintenance requirements differ for enrolled fields that are at a 65 ha threshold. For enrolled fields < 65 ha, we can only treat 50% of the field in year 1 and the remaining in year 2. For fields > 65 ha, we can only treat 33% of a field in year 1, 33% in year 2, and 33% must remain untreated. Thus, in Treatment Area 1 (Fig. 2), we will treat 140 ha in 2016 and 140 ha in 2017. In Treatment Area 2 (Fig. 2), we will treat 202 ha in 2016 and 202 ha in 2017.

Although there are numerous vegetation manipulation approaches to reduce non-native grass cover and increase plant species richness, we identified the following protocol to implement habitat treatments. First, during late-summer (after nest hatch), we will initiate treatments with mechanical tillage equipment (off-set disc) to reduce viable non-native perennial grass cover and assist with seed-bed preparation. Second, approximately 2-4 weeks after mechanical tillage, we will treat sites with a chemical aerial application of Plateau® and glyphosate to reduce non-native perennial grass and limit annual and perennial grass seed germination. We may need to treat with a second application of glyphosate. Lastly, in late-fall, we will drill a seed mixture of native and non-native grasses, forbs, and shrubs (Table 1) with a no-till drill.

Study Design and Data Analyses

We are conducting our research project on private land with willing landowners (Fig. 2). Based on previous experience, many landowners will likely have access and/or treatment restrictions, thus situations could arise that may affect the access, timing, and/or replication and randomization of treatments and controls. Possible scenarios could include, landowners choosing to discontinue involvement in the study, changes in landownership or land management influencing the location, size or seed composition of a treatment therefore, a flexible study design is needed.

The aforementioned scenarios would impact the primary tenants of experimental treatments; randomization and replication (Wiens and Parker 1995). To accommodate these potential issues, we will treat these modifications in the same manner as described by Eberhardt and Thomas (1991) and Wiens and Parker (1995) in describing the analyses of the effects of accidental environmental impacts. Since accidental environmental impacts are unplanned and not replicated or spatially and statistically balanced (Eberhardt and Thomas 1991, Wiens and Parker 1995), they are characteristically temporally or spatially impacted by pseudoreplication (Hurlbert 1984, Stewart-Oaten et al. 1986). Wiens and Parker (1995:1071) acknowledged the pseudoreplication of treatments in accidental environmental impact and the associated non-independence among samples and termed them "judicious pseudoreplication."

To accommodate judicious pseudoreplication and other study design challenges, an alternative study design has been selected that involves the comparison of an impact site before and after while accounting for issues with natural change by pairing it to a control (Eberhart 1976, Steward-Oaten et al. 1986) or reference site (Steward-Oaten and Bence 2001); a before-after control-impact design (BACI) (Smith 2002). Although there are criticisms of BACI designs and its inability to discriminate the effects of treatments with a single control (Underwood 1991, 1992, 1994), Steward-Oaten and Bence (2001) argued that criticisms are unwarranted because BACI controls are not true experimental controls in the statistical sense because they are not independent or randomly selected. They suggest that the controls in a BACI design are selected specifically for their correlative ability and thus can be used as covariates and not used to estimate variances of the effect estimates. Even though the BACI design is typically used in environmental impact assessments (Smith 2002), BACI designs have been recommended (Michener 1997) and applied (Maccherini and Santi 2012) in restoration ecology studies.

A BACI design with paired controls will be employed (Smith 2002). This design is somewhat similar to a typical repeated measures design with the following two-factor mixed-effect ANOVA model:

$$X_{ijk} = \mu + \alpha_i + \tau_{k(i)} + \beta_j + (\alpha\beta)_{ij} + \varepsilon_{ijk}$$

where μ is the overall mean, α_i is the effect of period (i = before or after), $\tau_{k(i)}$ represents the times within period ($k = 1, 2, \ldots t_A$, for i = after and $k = 1,2,\ldots,t_B$ for i = before), β_j is the effect of location (j = control or treatment), $(\alpha\beta)_{ij}$ is the interaction between period and location, and ε_{ijk} represents the error. The fixed effects include timing (before and after treatment), if the site is a treatment or control, and the interaction. The random effects include the before or after time period are nested within year and the treatment or control are nested within the replicated controls or treatments, and the interaction (Little et al. 2006).

BACI design assumptions include; the measurements within and across site and years are independent, normality of residuals, equality of variation at each site and year, and normality of year, site, year*site interaction effects. In BACI designs it is not necessary to be spatially or statistically balanced and the number of birds and transects can vary among sites and year and not all sites need to be measured in all years.

We have 2 control or reference sites (lek complexes; Fig. 2) that will have no habitat improvements. There will be degrees of habitat quality within the controls that include better quality (reclaimed mineland) and low to marginal quality (existing or expired CRP). Additionally, we have two treatment (impact) sites, (Figs. 2, 3). We are conducting sampling for at least two years before treatment (impact) and two years immediately post-treatment (impact).

Response variables will include nest survival (Rotella et al. 2004); adult and yearling monthly and annual survival; chick daily, monthly and annual survival/recruitment; and home range. Covariates will also include grass and forb cover and height and plant species richness. The long-term population response and associated demographic rates will be evaluated using population matrix models (Caswell 2001, Powell et al. 2000, Doherty et al. 2004, Sæther and Bakke 2000). Chick, juvenile, and adult/yearling survival will be estimated using the Kaplan-Meier (K-M) (Kaplan and Meier 1958) product-limit function with staggered entry (Pollock et al. 1989).

Female home range will be estimated using a nonparametric fixed kernel density estimator (Worton 1989, White and Garrott 1990) that is based on the distribution and concentration of locations (Janke and Gates 2013). Since bandwidth selection can influence home range estimates (Gitzen et al. 2006, Downs and Horner 2008) we will follow a procedure outlined by Janke and Gates (2013) and will compare 3 bandwidth estimators. The estimators will include least squares cross validation (Seaman and Powell 1996), reference bandwidth (Worton 1989), and likelihood cross validation (Horne and Garton 2006, Horne and Garton 2009) and they will be compared in relation to data fit across point patterns and sample sizes (Janke and Gates 2013).

RESULTS

We captured 105 female CSTG (78 adults: 27 yearlings). Our capture dates in 2016 were from 9 April–3 May (Fig. 4). We trapped on 8 dancing grounds in 4 study areas (Hayden; Big Elk 1: West Axial; Moffat County Road 53 and Temple; Iles Dome; Iles Dome 2, 3, and 4: Trapper; Trapper Mine 1, and 7). In 2015 (Apa 2015), we trapped at more dancing grounds and captures occurred earlier in time (Fig. 4). Adult and yearling female mass ($\bar{x}$ ± SE) was 671.3 ± 5.2 g (n = 78) and 620.8 ± 8.9 g (n = 27), respectively. Female mass appears to vary by study area (Fig. 5), by age, and spatially (Fig. 6).

From April through September 2016, we documented 31 and 6 adult and yearling female mortalities resulting in a six-month survival rate for adult females of 0.52 ± 0.05 (n = 100; 95% CI 0.43 - 0.61) and for yearling females of 0.49 ± 0.01 (n = 27; 95% CI 0.33 - 0.65) (Fig. 7). We pooled adult and yearling female survival yielding a female survival rate of 0.67 ± 0.04 (*n* = 127; 95% CI 0.58 - 0.76) (Fig. 8). We also estimated female survival for each study area (Fig. 9). Female survival appeared similar between 2015 (0.62 ± 0.01 (*n* = 107; 95% CI 0.52 - 0.72) and 2016 (Fig. 10).

We documented an overall nest initiation rate of 95% (*n* = 74/78) and 96% (*n* = 21/22) for adult and yearling females, respectively. Females that did not survive to the nesting season (1 June) were not included. We documented an overall 54.4% (*n* = 56/103) and 59% (n = 56/95) apparent nest and female success, respectively. Seven females renested once yielding 42.9% (n = 3/7) nest success and one female successfully nested on a second renest attempt.

Female movement in 2016 from the lek of capture to nest averaged 2.42 ± 0.48 km (*n* = 94; range 0.17-35.0 km) (Fig. 11). The median distance moved was 1.1 km (25% quartile = 0.71 km; 75% quartile = 2.2 km). Seventy percent (*n* = 66/84) of the nests were located within 2 km of the lek of capture (Fig. 11). Female movements in the West Axial study area resulted in 62% (*n* = 13/21) of the females nesting within 2 km of the lek of capture and 83% (*n* = 20/24), 62% (*n* = 15/24) and 72% (*n* = 18/25) of females were nesting within 2 km of the lek of capture at the Iles Dome, Trapper, and Hayden study areas, respectively (Fig. 11). Over the 2 years of our study 72.2% (*n* = 127/176) of the females nested within 2 km of the lek of capture and by study area 48.7% (*n* = 18/37), 87.8% (n=43/49), 75.6 (*n* = 34/45), and 71.1% (*n* = 32/45) moved within 2 km of the West Axial, Iles Dome, Trapper, and Hayden study areas, respectively (Fig. 12).

We captured 391 chicks from 56 broods with an overall mean mass of 15.5 ± 0.1 g (range 11.8-23.1) and the average age of broods was 2.4 ± 0.1 days (range 2-6 days). A majority of chicks (96%; *n* = 376/391) were captured 1-3 days post-hatch and included 93% (*n* = 52/56) of the broods. Thus, the mean mass for chicks from 1-3 days-of-age was 15.2 ± 0.1 g (range 11.8 – 20.0). Chick mean mass by study area was 14.6 ± 2.0 g (*n* = 76; range 12.0-21.3), 15.2 ± 0.1 g (*n* = 119; range 12.8-18.5), 15.5 ± 0.1 g (*n* = 110; range 11.8-20.0), and 16.5 ± 0.2 g (*n* = 86; range 13.1-23.1) at West Axial, Iles Dome, Trapper, and Hayden, respectively (Fig. 13). There was a clear shift in chick mass with chicks being heavier in 2016 compared with 2015 with no chicks smaller than 12 g.

We radio-marked 211 chicks resulting in an average number of 3.8 chicks marked per brood. We also PIT tagged 172 chicks. The average brood size at marking was 7.7 chicks (range 3-14). The average marking time per brood was 28 minutes resulting in an average marking time of 7 minutes per chick. We recaptured and/or marked 119 juveniles that averaged 27 days post-hatch (range 18-53 days post-hatch). Juvenile mean mass was 112.0 ± 5.0 g (range 36.0-404.0 g). Nineteen of the juveniles were not previously marked. The average time for marking juveniles and broods was 8 minutes and 19 minutes, respectively.

We conducted vegetation sampling at 98 nest and 98 random sites and 33 brood and 33 random sites. We are currently proofing and entering data.

DISSCUSSION

Due to the more normal winter than 2014/15 our trapping time frame was later than previously reported by Boisvert (2002) and Collins (2004) and captures from 2015 (Apa 2015). Our adult:yearling capture ratio (2.89:1) was higher than 2015 (0.84:1) and was lower than reported by Collins (2004; 5.0:1) and Boisvert (2002; 3.6:1). Adult and yearling female mass was similar to earlier reports (Boivert 2002, Collins 2004), but appeared slightly lower than 2015 (Apa 2015).

Our 2016 six-month female survival (0.52) was similar to reports by Collins (2004; 0.41-0.58) for birds in reclaimed minelands, but lower (0.70-0.79) than females in shrub steppe habitat at 150 days exposure post-capture. Our survival was similar to that reported by Boisvert (2002; 0.50). We documented a similar nest initiation rate to Collins (2004; 97%) and Boisvert (2002; 97%) which were higher than in 2015 (Apa 2015). Our apparent nest success was higher than nest success reported by Collins (2004; 42%) but slightly lower than Boisvert (2002; 63%).

We changed chick and juvenile transmitter size from the proposal and project recommendation in Apa (2015). We reduced chick transmitter size from 0.65 to 0.55 g and juvenile transmitter size from 3.9 to 2.4 g in an attempt to reduce the overall percentage of transmitter:body mass ratio. We were successful in reducing the transmitter:body mass ratio for chicks to 3.55% and juveniles to 2.14%. The lowering of the transmitter:body mass ratio was partly a result of the smaller transmitter but also due to chick mass increasing from 13.8 g in 2015 to 15.5 in 2016. Our transmitter percentage of body mass is still a lower percentage of chick mass than Manzer and Hannon (2007), which fit chicks with transmitters similarly and reported a transmitter mass of 6-8% of chick mass. Even though the transmitter percentage of body mass was 3.55%, as chicks age, and become flight capable, transmitter mass will decline to < 1% as chick mass increases. In no case this year did our transmitter:chick mass ratios exceeded 5% (a recommended standard) which is typically recommended for flight capable birds and may be more important when considering power requirements for flight (Cochran 1980, Caccamise and Hedin 1985, Fair et al. 2010). As recommended in Apa (2015) we reconsidered and changed the original project proposal transmitter size and successfully reduced the transmitter percentage of chick body mass.

At the writing of this report, data entry and proofing is continuing.

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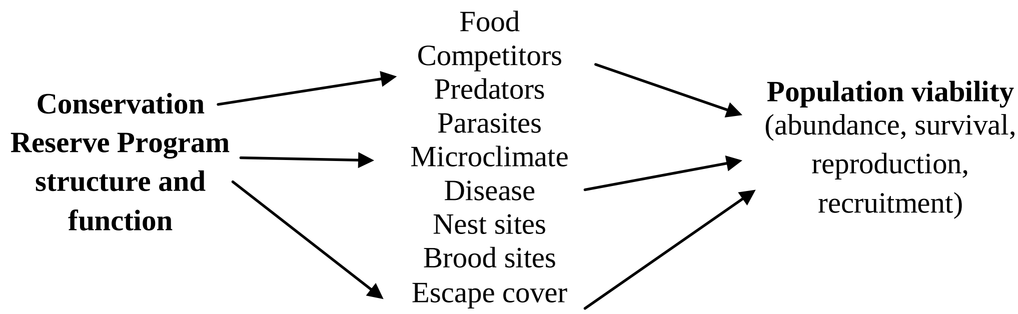

Figure 1. Mechanisms that link CRP structure and function to population viability (adapted from Marzluff et al. 2000).

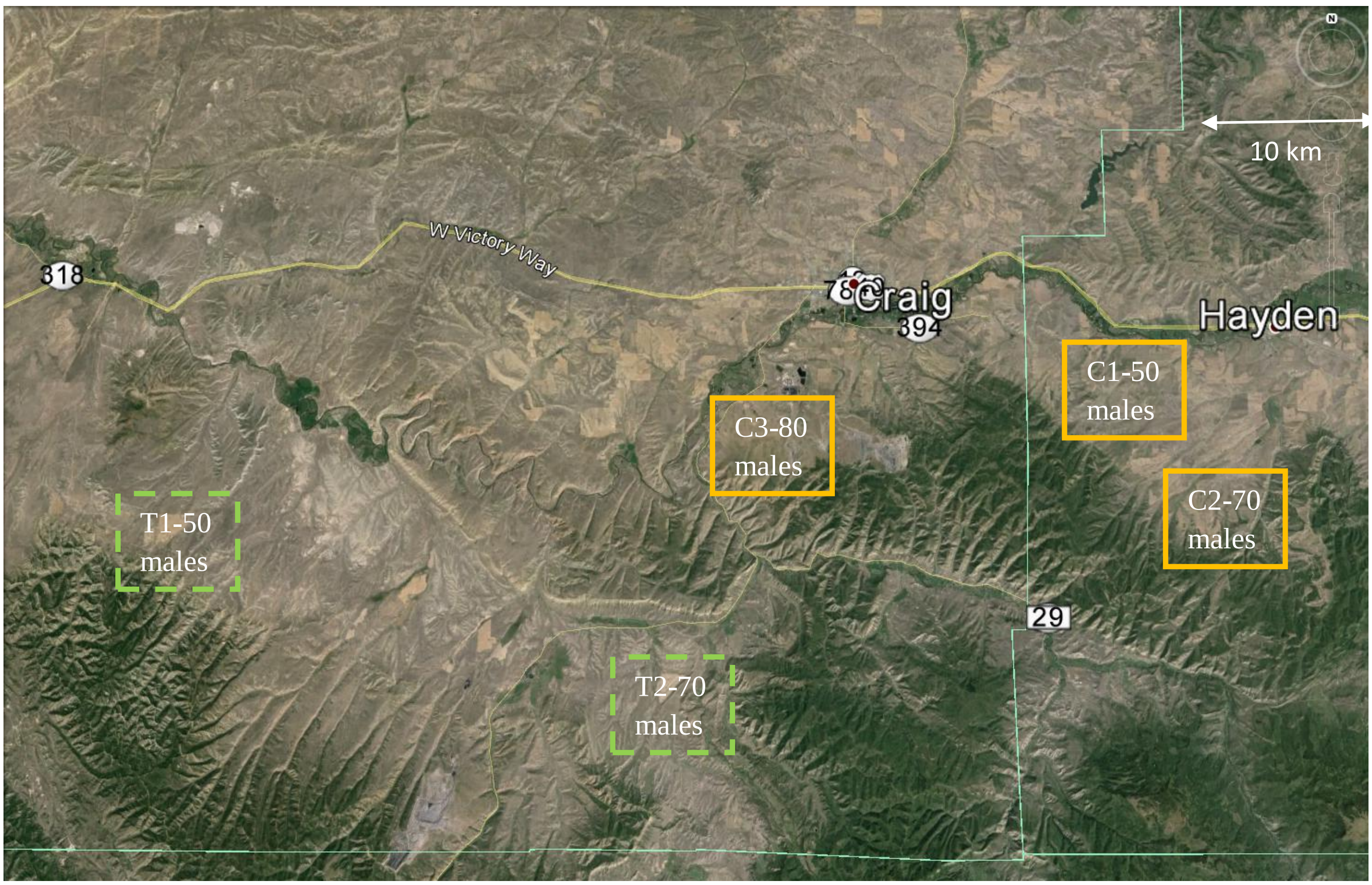

Figure 2. Study area location of treatment (T) and control (C) sites and the number of males on 2 or more dancing grounds in Moffat and Routt counties, Colorado. Study site C1 was not used in 2016.

Table 1. Plant scientific and common name and cultivar seeded in treatments in northwestern Colorado, 2016-2017.

Scientific Name	Common Name	Cultivar
Graminodes		
Dactylis glomerata	Orchard grass	Piaute
Leymus cinereus	Basin wildrye	Magnar/Trailhead
Poa secunda	Sandberg bluegrass	Sherman
Koeleria macrantha	Prairie Junegrass	Barkoel
Elymus lanceolatus ssp. lanceolatus	Thickspike wheatgrass	Critana
Achnatherum hymenoides	Indian ricegrass	
Elymus elymodies	Bottlebrush squirreltail	
Sporobolus crytandrus	Sand dropseed	
Forbs		
Sanguisorba minor	Small burnet	Delar
Onobrychis vicifolia	sainfoin	Eski/Melrose/Remont
Medicago sativa	Alfalfa	Falcata
Medicago sativa	Alfalfa	Ladak
Achillea millefolium	Western yarrow	
Linum lewisii	Lewis flax	Appar
Helianthus annuus	Common sunflower	
Cleome serrulata	Rocky Mountain beeplant	
Spaeralcea coccinea	Scarlet globemallow	
Shrubs		
Artemisia tridentata ssp. wyomingensis	Wyoming big sagebrush	
Artemisia tridentata ssp. tridentata	Basin big sagebrush	
Chrysothamus nauseosus	Rubber rabbitbrush	

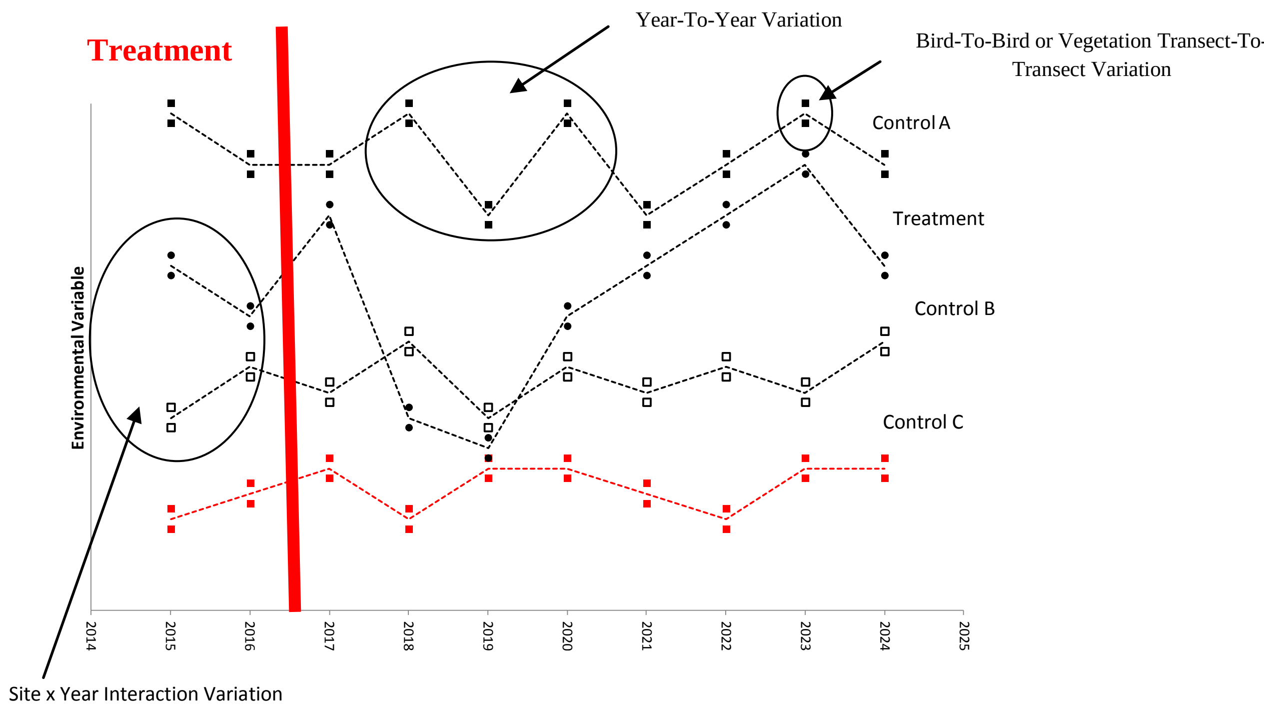

Figure 3. Conceptual schematic of a BACI design identifying the differing types of variation, treatment and control sites as well as the anticipated treatment in 2016 for Columbian sharp-tailed grouse habitat improvement. Only one treatment site is depicted.

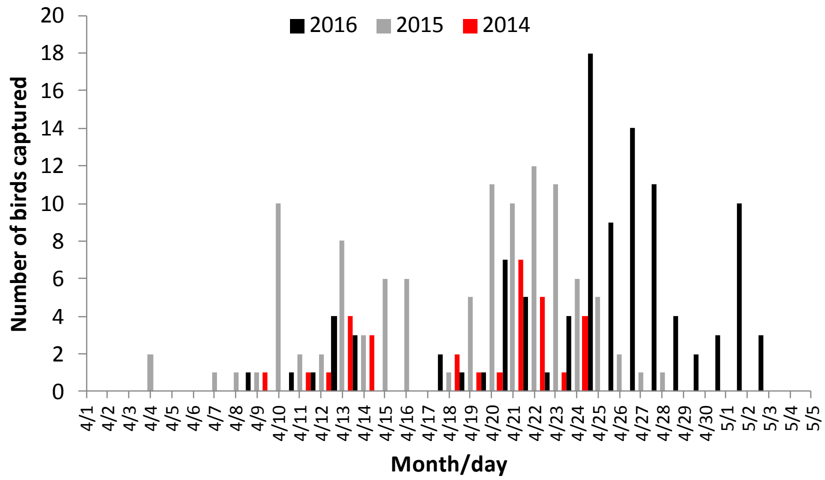

Figure 4. Number of female Columbian sharp-tailed grouse captured by date in northwestern Colorado, 2014-2016.

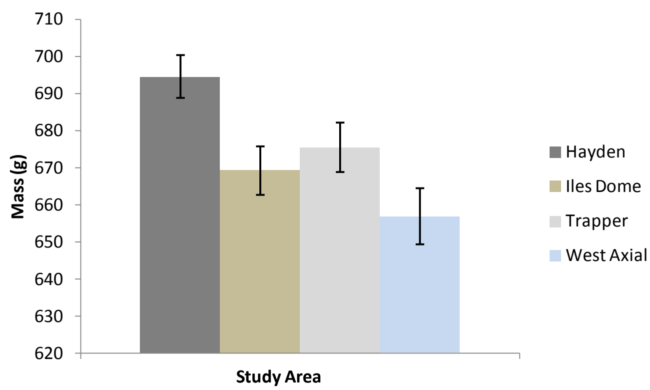

Figure 5. Mean mass ($\pm$ SE) of female Columbian sharp-tailed grouse at 4 study areas in northwestern Colorado, 2016.

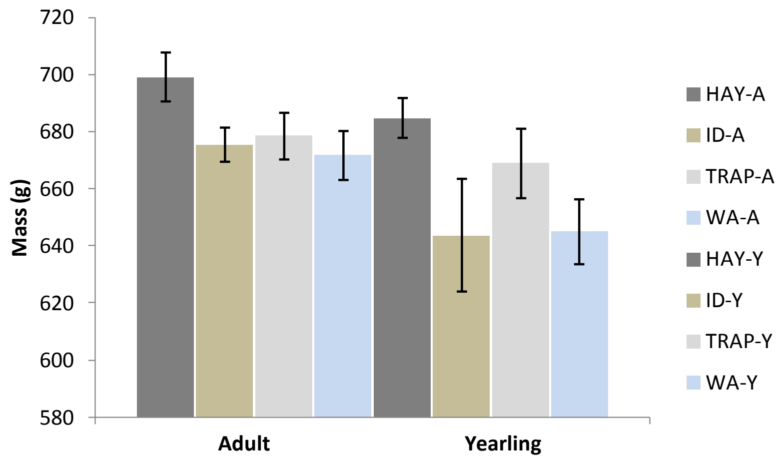

Figure 6. Mean mass (± SE) of female adult (A) and yearling (Y) Columbian sharp-tailed grouse at 4 study areas (HAY = Hayden; ID = Iles Dome; TRAP = Trapper; WA = West Axial) in northwestern Colorado, 2016.

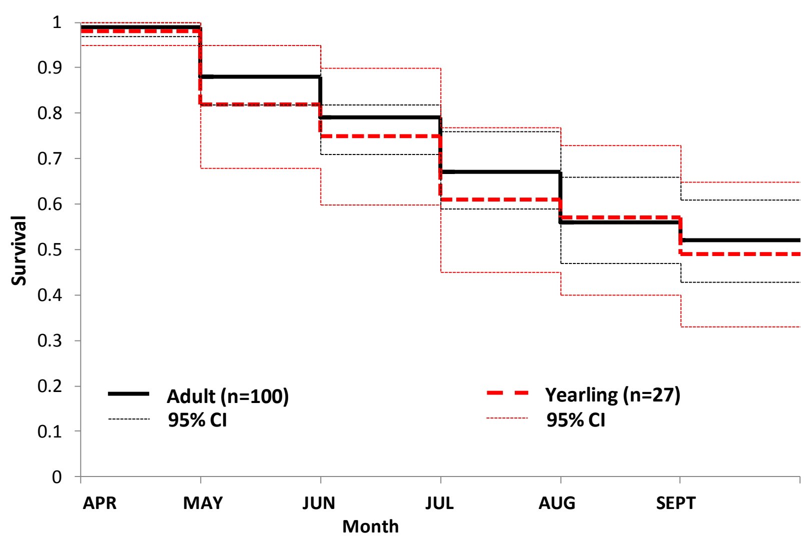

Figure 7. Kaplan-Meier product-limit monthly survival (± 95% CI) with staggered entry of adult (n = 100) and yearling (n = 27) female Columbian sharp-tailed grouse from April - September in northwest Colorado, 2016.

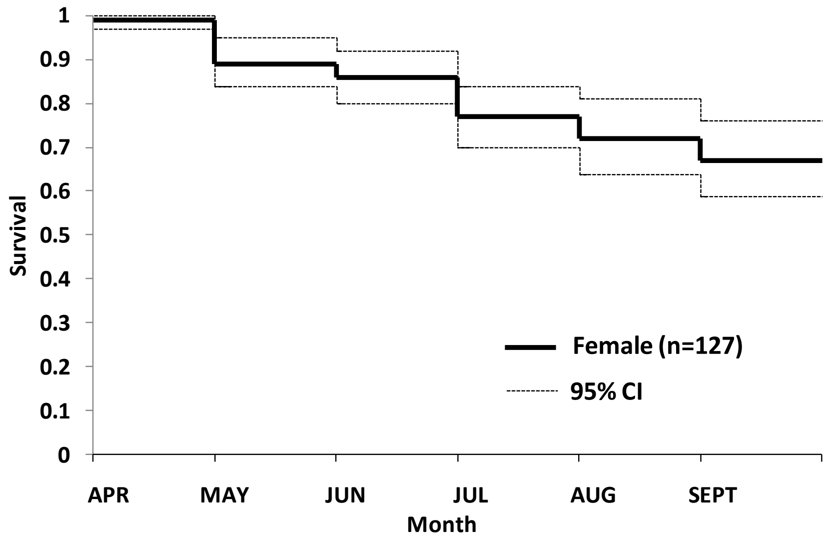

Figure 8. Kaplan-Meier product-limit monthly survival (± 95% CI) with staggered entry of female Columbian sharp-tailed grouse (*n* = 127) from April - September in northwest Colorado, 2016.

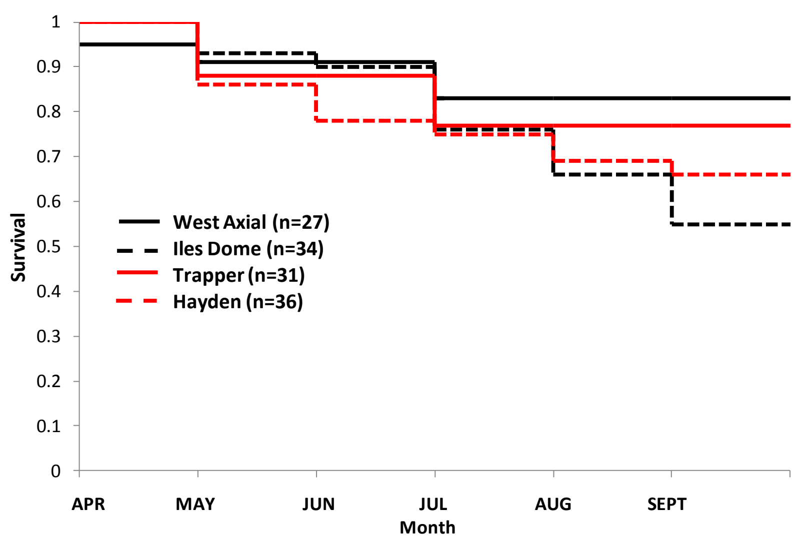

Figure 9. Kaplan-Meier product limit monthly survival with staggered entry of female Columbian sharp-tailed grouse from April – September for 4 study areas in northwestern Colorado, 2016.

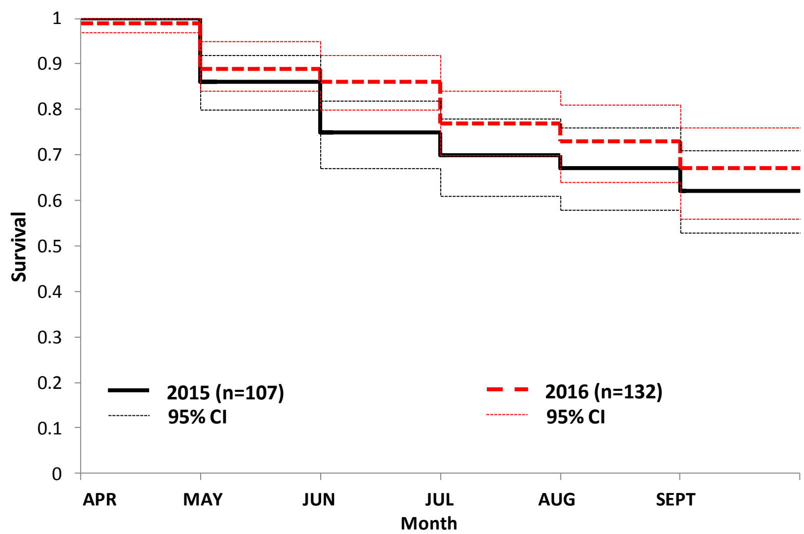

Figure 10. Kaplan-Meier product limit monthly survival with staggered entry of female Columbian sharp-tailed grouse from April – September in 2015 and 2016 pooled over 4 study areas, adults, and yearlings in northwestern Colorado, 2015-2016.

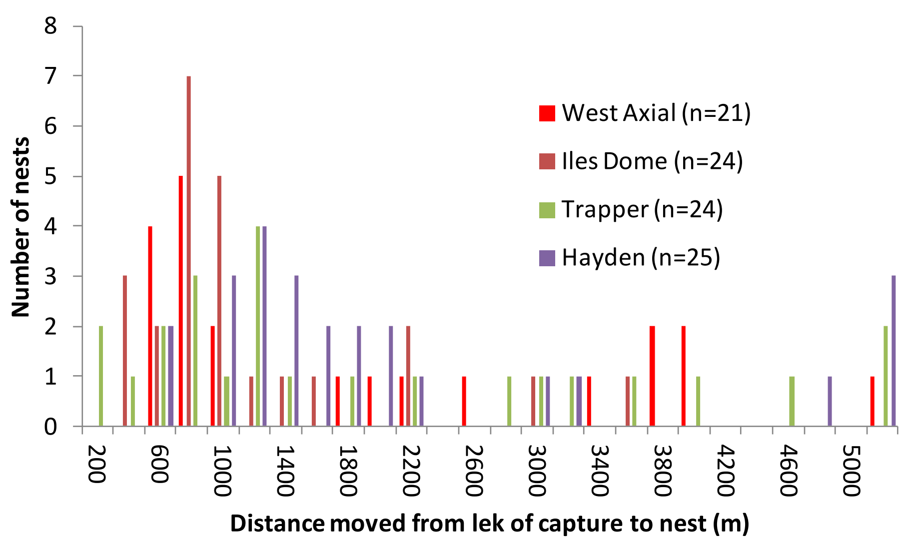

Figure 11. Frequency distribution of the number of Columbian sharp-tailed grouse nests by distance moved from the lek of capture by study area in northwestern Colorado, 2016.

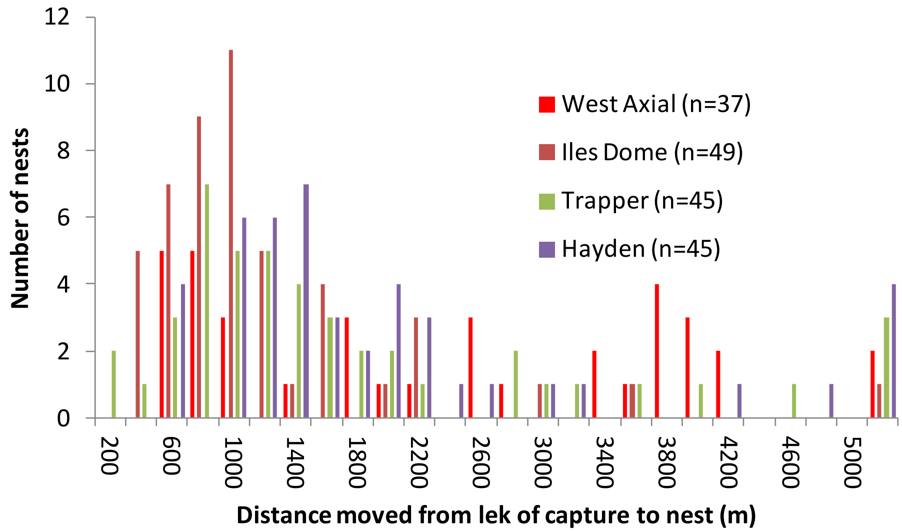

Figure 12. Frequency distribution of the number of Columbian sharp-tailed grouse nests by distance moved from the lek of capture by study area in northwestern Colorado, 2015-2016.

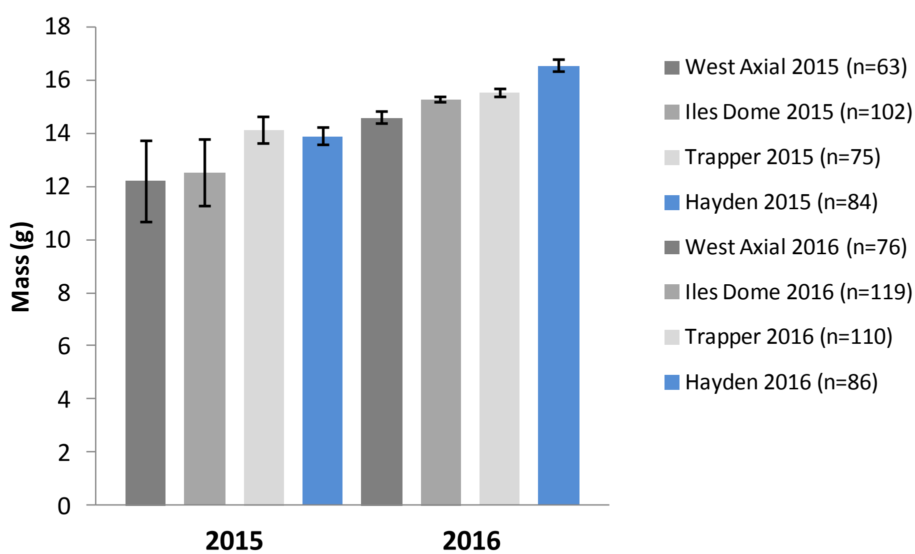

Figure 13. Mean (± SE) mass of chicks captured at 4 study areas in northwestern Colorado in 2015 and 2016.

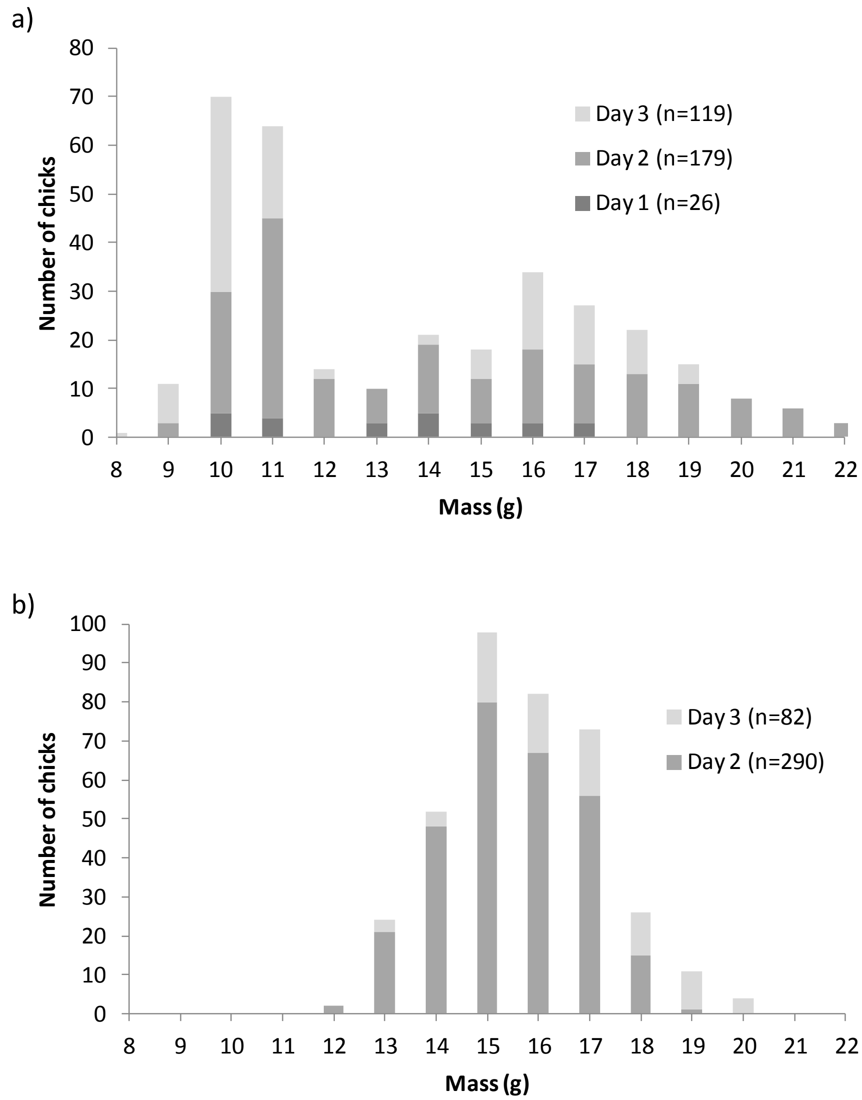

Figure 14. Frequency distribution of the number of Columbian sharp-tailed grouse chicks captured on day 1, 2, and 3 following hatch by mass in northwestern Colorado in 2015 (a) and 2016 (b).

Appendix A

Figure A-1. The 2016 Columbian sharp-tailed grouse field crew. Staff included from left to right, (back row) Jessica Shapiro, Dakota Vaccaro, Brittany Austin, Rebecca deVergie, Vincent Johnson, (front row) Rachel Harris, Brady Neiles, Anna Butler, and Ariana Dickson.

Figure A-2. Male Columbian sharp-tailed grouse conducting breeding display (dancing). Photo courtesy of Chris Yarbrough.

Figure A-3. One day-old Columbian sharp-tailed grouse chick after being fitted with a 0.65 g VHF micro-transmitter.

Figure A-4. Twenty day-old Columbian sharp-tailed grouse juvenile fitted with a 3.9 g VHF micro-transmitter that replaces the chick transmitter seen in Figure A-3.

Figure A-5. Three month old subadult Columbian sharp-tailed grouse being fitted with an adult 15 g transmitter that replaces the 3.9 g juvenile transmitter that will be removed (see Figure A-4).

Figure A-6. Staff making final adjustments to Columbian sharp-tailed grouse trapping configuration. Photo courtesy of Ariana Dickson.