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WILIDLIFE RESEARCH REPORT

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Author: A. D. Apa

Personnel: Jim Haskins and Bill deVergie, Area Wildlife Managers; Brad Petch, Senior Terrestrial Biologist; Trevor Balzer Sagebrush Habitat Coordinator; Kathy Griffin, Grouse Coordinator; Liza Rossi, Brian Holmes, and Jeff Yost, Terrestrial Biologists, Michael Warren, Energy Liaison; Becky Jones, Biologist-RMBO/NRCS/CPW

EXTENDED ABSTRACT

The Columbian sharp-tailed grouse (CSTG, *Tympanuchus phasianellus columbianus*) is one of 6 subspecies of sharp-tailed grouse in North America. Historically its distribution ranged from the northwest in British Columbia to the southwest in Colorado. Isolated populations exist (or formally existed) in Washington, Idaho, Wyoming, Colorado, Montana (extirpated), Utah, Nevada (reintroduced) and Oregon (reintroduced) occupying 10% of its former range. Habitat loss and degradation from anthropogenic activities are cited as the primary reasons for its decline with the conversion of native shrub plant communities to agricultural production being the most prevalent. 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. 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 to assess 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. My 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) and short-term (2 years) demographic and spatial parameters in existing non-native grass dominated communities and compare with treated sites, and 2) ascertain the long-term (5-7 year) post-habitat enhancement, demographic and spatial parameters in non-native grass dominated communities and compare with treated sites. The goal of my research is to conduct treatments (habitat improvements) in two lek complexes (T1 and T2). The actual location and placement of the habitat enhancement will depend upon landowner permission and agency funding. Treatments will be in collaboration with NW Regional management staff and the Northwest Region Habitat Coordinator. A Before-After Control-Impact (BACI) design with paired controls will be employed. My study area is located in northwestern Colorado, specifically in southwestern Routt and southeast Moffat counties. 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 (Fig. 2). Female CSTG were captured 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) that range in size from 10 – 45 males. Trapping also occurred on dancing grounds in two study sites in Routt county (C1, C2) that ranged in size from 6 – 24 males. I fitted females with 12 g elastic necklace-mounted radio transmitter equipped with a 12-hour mortality circuit having an 8.5 month nominal battery life. I monitored movements every 1-3 days with hand-held Yagi antennas attached to a receiver. When monitoring revealed a successful hatch, I attempted to capture all chicks in the brood within 24 hours. I randomly selected 4 chicks/brood and fit a 0.65 g backpack style transmitter using sutures along the dorsal midline between the wings (Fig. A-3). I captured juveniles when they reached 20-23 days-of-age at approximately two hours before sunrise while juveniles are brooding with the female. I removed chick transmitters and replaced them with a 3.9 g back-pack style juvenile transmitter (Fig. A-4). I sampled vegetation at all nest and a sample of brood sites. I captured 109 female CSTG (49 adults: 58 yearlings: 2 unknown) from 1-28 April 2015 on 11 dancing grounds in 5 study areas. Adult and yearling female mass ($\bar{x}$ ± SE) was 694.0 ± 5.6 g (n = 58) and 680.2 ± 6.9 g (n = 49), respectively. From April through September 2015, I documented 23 and 17 adult and yearling female mortalities resulting in a 6-month adult female survival rate of 0.61 ± 0.01 (n = 59; 95% CI 0.48 - 0.74) and a yearling survival rate of 0.64 ± 0.01 (n = 48; 95% CI 0.48 - 0.79). I pooled female survival yielding a female survival rate of 0.62 ± 0.01 (n = 107; 95% CI 0.52 - 0.72) (Fig. 6). Female survival was similar among study areas. I documented an overall nest initiation rate of 82% (n = 40/49) and 91% (n = 40/44) for adult and yearling females, respectively. I documented 60% (n = 24/40) and 61% (n = 25/41) apparent nest success for adult and yearling females, respectively. Only one yearling female initiated a renest and it was unsuccessful. Female movement from the lek of capture to nest averaged 2.01 ± 0.32 km (n = 81; range 0.29 - 24.48 km). The median distance moved was 1.3 km (25% quartile = 0.83 km; 75% quartile = 2.0 km) (Fig. 10). Seventy-four percent (n = 61/82) of the nests were located within 2 km of the lek of capture. A slightly different scenario presented itself among study areas. Female movements in the West Axial study appeared to move further with only 31% (n = 5/16) of females nesting within 2 km of the lek of capture while 92% (n = 23/25), 91% (n = 19/21) and 70% (n = 14/20) of females nesting within 2 km of the lek of capture at the Iles Dome, Trapper, and Hayden study areas, respectively. I captured 355, chicks from 49 broods with an overall mean mass of 13.8 ± 0.8 g (range 8.0 – 30.4) that ranged in age from 1-8 days. A majority of chicks (91%, n=324/355) were 1-3 days-of-age and included 86% (n = 42/49) of the broods. Thus, the mean mass for chicks from 1-3 days-of-age was 13.2 ± 0.2 g (range 8.0 – 21.6). Chick mean mass by study area was 12.3 ± 1.5 g (n = 63; range 9.2 – 17.0; 95% CI 11.6-12.8), 12.5 ± 1.2 g (n = 102; range 8.0 – 21.2; 95% CI 11.9-13.1), 14.1 ± 0.5 g (n = 75; range 9.0 – 21.6; 95% CI 13.1-15.1), and 13.9

± 0.3 g (*n* = 84; range 9.4 – 18.7;95% CI 13.2-14.5) at West Axial, Iles Dome, Trapper, and Hayden, respectively. Seventy-five percent (*n* = 243/324) of chicks captured were ≤16 g and 41% weighed 10-11 g (Fig. 13). Thus, the percentage of body mass for transmitters was as high as 8% for chicks weighing 8 g (only 1 was that small), but 41% (*n* = 134/324) would have had a transmitter mass of 6.5%. I radio-marked 179 chicks resulting in an average number of chicks marked/brood of 3.7 chicks. Total average brood size was 7.5 chicks (range 2 - 13). I recaptured and marked 76 juveniles at approximately 18 - 21 days-of-age. At the time of this report I have not estimated survival for chicks or juveniles. I conducted vegetation sampling at 66 nest sites and 69 random sites. Due to logistical issues, I did not conduct vegetation sampling at brood sites. My 6-month female survival (0.61) was slightly higher than previous reports (2004;0.41 - 0.58) for birds in mineland reclamation, but lower (0.70 - 0.79) than females in shrub steppe habitat at 150 days exposure post-capture. In contrast, my survival was higher than other reports (2002; 0.50). I documented a similar, but slightly lower, nest initiation rates than (2004;97% and 2002; 97%) which could be explained by the larger number of yearlings females in my sample. My apparent nest success was higher than one previous report (2004;42%) but similar to another (2002;63%). Transmitter size was higher than my recommended 5% of body mass which is a concern and was an unexpected result based on data from my pilot study. In previous studies chick mass ranged from 15 - 19 g, which is similar to reports on plains sharp-tailed grouse. As chicks age, and become flight capable, transmitter mass will decline to < 1% as chick mass (85- 130 g) increases. Although some transmitter:chick mass ratios exceeded 5% (a recommended standard), this percentage is typically recommended for flight capable birds and may be more important when considering power requirements for flight. Regardless, these results strongly suggest that the day-old chick transmitter size (0.65 g) needs to be reconsidered. Other transmitter sizes are available that range in size from 0.2-0.55 g. The 0.2, 0.3, and 0.5 g transmitters are of a glue-on style and to be retrofit for suture style will require an increase of 0.05 g/transmitter. Clearly, a decrease in transmitter weight will have a concomitant decrease in battery life from 36 days for 0.65 g to 12 days for 0.20 g with a pulse rate of 30 ppm. This is the first of four planned field seasons; two before treatment and two following treatment.

WILDLIFE RESEARCH REPORT

COLUMBIAN SHARP-TAILED GROUSE DEMOGRAPHIC RESPONSE TO HABITAT IMPROVEMENTS

ANTHONY D. APA

INTRODUCTION

The Columbian sharp-tailed grouse (CSTG, *Tympanuchus phasianellus columbianus*) is one of 6 subspecies of sharp-tailed grouse in North America (Connelly et al. 1998). Historically its distribution ranged from the northwest in British Columbia to the southwest in Colorado (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) 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 its decline (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.

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 efforts 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 species of "state special concern." 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-1950's to mid-1960's many of the native sagebrush communities on private land were converted to agricultural production (Braun et al. 1976). These practices continued into the mid-1980's until the 1985 Farm Bill provided an opportunity for private landowners to enroll highly erodible lands into the CRP which ultimately removed these agricultural lands from production (Negus et al. 2010). Since the goal was to stabilize erodible soils, many CRP planting seed mixes included only 2-3 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). In some situations in Washington, 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 an issue for 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). Additionally, some CRP plantings

in Idaho were sufficiently diverse to support CSTG (Apa 1998) and facilitate range expansion (Mallett 2000).

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 (Boisvert 2002) or native rangeland (Collins 2004). Mineland reclamation provides 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 mineland reclamation 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 mineland reclamation. Boisvert (2002) also reported higher CSTG productivity in mineland reclamation habitat. Nest success was nearly five times higher for females nesting in mineland reclamation 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 mineland reclamation. 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 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 habitat improvements (quality) through wildlife habitat enhancement and/or management 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 that influence demography as possible, 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). To better understand and improve the predictive ability of habitat quality improvements on population viability, the mechanisms responsible for these changes need additional understanding (Raphael and Maurer 1990, Marzluff et al. 2000; Fig 1.).

I define 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 mineland reclamation (75 ha) when compared to CRP (112 ha). These life history traits and relatively small movements facilitate a relatively rapid response to habitat management, ultimately providing managers and researchers an opportunity to work collaboratively to investigate a mechanistic response to landscape level habitat quality improvements.

To evaluate the demographic and population response of CSTG to breeding and summer/fall 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 an 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 inprecise estimates of chick survival because of unknown detection probabilities associated with cryptic chicks combined with a no movement defensive strategy to avoid detection. Other issues can bias chick survival estimates and include chick exchange between broods observed with greater sage-grouse (*Centrocercus urophasianus*) (Dahlgren et al. 2010, Thompson 2012).

To obtain a more reliable estimate of chick survival, my field methods will include the use of VHF micro transmitters attached to day-old chicks to obtain survival estimates using techniques established with surrogate species (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

My 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.
3. Ascertain the long-term (5-7 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

My study area is located in northwestern Colorado, specifically in southwestern Routt and southeast Moffat counties (Fig. 2). It 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 – Female CSTG were captured in the spring using 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 have 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. Traps were opened ½ hour before sunrise and closed/blocked at the cessation of trapping each morning. Trapping was timed to coincide with the peak of female attendance (Giesen et al. 1982, Giesen 1987, R. Hoffman, retired CPW, personal communication).

I fitted females with 12 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 < 2% (range 1.7 – 1.9%) of an adult or yearling female body mass. I bent the 16 cm antenna down the back to lie between the wings and down the back of the grouse. Captured grouse were classified by gender (Snyder 1935, Henderson et al. 1967) and age (Ammann

1944). I aged females as yearling or adult by examining the condition of the outer primaries (Ammann 1944). I collected mass (± 1 g) data by placing a restrained individual in a cotton bag and weighing it on an electronic balance.

I fitted all females with individually numbered aluminum leg bands (size 12) attached on the tarsus. I released all captured males. I processed individuals and released them at the point of capture. When releasing birds, I quickly and quietly backed away until the bird walked, ran, or flushed away.

Nest Monitoring and Chick Capture – I monitored movements every 1-3 days and general locations were obtained using triangulation from a ≥ 30 m distance (to minimize disturbance) with hand-held Yagi antennas attached to a receiver. I 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 I assumed nest initiation or incubation. I attempted to make visual observations of females on nests at 7-10 days post-incubation confirmation, but visibility depended on vegetation density. I monitored incubating females 2-3 times/week to monitor nest fate. I monitored nesting by using telemetry at two points at right angles from one another 10 -20 m distance (25-26 day incubation period) from the incubating female.

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

I weighed (± 0.01 g) all captured chicks using an electronic scale. I randomly selected 4 chicks/brood and fit a 0.65 g backpack style (model A1025, 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, I swabbed the suture site with isopropyl alcohol, and inserted two sterile, unused 20-gauge needles subcutaneously and perpendicular to the dorsal mid-line. I threaded the monofilament suture material (Braunamide: polyamide 3/0 thread, pseudo monofilament, non-absorbable, white) through the needle barrels. I then removed the needles and tied off the suture material using a square knot and removing excess suture material. I applied one drop of cryanocrylate glue on the knot. I monitored the brood female during brood processing to assure that she remains in the near vicinity.

I determined chick and brood positions by first locating females and circling at a 25 m radius. I also identified the position (i.e., distance) of radio-marked chicks in relation to the female. I attempted to find all chicks that are separated or missing from broods to determine fate and/or cause of mortality and I attempted to obtain brood locations equally among 4 time periods: brooding (< 2 hour after sunrise or before sunset), morning (0800-1100), mid-day (1100-1400), and afternoon (1400-1800).

I attempted to capture juveniles when they reached 20-23 days-of-age at approximately two hours before sunrise while juveniles are brooding with the female (Apa 2014). I circled the female and brood using radio telemetry approaching slowly with the aid of a "red light" on a head lamp and the location will be marked with yellow glow sticks. Once I obtained a visual location, the female and brood were captured using a 1.5 m diameter hoop net. I restrained all captured juveniles and released the female at the point of capture to avoid injury of juveniles.

I removed chick transmitters and replaced it with a 3.9 g back-pack style juvenile transmitter (Model A1080, Advanced Telemetry Systems, Isanti, MN) (Fig. A-4). I 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). I selected a new suture site near the previous suture site. I weighed all captured juveniles. I 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 has a nominal battery life of 8.5 months and consisted of 3.0 - 4.6% of chick mass (Apa 2014).

I captured 4-month old juveniles using a different approach. I located juveniles in the late-afternoon before an evening capture attempt. At 1-2 hours following sunset, I located the juveniles using radio-telemetry. Six staff were needed for a successful capture. Using radio telemetry, 4 people with long-handled hoop nets and 2 with spot-lights (1 with radio-telemetry) circled the bird with the 2 spot-lights 180^{o} from each other. People with nets were at 12, 3, 6, and 9 o'clock around the bird. Once I identified the estimated location, 2 glow sticks were placed near the estimated location all people converged on the juvenile with the aid of the spotlights. Once captured, the juvenile was fitted with an adult necklace mounted transmitter as earlier described (Fig. A-5). Juveniles were flushed no more than 3 times (including the initial flush) sequentially in a single evening.

Aerial locations and/or detections (survival) were obtained as needed for missing birds and will be obtained once/month throughout the research. All trapping and handling protocol were approved by the CPW Animal Care and Use Committee (Permit # 02-2015).

Habitat Quality

Vegetation Sampling – I sampled vegetation at all nest and a sample of brood sites. I placed four, 10-m transects in the cardinal directions intersecting at the nest bowl. Sampling was conducted as soon as logistically possible, within 7 days of nesting cessation (successful or unsuccessful), or the last brood location. I sampled paired random site vegetation sampling within 7 days of its paired sample. Abiotic site characteristics such as date, time, UTM coordinates, slope, aspect, and elevation were also recorded.

Overstory horizontal and vertical structure – When present, I 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.

Understory horizontal and vertical structure – I 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). I 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%. I 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. Grass and forb height was measured along the transect, and I measured 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.

Females with broods were located 1-2 times per week. Females with broods were circled, the intersection point of flags placed in the cardinal directions will be used to identify the center of the brood location which will determine the intersection point of the transects. Habitat measurements were conducted at as many brood locations as possible with equal sampling across individuals retain sample independence and avoid sampling autocorrelation issues.

I 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 which will serve as sampling locations for random sites. Cells with grouse locations were not considered as part of the random sample. The same vegetation data collection techniques were conducted on at least one paired random location for each nest and brood site.

Treatments

The goal of this research is to conduct treatments (habitat improvements) in two lek complexes (T1 and T2; Figs. 2, 3). The actual location and placement of the habitat enhancement will depend upon

landowner permission and agency funding. Treatments will be in collaboration with NW Regional management staff and the Northwest Region Habitat Coordinator (NWRHC). Treatments will be focused 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). Although the NWRHC will prescribe and conduct treatments in collaboration with CSTG experts, a possible approach could include a disking/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 4 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 3 years post treatment, but recommended that research should be delayed as much as 5 years post-treatment to yield more conclusive results of bird response. Treatments will be initiated between the fall of 2016 and the fall of 2017.

ANALYSIS

Study Design and Data Analyses

The research project is conducted 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 impact 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, I 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 (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,…t_A, for i = after and k = 1,2,…,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 are nested within year, 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 measure in all years.

I will have three control or reference sites (lek complexes; Figs. 2, 3) that will have no habitat improvements. There will be degrees of habitat quality within the controls that include better quality (mineland reclamation) and low to marginal quality (existing or expired CRP). Additionally, I will have two treatment (impact) sites, (Figs. 2, 3), but these treatment sites are still in development and not finalized (location, seed mix, and treatment approach) until there is additional communication with the landowners. I will conduct sampling for at least two years before treatment (impact) and two years immediately post-treatment (impact). I will not conduct active research for 5 - 7 years following treatment allowing for vegetation establishment and maturation. Once the treatment has matured, the long-term portion of the after treatment (impact) study will be conducted in the same manner as the before and immediately after treatment.

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) I 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 data fit across point patterns and sample sizes (Janke and Gates 2013).

RESULTS AND DISSCUSSION

Results - I captured 109 female CSTG (49 adults: 58 yearlings: 2 unknown) from 1-28 April 2015 on 11 dancing grounds in 5 study areas (South Hayden; Big Elk 1 and Postovit: South West Hayden; Smuin OGW1 and Haskins: West Axial; Moffat County Road 53 and Temple: Iles Dome; Iles Dome 3 and Iles Dome 2: Trapper; Trapper Mine 7 and Trapper Mine 1). For the purposes of this progress report, data from the South Hayden and West Hayden study areas were combined (Hayden). I captured a majority (>90%) of females from 10-25 April 2015 (Fig. 4). Adult and yearling female mass ($\bar{x}$ ± SE) was 694.0 ± 5.6 g (n = 58) and 680.2 ± 6.9 g (n = 49), respectively.

From April through September 2015, I documented 23 and 17 adult and yearling female mortalities resulting in a 6-month adult female survival rate of 0.61 ± 0.01 (n = 59; 95% CI 0.48 - 0.74) and a

yearling survival rate of 0.64 ± 0.01 (*n* = 48; 95% CI 0.48 - 0.79) (Fig. 5). I pooled female survival yielding a female survival rate of 0.62 ± 0.01 (*n* = 107; 95% CI 0.52 - 0.72) (Fig. 6). Specifically, female survival among study areas was also similar (Fig. 7).

I documented an overall nest initiation rate of 82% (*n* = 40/49) and 91% (*n* = 40/44) for adult and yearling females, respectively. Females that did not survive to the nesting season (1 June) were not included. I documented 60% (*n* = 24/40) and 61% (n = 25/41) apparent nest success for adult and yearling females, respectively. Only one yearling female initiated a renest and it was unsuccessful.

Female movement from the lek of capture to nest averaged 2.01 ± 0.32 km (*n* = 81; range 0.29 - 24.48 km) (Fig. 8). The median distance moved was 1.3 km (25% quartile = 0.83 km; 75% quartile = 2.0 km). Seventy-four percent (*n* = 61/82) of the nests were located within 2 km of the lek of capture (Fig. 8). A slightly different scenario presented itself among study areas. Female movements in the West Axial study appeared to move further with only 31% (*n* = 5/16) of females nesting within 2 km of the lek of capture while 92% (*n* = 23/25), 91% (*n* = 19/21) and 70% (*n* = 14/20) of females nesting within 2 km of the lek of capture at the Iles Dome, Trapper, and Hayden study areas, respectively (Fig. 8). This longer movement in the West Axial study areas by females was apparent with the mean (Fig. 9) and median distances (Fig. 10).

I captured 355, chicks from 49 broods with an overall mean mass of 13.8 ± 0.8 g (range 8.0 – 30.4) that ranged in age from 1-8 days. A majority of chicks (91%, n=324/355) were 1-3 days-of-age and included 86% (*n* = 42/49) of the broods. Thus, the mean mass for chicks from 1-3 days-of-age was 13.2 ± 0.2 g (range 8.0 – 21.6) (Fig. 11). Chick mean mass by study area was 12.3 ± 1.5 g (*n* = 63; range 9.2 – 17.0; 95% CI 11.6-12.8), 12.5 ± 1.2 g (*n* = 102; range 8.0 – 21.2; 95% CI 11.9-13.1), 14.1 ± 0.5 g (*n* = 75; range 9.0 – 21.6; 95% CI 13.1-15.1), and 13.9 ± 0.3 g (*n* = 84; range 9.4 – 18.7;95% CI 13.2-14.5) at West Axial, Iles Dome, Trapper, and Hayden, respectively (Fig. 12). Seventy-five percent (*n* = 243/324) of chicks captured were ≤16 g and 41% weighed 10-11 g (Fig. 13). Thus, the percentage of body mass for transmitters was as high as 8% for chicks weighing 8 g (only 1 was that small), but 41% (*n* = 134/324) would have had a transmitter mass of 6.5% (Fig. 14).

I radio-marked 178 chicks resulting in an average number of chicks marked/brood of 3.7 chicks. Total average brood size was 7.5 chicks (range 2 - 13). The average of chicks marked was 2.85 ± 0.09 days post-hatch (range 1-7; n=178). Chick survival for 20 days post-mark was 0.34 (95% CI: 0.25-0.43; Fig 15). I recaptured and/or marked 84 juveniles that averaged 27.10 ± 0.48 days post-hatch (range 20-39). Juvenile survival from remark (or mark) to 120 days post-hatch was 0.37 (95% CI: 0.22-0.52) (Fig. 16).

I conducted vegetation sampling at 66 nest sites and 69 random sites. Due to logistical issues, I did not conduct vegetation sampling at brood sites.

Discussion – Due to the mild winter my trapping time frame was considerably earlier than previously reported by Boisevert (2002) and Collins (2004) and lasted nearly one month. My adult:yearling capture ratio (0.84:1) was different than reported by Collins (2004; 5.0:1) and Boisvert (2002; 3.6:1), but adult and yearling female mass was similar to earlier reports (Boivert 2002, Collins 2004).

My 6-month female survival (0.61) was slightly higher than reported by Collins (2004;0.41 - 0.58) for birds in mineland reclamation, but lower (0.70 - 0.79) than females in shrub steppe habitat at 150 days exposure post-capture. In contrast, my survival was higher than reported by Boisvert (2002; 0.50). I documented a similar, but slightly lower, nest initiation rates than Collins (2004;97%) and Boisvert (2002; 97%) which could be explained by the larger number of yearlings females in my sample. My

apparent nest success was higher than nest success reported by Collins (2004;42%) but similar to Boisvert (2002;63%).

Transmitter size was higher than the recommended 5% of body mass which is a concern and was an unexpected result (Apa 2014). Manzer and Hannon (2007) fit chicks with transmitters similarly and reported a transmitter mass of 6 - 8% of chick mass (13.7 - 18 g) which is similar to my transmitter mass ratio. Manzer and Hannon (2007) also fit chicks with larger (1.1 g) transmitters which resulted in a higher percent of body mass than I report. In previous studies chick mass ranged from 15 - 19 g (Apa 2014) which is similar to PSTG (Manzer and Hannon 2007). Manzer and Hannon (2007) reported PSTG day-old chick mass (range; 14-18 g). I anticipated that transmitter mass would consist of 3 - 4% of the day-old non-flight capable chick mass and decline as chicks age. As chicks age, and become flight capable, transmitter mass will decline to < 1% as chick mass (85- 130 g) increases (Apa 2014). Although some transmitter:chick mass ratios exceeded 5% (a recommended standard), this percentage 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). Regardless, these results strongly suggest that the day-old chick transmitter size (0.65 g) needs to be reconsidered. Other transmitter sizes are available that range in size from 0.2-0.55 g (Fig. 14). The 0.2, 0.3, and 0.5 g transmitters are of a glue on style and to be retrofit for suture style will require an increase of 0.05 g/transmitter. Clearly, a decrease in transmitter weight will have a concomitant decrease in batter life from 36 days for 0.65 g to 12 days for 0.20 g with a pulse rate of 30 ppm.

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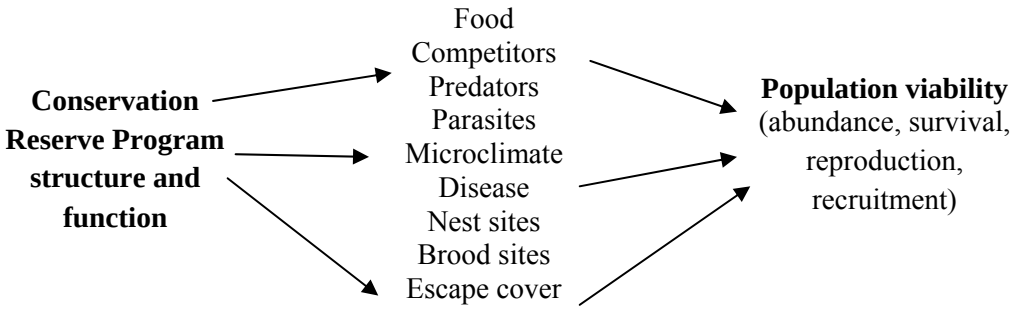

Figure 1. Mechanisms that link CRP structure and function to population viability (adapted from Marzluff et al. 2000).

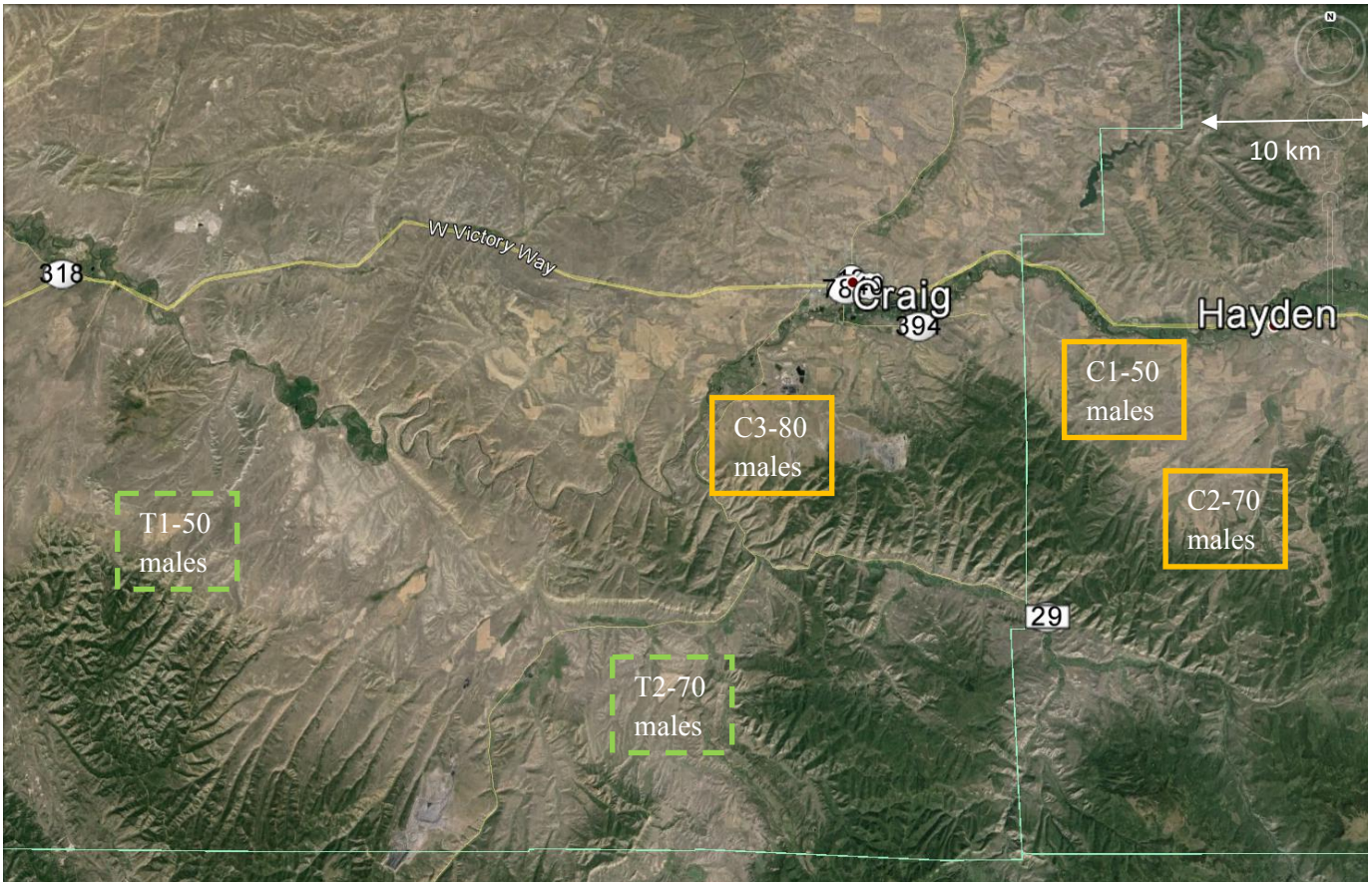

Figure 2. Study area location of treatment (T) and control (C) sites and the number of males on 2 or more dancing grounds in Moffat and Routt counties, Colorado.

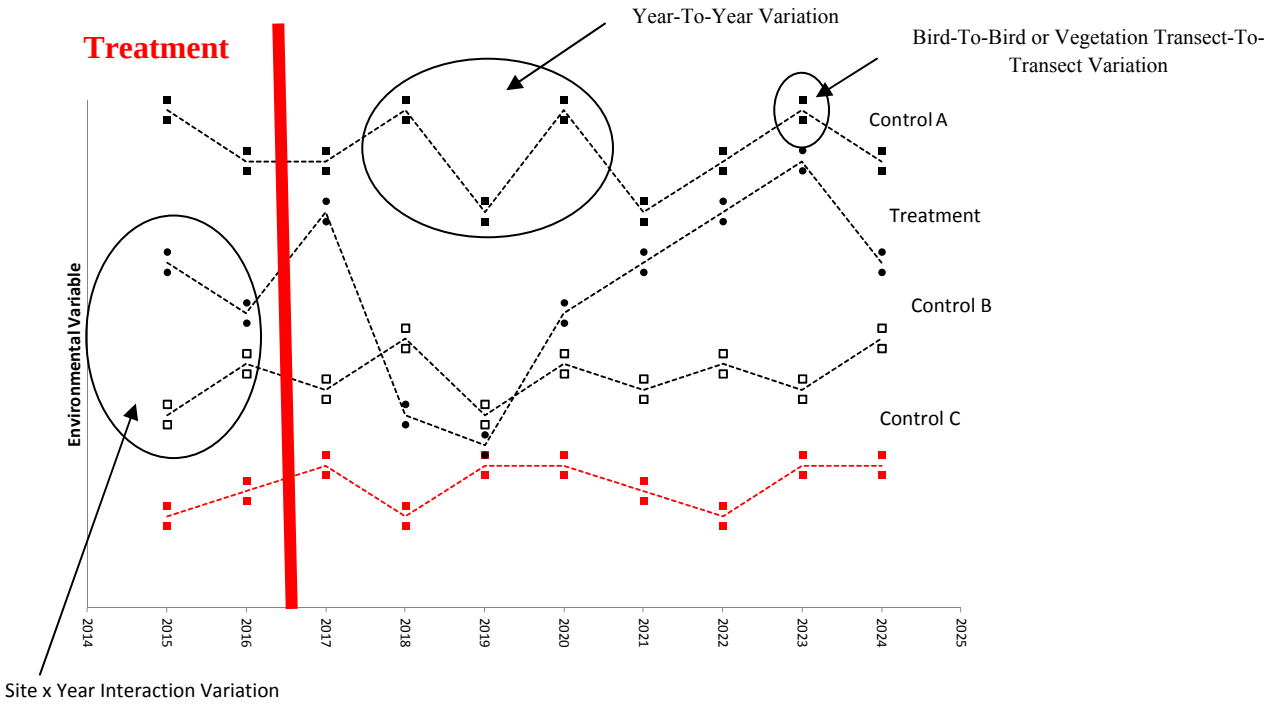

Figure 3. Conceptual schematic of a BACI design identifying the differing types of variation, treatment and control sites as well as the anticipated treatment in 2016 for Columbian sharp-tailed grouse habitat improvement. Only one treatment site is depicted.

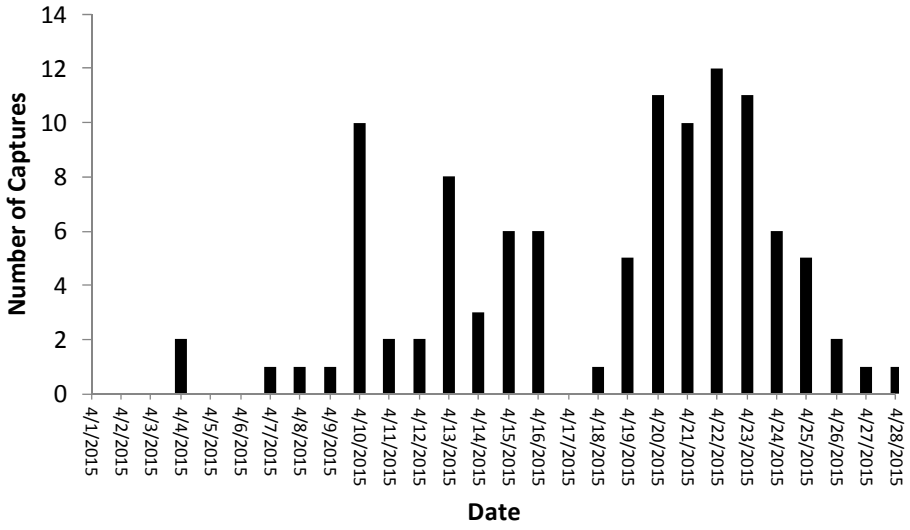

Figure 4. Number of female Columbian sharp-tailed grouse captured by date in five study areas in northwestern Colorado, 2015.

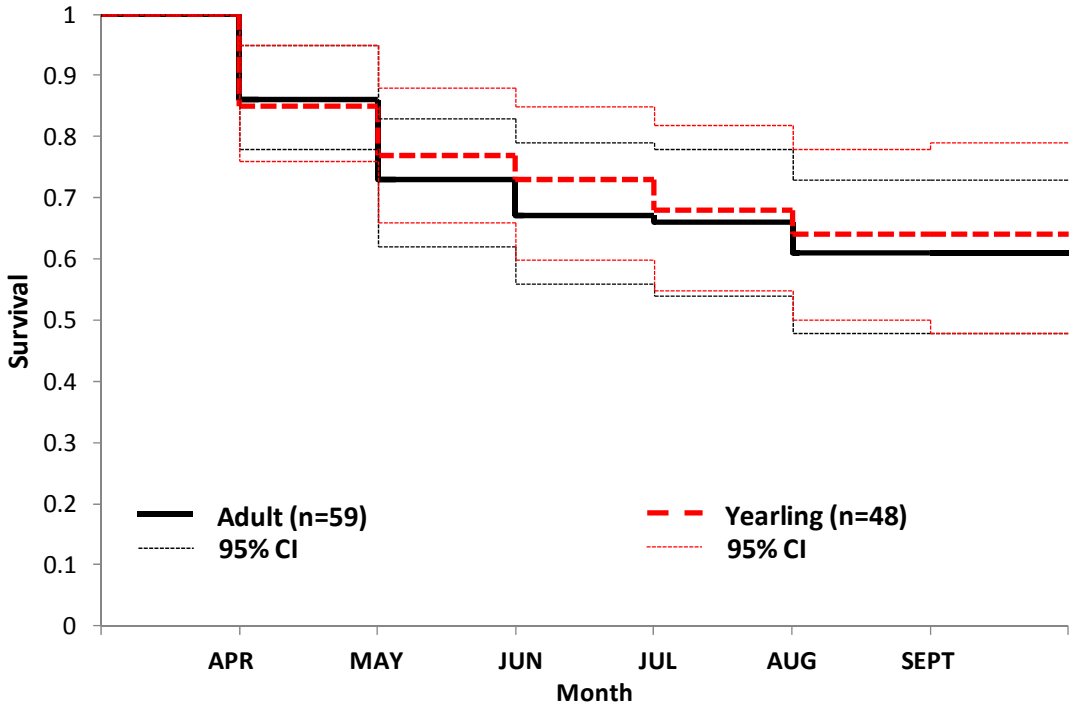

Figure 5. Kaplan-Meier product-limit monthly survival (± 95% CI) with staggered entry of adult (n = 59) and yearling (n = 48) female Columbian sharp-tailed grouse from April - September in northwest Colorado, 2015.

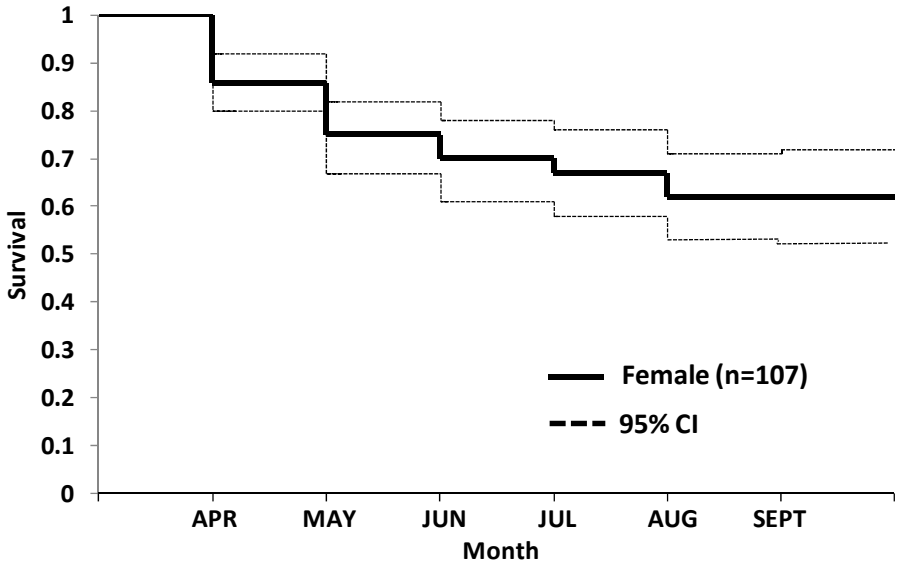

Figure 6. Kaplan-Meier product-limit monthly survival (± 95% CI) with staggered entry of female Columbian sharp-tailed grouse (*n* = 107) from April - September in northwest Colorado, 2015.

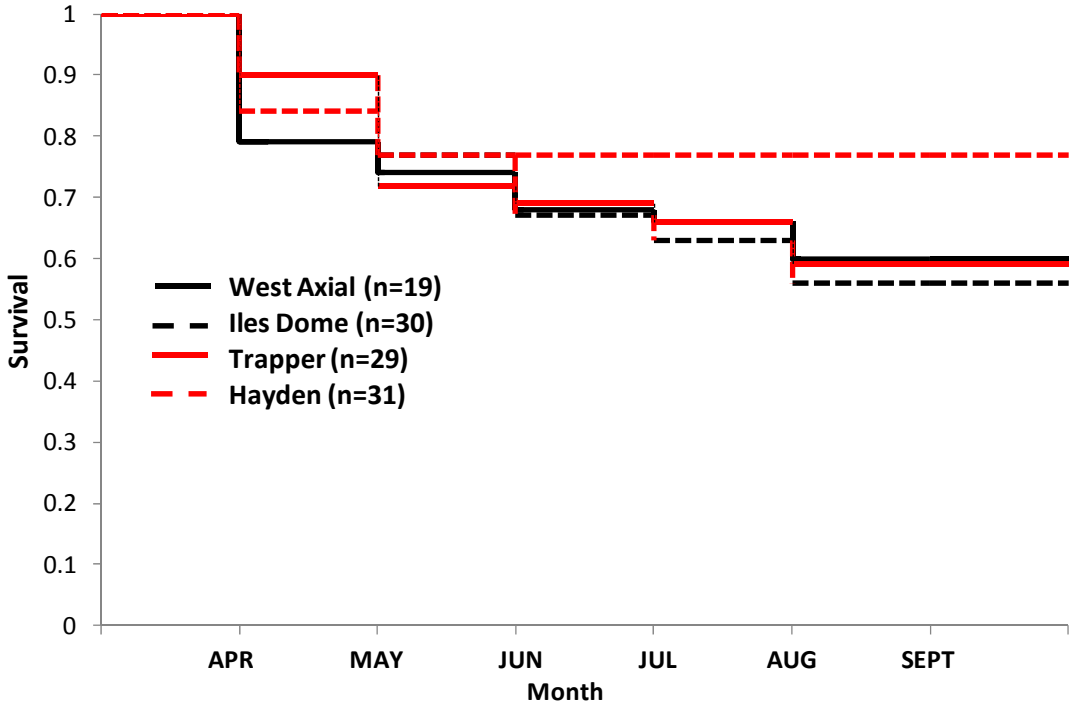

Figure 7. Kaplan-Meier product limit monthly survival with staggered entry of female Columbian sharp-tailed grouse from April – September for 4 study areas in northwestern Colorado, 2015.

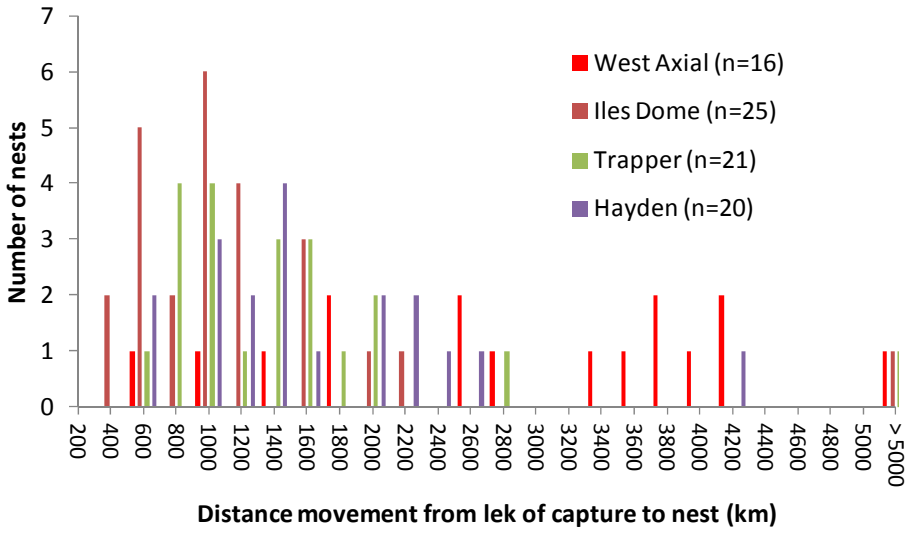

Figure 8. Frequency distribution of the number of Columbian sharp-tailed grouse nests by distance moved from the lek of capture by study area in northwestern Colorado, 2015.

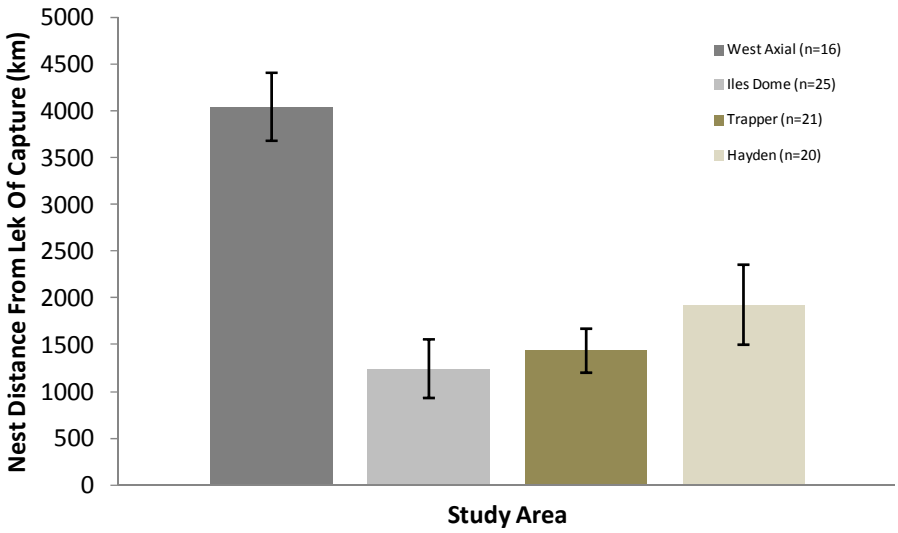

Figure 9. The mean (± SE) distance moved by female Columbian sharp-tailed grouse to nest from the lek of capture at four study areas in northwestern Colorado, 2015.

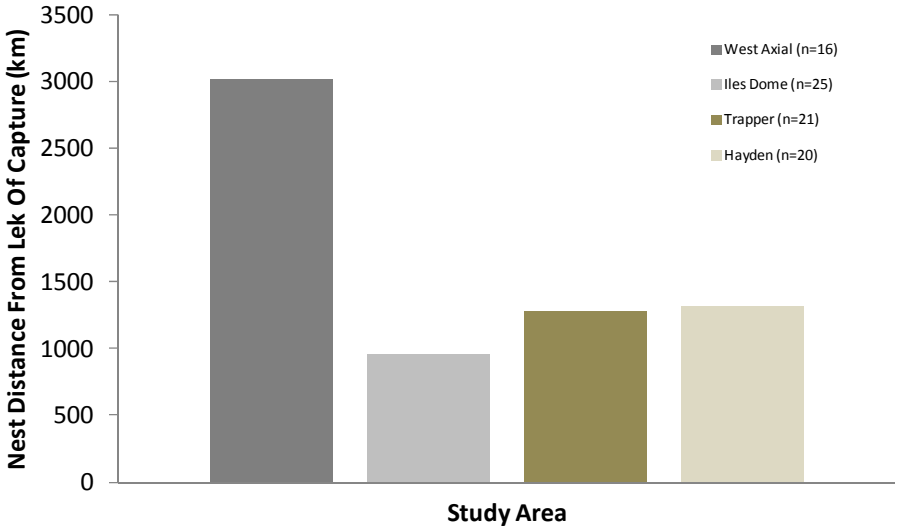

Figure 10. The median distance moved by female Columbian sharp-tailed grouse to nest from the lek of capture at four study areas in northwestern Colorado, 2015.

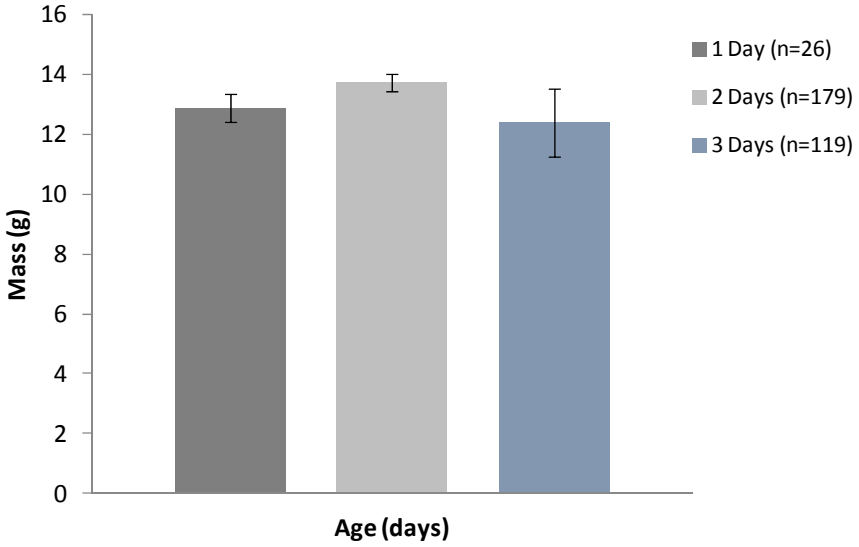

Figure 11. Mean (± SE) mass of Columbian sharp-tailed grouse chicks at 1, 2, and 3 days-of-age in northwestern Colorado, 2015.

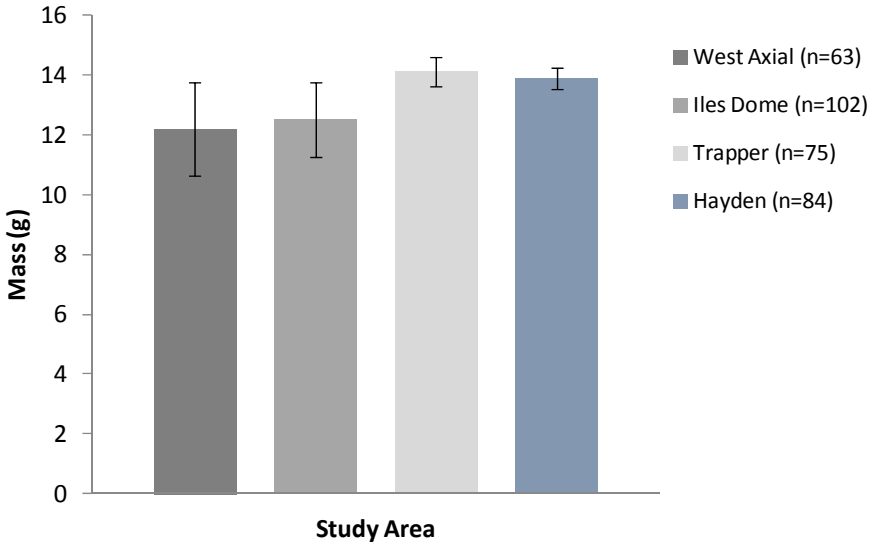

Figure 12. Mean (± SE) mass of 1-3 day-old Columbian sharp-tailed grouse chicks at 4 study areas in northwestern Colorado, 2015.

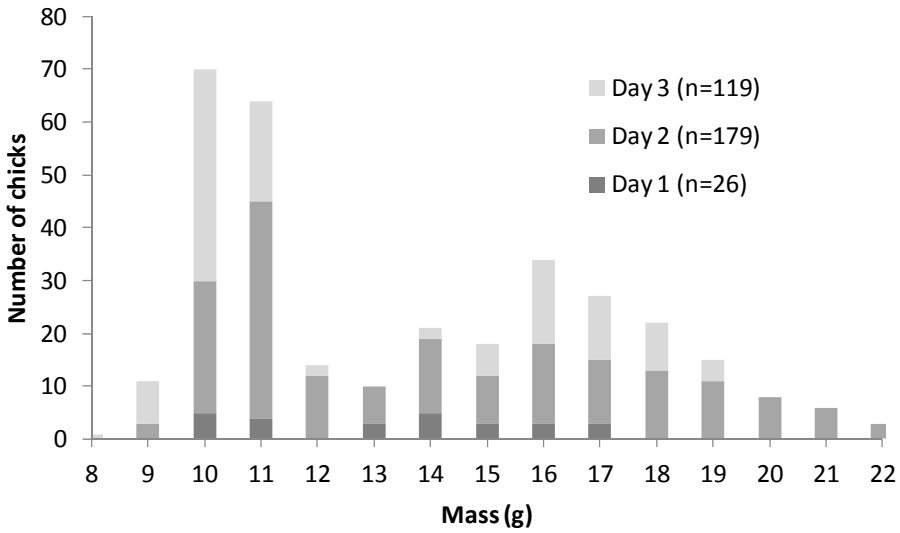

Figure 13. Frequency distribution of the number of 1, 2, and 3 day-old Columbian sharp-tailed grouse chicks by mass in northwestern Colorado, 2015.

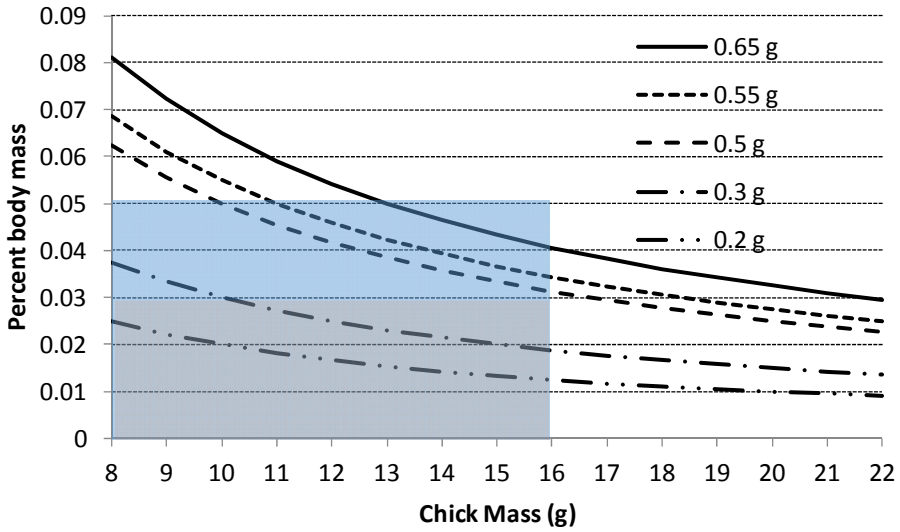

Figure 14. A depiction of Columbian sharp-tailed grouse chick mass and percent of body mass of 5 different micro transmitters.

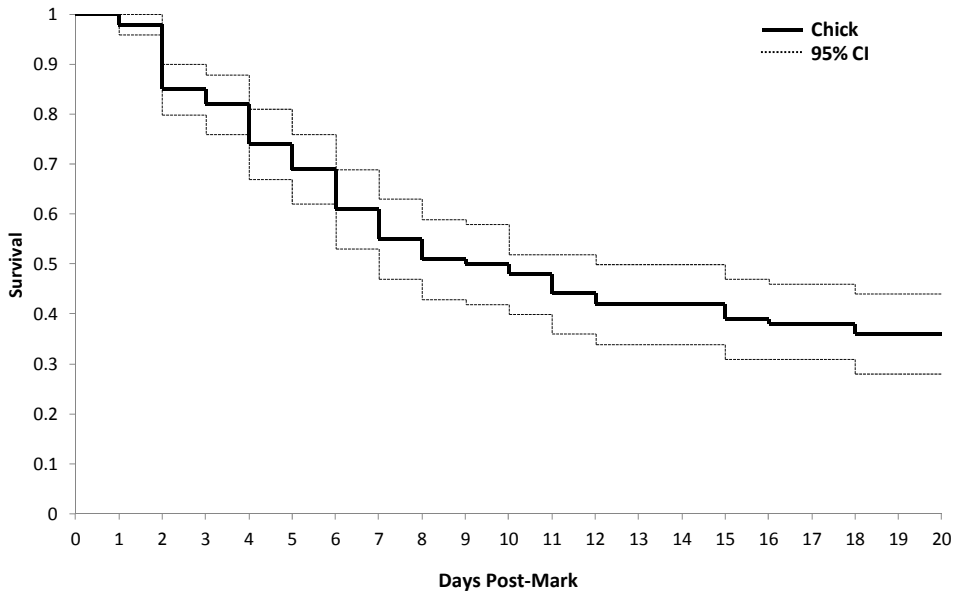

Figure 15. Kaplan-Meier product-limit chick daily survival and 95% confidence interval for 20 days post-mark with 0.65 g chick transmitters in northwestern Colorado, 2015.

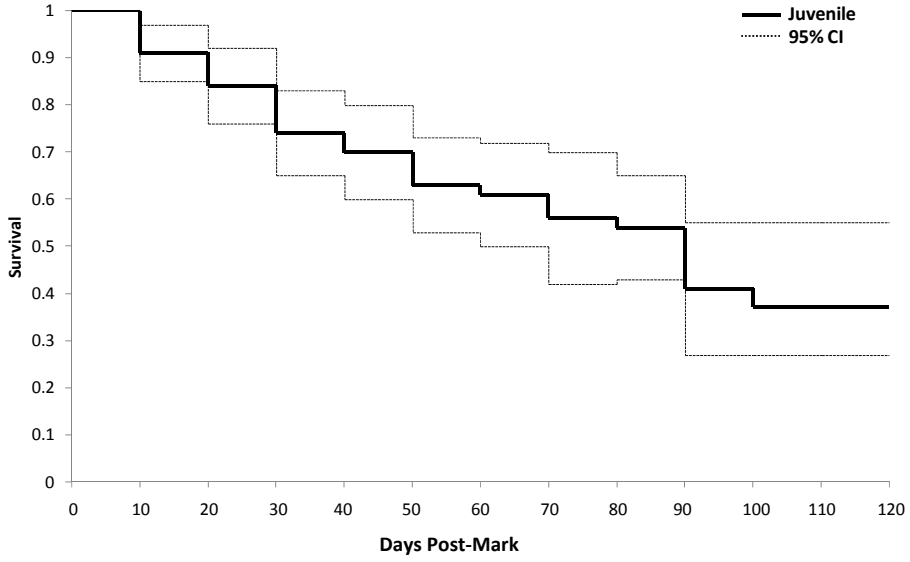

Figure 16. Kaplan-Meier product-limit daily juvenile survival and 95% confidence interval for 120 days post-mark with 3.9 g juvenile transmitters in northwestern Colorado, 2015.

Appendix A

Figure A-1. The 2015 Columbian sharp-tailed grouse field crew. Staff included from left to right, (back row) Nick Rochon, Melissa Maleckar, Shane Petch, Rachel Harris, (front row) Elizabeth Tray, Kiera Kauffman, and Ariana Dickson.

Figure A-2. Male Columbian sharp-tailed grouse conducting breeding display (dancing). Photo courtesy 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 12 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.