Appendix D: Fisher Team Final Report

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Fishers (*Pekania pennanti*) are a medium-sized mammalian carnivore with a pre-European distribution encompassing the boreal forest zone of Canada, the Great Lakes region and northeastern United States, a relatively limited portion of the Rocky Mountains in the United States, and mountainous areas of Washington, Oregon, and California, USA (Powell 1993). Ecologically, fishers are a mature or old forest-obligate species (Zielinski et al. 2005), and in central to eastern Canada and the northeastern United States their numbers were reduced historically by the combination of intensive trapping and loss of forest habitats (Powell 1993, Powell and Zielinski 1994). The species is uncommon to rare in the western United States. It is listed as a sensitive species by the Oregon Department of Fish and Wildlife and endangered by Washington State. In July 2015, the California Fish and Game Commission voted to list the southern Sierra Nevada fisher population as threatened, and the species is currently a candidate for listing under the US Endangered Species Act. In advance of federal and state listing decisions, conservation planning has been underway in California since 2013 to develop an approach to maintaining viable populations of fishers in both northwestern California and in the southern Sierra Nevada. Information from the SNAMP Fisher Project (published manuscripts, submitted manuscripts, and unpublished data) described herein has been included in a Southern Sierra Nevada Fisher Conservation Assessment developed by the Conservation Biology Institute, with input from a team of 13 fisher researchers and scientists.

The SNAMP Fisher Project was initiated by the UC Berkeley Science Team in Fall 2007, in association with multiple other SNAMP science programs, to provide an independent evaluation of how vegetation management, prescribed by the 2004 Sierra Nevada Forest Plan Amendment, affects fire risk, wildlife, forest health and water. A major goal of the SNAMP Fisher Project was to determine whether current rates of survival and reproduction will allow fishers to persist in the Sierra Nevada in the context of active forest management to reduce fuels and the risk of catastrophic wildfire.
Our approach for assessing how fishers would respond to Strategically Placed Landscape Area Treatments (SPLATs) was designed to be multifaceted including (1) life history responses to fuels reduction (changes in survival, reproduction/fecundity, lifespan), (2) changes in local scale habitat use within individual home ranges, and (3) shifts or changes in habitat use at the home range scale of animal resource use/resource selection.

Illustration D2: Fuel reduction management treatments observed in the SNAMP Fisher Study area; mastication/mowing, control burning, commercial thinning

A range of standard methods were used in the study to live-trap, radiocollar and monitor survival status of individual fishers. Monitoring was accomplished almost entirely by fixed-wing aerial radiotelemetry, supported by an “in house” aviation program developed specifically for SNAMP Fisher and administered by the USDA Forest Service. Ground-based radiotelemetry was used to monitor female fishers during denning seasons, and to recover carcasses of deceased fishers. Cameras were systematically placed throughout the study area at the center points of 1-km² grid cells. Grid cells within the SNAMP study area and the key watershed region were surveyed annually, while grid cells outside these areas were surveyed opportunistically. We used the camera survey data to support an occupancy analysis, investigating the impacts of different forest management actions on fisher occupancy, persistence, and extinction.

A total 110 individual fishers were captured and radiocollared from Dec 2007 to Dec 2013 (62 females, 48 males). Sixty-six (60%) of the 110 individual fishers radiocollared during the study were known to have died, including 32 females and 34 males. On average 10.5 radiocollared fishers died in each population year over the course of the study, and the most common cause of death was predation by felid carnivores (bobcats, *Lynx rufus*, and mountain lions, *Puma concolor*). Two radiocollared fisher deaths were roadkills on Highway 41, and five others were directly linked to anticoagulant rodenticides being used in association with illegal marijuana grow sites in the Sierra National Forest.
Seventy-six (85%) breeding-age female fishers either exhibited denning behavior \((n = 63)\) or were determined to have denned and weaned at least 1 kit. Among the 76 breeding-age females that initiated denning, 64 (84%) were identified as weaning kits. Overall, 72% of adult female fishers for which reproductive status was known produced at least 1 weaned kit. We were able to determine litter size for 48 of 59 denning females. A total of 73 kits were known produced, with an average litter size of 1.5.

Fisher population sizes ranged from 48 in 2010 to 62 in 2012, whereas mean population density ranged between 0.072 fishers/km\(^2\) in 2010 and 0.093 fishers/km\(^2\) in 2012. Lambda across all years was 0.90, which was suggestive of general population decline, however, the annual and cumulative 95% confidence intervals all overlapped with 1.0.

Camera surveys were completed in 905 unique 1-km\(^2\) grid cells throughout the overall study area, including 56 grid cells within the southern region of Yosemite National Park. Fishers were detected in 448 of the unique grid cells surveyed, which helped to identify that fishers in this part of the southern Sierra Nevada were most common between 4500 and 6500 feet elevation (1372 and 1981 m elevation). Occupancy estimates for multi-year surveyed grid cells corrected for imperfect detection < 1.0 ranged from 0.62 to 0.80.

Occupancy modelling indicated that fishers reduced their use of forest patches exposed to higher levels of restorative fuel reduction; i.e., persistence of occupancy declined with additional acreage treated for fire resiliency. However, neither restorative nor extractive (i.e., commercial thinning) fuel reduction was related to either initial probability of occupancy or local extinction. We found that SPLATs caused an immediate 6% reduction in potential fisher habitat. However, they also moderated the impact of fire, resulting in greater available fisher habitat within 30 years. In the absence of simulated fire, the amount of habitat steadily increased over time due to forest succession, and was actually slightly greater on the treated landscape in year 30 than in year 0.

The combination of an overall negative population growth rate and the relatively small abundance estimate \((n = 93, \text{ range} 80-107)\), warrants concern for the long term viability of the fishers in the region. Any small population will be at high risk to stochastic events such as disease and large perturbations to critical habitats (e.g., forest fires or drought; Noss et al. 2006), and genetic limitation
resulting from genetic drift after founder events (Tucker et al. 2014) will hinder population recovery and expansion (Reed et al. 2003). Minimum viable population size has been under debate (Shoemaker et al. 2013, Reed and McCoy 2014), but at <500 individuals (Spencer et al. 2015), the current southern Sierra Nevada fisher population will likely require active management and conservation measures to maintain a positive growth rate across its entire range. The estimated population growth rate in the SNAMP Fisher study area reaffirms the vulnerability of the small, isolated population to external threats (Spencer et al. 2015), especially wildfires that are likely to increase in frequency and intensity with climate change. Moreover, the SNAMP Fisher study spanned a limited period of six years during which multiple novel threats to fisher survival within the study area were identified, and when three large wildfires significantly reduced availability of suitable habitat for fishers immediately to the south and north of the study site. We recommend continuous monitoring of the status of fisher populations in the southern Sierra Nevada region. Development of ways to mitigate for major threats to fisher survival and fisher habitats and population viability analyses are necessary for evaluating the long-term prospects for fishers in the southern Sierra Nevada. Data from the SNAMP Fisher study have provided important new insights on the status of a fisher population at the north margin of their current distribution in the southern Sierra Nevada Range, which will be useful towards developing a comprehensive conservation strategy for fishers in California.
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Introduction

Background

Fishers are a medium-sized mammalian carnivore with a historic distribution encompassing the boreal forest zone of Canada, the Great Lakes region and northeastern United States, a relatively limited portion of the Rocky Mountains in the United States, and mountainous areas of Washington, Oregon, and California, USA (Powell 1993). In the west coast states, indigenous fishers currently exist in three remnant populations in southern Oregon, northern California, and the southern Sierra Nevada, California (Zielinski et al. 2005). In California the fisher occupies less than half of its historical range as described by Grinnell in the early 1900s (Grinnell et al. 1937), and the two remnant populations are separated by approximately 400 km. This fragmentation had been considered to be due to widespread timber harvest and fur trapping during the early to mid-1900s (Zielinski et al. 2005), but recent genetic research suggests that the northern California and southern Sierra Nevada populations may have been genetically isolated prior to European settlement (Tucker et al. 2012). Whether this isolation stemmed from extirpation of fishers in the central Sierra Nevada, or from reduced genetic flow due to local topographic obstacles is unknown. Notwithstanding uncertainty regarding the timing or cause of the range retraction, there may be fewer than 500 total fishers in the southern Sierra Nevada population (Spencer et al. 2011), where the species currently occupies approximately 4,400 km² of mid-elevation, mixed-coniferous forest (Spencer et al. 2015).

In October, 2014 the US Fish and Wildlife Service proposed listing the West Coast Fisher Distinct Population Segment, meaning fishers in California, Oregon, and Washington, as threatened under the federal Endangered Species Act (http://federalregister.gov/a/2014-23456). The public comment period on the proposed rule closed in May, 2015, and a final decision is scheduled for April, 2016. The fisher is currently listed as state endangered by the Washington Department of Fish and Wildlife. In California and Oregon, the fisher is currently a candidate species for listing under the state Endangered Species Acts, and on August 5, 2015 the California Fish and Game Commission voted to list the southern Sierra Nevada fisher population as threatened.

Fisher Range and Population Trends

Grinnell et al. (1937) described the original range of the fisher in California as including the
entire western slope of the Sierra Nevada, the southern Cascades, Klamath Mountains, and northern Coast Range, a total area of ~100,000-110,000 km² (Spencer et al. 2015) (Fig. D1). Lofroth et al. (2010) estimated that the current range of the fisher in California represents <50 percent of the historical range, and fishers are currently absent from most of the northern and central Sierra Nevada, leaving a ~400-km gap separating the two remnant populations in the state (Zielinski et al. 1995) (Fig. D1), one in the northern Coast Range and one in the southern Sierra Nevada (Spencer et al. 2015). Spencer and Zielinski (in review) used an updated fisher locality database to estimate their current geographic range in California at 55,000-60,000 km², with ~45,000-50,000 km² in northern California and 10,000-12,000 km² in the southern Sierra Nevada. Although the range areas estimated by Spencer and Zielinski (in review) included a mix of suitable and unsuitable habitats, the analysis suggested a 30-50 percent reduction compared to the historical range of the species.

The southern Sierra Nevada fisher population is small (~500 total individuals and <300 adult fishers; Spencer et al. 2011), but appears to be stable over about the past decade (Zielinski et al. 2013). Following substantial population contractions in the past (Knaus et al., 2012), fishers in this part of California may have expanded in the late 20th century (Tucker et al. 2014). The overall distribution of fisher in the southern Sierra Nevada has been monitored using a combination of track plates and motion detecting cameras since the mid-1990s (Truex et al. 1998, Zielinski et al. 2005, Jordan 2007). Zielinski et al. (2013) analyzed occupancy records from this effort for the period 2002 to 2012, when a systematic survey design was
in place, and found no detectable change in occupancy for the entire area or for any of the three subareas examined. Furthermore, the authors stated that “Constant and positive persistence values suggested that sample units rarely changed status from occupied to unoccupied or vice versa” (Zielinski et al. 2013). However, this evidence can be interpreted one of two ways. On one hand, the lack of a decreasing trend may indicate a small but stable population. On the other, the lack of an increasing trend may indicate that despite fishers being protected from fur harvest for over 60 years during a time when large scale clearing of forest habitat was diminished (Collins et al. 2010), the population isolate is not showing evidence of recovery. Interpretation is further complicated by a combination of genetic patterns and survey data suggesting that the population north of the Kings River may have expanded during the 1990s, before the regional monitoring program was established (Tucker et al. 2014).

Insight from prior research in the High Sierra District, Sierra National Forest (KRFP study, ≈60 km southeast of the Key Watersheds) suggests that fisher population densities range from 0.07 to 0.28 fishers/km² (Jordan et al. 2011, Thompson et al. 2012). Records from research in northern California (Hoopa Study) indicate the potential for fisher densities to change rapidly. In the Hoopa Valley area of Northern California, fisher densities were estimated at 0.52 fishers/km² in 1998, but fell to 0.14 fishers/km² in 2005 (Matthews et al. 2013). Due to the apparent variability in density estimates, developing precise density estimates for different subpopulations and in different habitat types is critical for effective management.

**Management and Conservation Planning**

Federal and state resource agencies are currently developing strategies to aid in the maintenance of viable populations of fishers in both northwestern California and in the southern Sierra Nevada. It is possible that the isolated population of fishers in the southern Sierra Nevada will be impacted as the USDA Forest Service implements fuel reduction measures (Strategically Placed Land Allocation Treatments; SPLATS) to mitigate risk of catastrophic wildfire (Scheller et al. 2011). Fuel reduction treatments are becoming the dominant forest management activity in western forests in response to increases in the frequency of intense, stand-replacing forest fires over the past several decades (Mallek et al. 2013, Safford 2013). Advances in fire modeling have greatly improved managers’ ability to plan and evaluate various landscape fuel treatment scenarios intended to reduce fire risks (Collins et al. 2010, Scheller et al. 2011). However, there remains a considerable gap between
modeling landscape-scale fuel treatments and implementing them due to concern over the status of rare and uncommon species associated with multi-storied, late-seral stage forests, such as the fisher and spotted owl (*Strix occidentalis occidentalis*) (Naney et al. 2012, Truex and Zielinski 2013). Presence of fishers has strongly influenced managers’ ability to delineate landscape-scale fuel treatments in this fire-prone region (Collins et al. 2010, Thompson et al. 2011). The Sierra Nevada Forest Plan Amendment (SNFPA) represents the most recent attempt to reconcile the need to reduce fuel loadings in Sierra Nevada mixed-conifer forests and retain characteristics of late-successional forests that are important for these species. The strategy involves a network of “Strategically Placed Area Fuel Treatments” (SPLATS) that allow up to a 60% reduction in basal area and a 30% reduction in canopy cover in Sierra mixed conifer forest. In the long-term, this strategy may increase availability of important habitats for species such as the fisher and California spotted owl by reducing wildfire-induced losses (Spencer et al. 2011), but treatments may also impact habitat quality for fishers in the near-term (Thompson et al. 2011).

To provide a framework for balancing the habitat needs of fishers with fuel treatments intended to reduce fire risks, SNAMP initiated a coordinated effort to assess the effects of fuel treatments on many environmental features including the fisher, spotted owl, forest health, and water quality and quantity in the central Sierra Nevada. SNAMP began in 2007 and was designed to evaluate the effects and effectiveness of fuel treatments implemented according to the revised Sierra Nevada Framework (USFS 2004) under a design that incorporated stakeholder participation. SNAMP is a landscape-scale, ecosystem-level experiment in natural resource management and involves a Before-After-Control (BACI) design developed specifically to assess the impacts of SPLATs on the overall forest ecosystem (Popescu et al. 2012).

**Study Goals and Primary Objectives**

1. Estimate population parameters including age and sex-specific survival, and fecundity
   a. What are the vital rates (reproduction, survival, population growth rates)?
   b. What is the population size and density in the study area?
   c. What are the patterns of dispersal movements?
2. Identify population limiting factors in the region encompassed by the study area
   a. What are the causes of mortality? Are predators, parasites or diseases important?
   b. What are the reproductive rates?
c. What are the patterns of home range, dispersal, and habitat use?

3. Evaluate effects of SPLATS on occupancy, survival and fecundity
   a. Characterize resource use by fishers; do SPLATs influence habitat use
   b. What are the patterns of fisher occupancy in relation to forest management?
   c. Do patterns of fisher occupancy change before and after by SPLATS?
Site Description and Study Area

The SNAMP Fisher Project study area is at the northern end of the southern Sierra Nevada fisher population in California, encompassing the area bounded by the Merced River in the north and the San Joaquin River in the south (Fig. 2). It consists of three nested landscapes: the SNAMP study area (34 km²), the key watershed region (128 km²), and the SNAMP Fisher study area (1300 km²). We expanded into the key watershed region to facilitate an evaluation of the impacts of fuel and vegetation management on fisher survival, habitat selection, and reproduction. The key watershed region

Figure D2: Overall Study Area of the SNAMP Fisher Project, including administrative boundaries and the outer boundary of the “Key Watersheds” focal study area in the approximate center of the map region.
encompassed three Forest Service projects expected to occur in the study period near the communities of Fish Camp (Fish Camp Project), Sugar Pine (Sugar Pine Project), and Cedar Valley (Cedar Valley Project). The four Key Watersheds are the Sugar Pine, Nelder Creek, Rainier Creek and White Chief Branch watersheds (Fig. D3).

Due to the large movement capacity and space use of fishers, further expansion into the SNAMP Fisher study area was necessary to reach and maintain the stated goal of monitoring 20 radiocollared animals at all times to obtain more precise estimates of demographic rates. This larger study area encompasses a mix of public and private land and is topographically complex with elevations ranging from 758 m to 2652 m. Administratively, the focal study area for the study is the non-wilderness region of the Bass Lake Ranger District in the Sierra National Forest, however it extends into the southern portion of Yosemite National Park. Field work was carried out between 1,000 m and 2,400 m in elevation, corresponding to fisher occurrence in the region. This elevation gradient corresponds with a mix of hardwoods (California bay \textit{[Umbellularia californica]}, Canyon live oak \textit{[Quercus chrysolepis]}, CA black oak \textit{[Quercus kelloggi]}, and several conifer species at lower elevations (ponderosa pine \textit{[Pinus ponderosa]}, incense cedar \textit{[Calocedrus decurrens]}; California Wildlife Habitat Relationship system MHW, PPN, and MHC habitat types). Between 1300 and 1900 m habitat consists of a mix of multiple conifers (Jeffrey pine \textit{[P. jeffreyi]}, white fir \textit{[Abies concolor]}, incense cedar), and hardwoods (black oak, white alder \textit{[Alnus rhombifolia]}, mountain dogwood \textit{[Cornus nuttallii]}; CWHR Habitat types SMC, MHC, PPN). Above 1900 m habitat transitions into red fir \textit{[Abies magnifica]} and lodgepole pine \textit{[P. contorta]} (CWHR Type RFR). Common shrubs and tree-like shrubs include whiteleaf manzanita \textit{[Arctostaphylos viscida]}, greenleaf manzanita \textit{[Arctostaphylos patula]}, mountain misery \textit{[Chamaebatia foliolosa]}, bush chinquapin \textit{[Chrysolepis sempervirens]}, mountain whitethorn \textit{[Ceanothus cordulatus]}, and snowberry \textit{[Symphoricarpos mollis]}. Giant sequoia \textit{[Sequoiadendron giganteum]} are present, but primarily restricted to the Nelder Grove Historic Area within the Nelder Creek watershed. Permanent streams in the Key Watersheds are important for fishers and other wildlife and include Big Creek and Rainier Creek in the Rainier Creek watershed, Lewis Creek in the Sugar Pine watershed, and California Creek and Nelder Creek in the Nelder Creek watershed.
Methods

Vital Rates and Basic Population Parameters

Field Methods

Live Trapping.—Although noninvasive methods can be used to generate important data on wildlife populations (Long 2008), estimating vital rates (survival, reproduction, dispersal) almost always requires trapping to radiocollar and then closely monitor the study animals. We followed...
standard live-trapping procedures previously developed for fishers in California (Jordan 2007, Matthews et al. 2013a), with only a few minor changes. Individual fishers were live-captured in steel mesh traps (Tomahawk Live Trap Company, Tomahawk, WI) modified to include a plywood cubby box to provide the animals with a secure refuge where they were less likely to injure themselves (Wilbert 1992). Trapping to mark animals with radiocollars was focused during the fall and winter seasons between December 2007 and March 2012. Also, with the exception of the first year of the study when we needed to capture fishers to initiate the study, we did not trap during the spring denning period (late March to mid-June) to minimize disturbance to reproduction. Live traps were baited with venison, and checked daily by late morning. Captured animals were restrained in a handling cone, and sedated using a mixture of Ketamine hydrochloride and Diazepam (1 mg Diazepam/200 mg Ketamine) injected intramuscularly. Sedated fishers were weighed, classified by age and sex based on examination of teeth and genitalia, and measured for standard morphological features. Small samples of ear tissue were collected for microsatellite DNA analysis using a sterile dermal biopsy punch. Several strands of hair were removed from the nape and rump region, also for DNA analysis. Hair samples were stored in a dry paper envelope, whereas tissue samples were stored in 95% ethanol until analysis at the USDA Forest Service Wildlife Genetics Lab (Rocky Mountain Research Station, Missoula MT). Teats on females were measured for base diameter and height using digital calipers (±1mm), and those data were used to identify females that weaned at least 1 kit when they had not been monitored during the denning period (Matthews et al. 2013b). Each animal was permanently identified by subcutaneous insertion of passive integrated transponder (PIT) tags (Biomark, Boise, ID), and fitted with a radio collar (Holohil Systems Model MI-2M, Ontario, Canada) modified by attaching small bands (0.5-1.0 cm) of infrared reflective tape (3M® Scotchlite™) along the lengths of the antennas. Custom breakaway devices were inserted into radiocollars fitted to juvenile fishers to reduce the risk of injury or strangulation between recaptures (Thompson et al. 2012). Bands of infrared reflective tape and breakaways were modifications, not used in previous studies. After handling, we returned animals to the cubby box and released them at the point of capture after recovery from anesthesia. Capture and handling procedures followed American Society of Mammologist guidelines (Sikes and Gannon 2011), and were approved by the Institutional Animal Care and Use Committee of the University of California, Berkeley (protocol R139).

Live-trapping is labor intensive, and the effort was designed to gain advantage from detections of non-collared fishers at cameras. Live traps were most frequently placed in the same area of camera
stations after cameras had been removed (to prevent interference with camera surveys). Data from camera detections were used to design linear traplines of 5-10 traps bracketing positive detection stations. Distance of separation between traps was typically ≥500 m, and traplines were usually successful at capturing targeted animals within five nights of trapping. Live-trap success was further enhanced in later years of the study by placing traps in locations where fishers had been captured in the past. Trap success was also enhanced by cleaning and sanitizing traps after captures. In winter, snow falling from tree branches can ice up the treadle mechanism inside live traps. We used lightweight, rectangular canvas tarps to protect the inside of the live traps from falling snow, and debris used to camouflage the traps. Traplines were generally removed the day after targeted fishers were captured, and always after 10 nights of trapping when no fishers were captured.

Illustration D4: Camouflaged live trap near the base of a white fir tree, and a radiocollared fisher being released after processing.

Aerial Telemetry and Radiotelemetry Monitoring.—Tracking radiocollared animals from an aircraft is an alternative to locating them from the ground by homing or triangulation (Thompson et al. 2012). Researchers have been using fixed-wing aircraft to locate wildlife since the early 1970s (Mech 1974). The unique ability of observers in aircraft to rapidly search and locate radiocollared animals over large and inaccessible areas while allowing for nearly line of sight reception between transmitter and receiver makes aerial radio telemetry an effective research technique in general (Gilmer et al. 1981), and specifically for studying fishers, which often occur in remote mountainous areas where access can be difficult (Weir and Corbould 2008). Partly for these reasons, we used fixed-wing airplanes to monitor and relocate radiocollared fishers for the entirety of the SNAMP Fisher Project. Beginning in December 2007, we worked with USDA Forest Service Supervisory Pilot John Litton to
develop an aviation program in support of SNAMP Fisher, which was fully established in August 2008 when a full time pilot was hired and the first of two dedicated aircraft were based at the Mariposa-Yosemite Airport in Mariposa, CA.

The two USDA Forest Service-owned aircraft acquired for supporting the project were a Cessna 185 (Cessna Aircraft Co., Wichita, KS) and a Piper PA-18 Super cub (Piper Aircraft Inc., Vero Beach FL). Two aircraft were considered necessary to maintain continuous monitoring of radiocollared fishers when routine maintenance or engine repair was necessary (John Litton, personal communication).

Illustration D5: Forest Service-owned Piper Supercub (left) and Cessna 185 (right) on the tarmac at the Mariposa Airport, California.

Illustration D6: Forest Service Cessna 185 airplane, and antenna configuration on the right side wing strut.

The optimal search procedure used when locating animals from light aircraft varies depending on the number of animals tracked, and the antenna configuration supported and approved for the airplane being used (Gilmer et al. 1981). Additional details are provided elsewhere (Thompson et al. 2012), but we used two, 2-element H antennas (Telonics Inc., Mesa, AZ) mounted in a sideways configuration on each wing strut, and a single 3-element Yagi antenna (Advanced Telemetry Systems, Isanti, MN) mounted forward-facing on the right wing strut. This antenna configuration was effective in allowing the pilot and biologist to search for radiocollared fishers using the Yagi antenna (detection
range 5-20 km), and then switching to the H-antennas to localize to a relatively precise location above the animal using a circling technique (Seddon and Maloney 2004).

Fixed-wing flights (aerial telemetry missions) to locate radiocollared fishers in the study area were scheduled in advance for 4-6 missions/week, depending on weather conditions considered safe for departure and return to the Mariposa-Yosemite Airport. Flights typically occurred in the morning hours, and lasted 2-3 hours. Afternoon telemetry flights were relatively infrequent, and the large majority of aerial radiotelemetry locations were acquired in the AM period of the day. As part of each aerial-telemetry mission, we systematically searched for all active radiocollars deployed on fishers in the study area. Biologists in the airplane recorded (1) active/inactive status for each fisher, (2) time of location, (3) an estimated UTM location for each fisher (typically logged into a handheld GPS unit; Garmin 60 CSx, Olathe, KS), (4) a qualitative ranking for each location (poor, fair, good, excellent), and (5) a record of any radiocollared fishers that were not located. Additional descriptive details were often recorded that related to the nature of weather conditions influencing the aircraft at the time of the location (turbulence, “bumpy”, clouds occluding visibility to the ground, etc.), or if the animal had moved an unusual distance or to an atypical area. At the end of each aerial telemetry mission, the biologists summarized details on departure and return times and weather and flight conditions during the flight.

Aerial radiotelemetry can be efficient for locating animals that range over large areas in difficult terrain (Gilmer et al. 1981), but the accuracy, or precision of aerial telemetry locations is generally less than for ground-based radiotelemetry (e.g., triangulation; Koen 2005, Gantz et al. 2006). Location error from fixed-wing airplanes varies with flight speed, elevation above ground level, pilot and biologist experience, and signal reflection in rugged topography (Thompson et al. 2012). We assessed error for aerial radiotelemetry locations on the SNAMP Fisher project by calculating the Euclidean distance between GPS locations logged by biologists in the airplane and positions of test collars placed at known locations on the ground. Test collar locations were generally radiocollars that were placed in locations unknown to the biologist in the airplane; biologists were required to regularly estimate positions for test collars during aerial telemetry missions. Other aerial radiotelemetry locations used to quantify accuracy included dropped radiocollars, carcass locations, fishers in live traps, and female fishers in a cavity in a den tree whose locations were also unknown to the biologist in
the airplane.

Fisher Reproduction

*Background.*—Den sites, where female fishers bear and raise their kits, are a critical habitat element for fisher populations in California. Females typically use more than one den during the denning season (late March to mid-June). Natal dens are where adult female fishers give birth and initially care for young, and they may then move kits to one or more maternal dens from early April to June until they are weaned (Powell et al. 2003, Matthews et al. 2013a). Reproductive dens, both natal and maternal, are nearly always cavities in large trees, live or dead, and are found in forest stands with dense canopy cover and complex multi-layered structure (Zhao et al. 2012). Suitable denning sites are likely a subset of suitable resting sites because the requirements are more stringent: (1) den cavities must be large enough to shelter both mother and kits for weeks rather than days; (2) the female needs to provision her young while they are restricted to the den, so dens must be located close to high-value foraging areas; (3) cavity entrances must be small enough to exclude males; and (4) denning begins in late March-early April, when temperature are colder and slope position may be more critical in assisting with kit thermoregulation.

*Identifying den trees and evidence of reproduction.*—Female fishers exhibiting behavior consistent with denning were identified during late March-mid April and then monitored. Denning behavior was characterized by an abrupt change from a pattern of successive aerial radiotelemetry locations being dispersed within a female’s home range, to a pattern where locations were spatially clustered (3-5 locations within 500 m over a 7-day period; Zhao et al. 2012). When clustering of locations occurred, a biologist navigated to the area with a handheld Global Positioning System device to investigate. Standard ground-based radiotelemetry techniques with a handheld receiver (model R1000; Communication Specialists, Inc. Orange, California) and an H-type antenna were then used to home towards telemetry signals of radiocollared females. Once a collared female was isolated in an area, the biologist circled the fisher until the individual tree or snag was identified (Matthews et al. 2013a). When female fishers were localized to a possible den structure, 2-4 automatic “den cameras” that had been cleaned and de-scented were attached to nearby trees and focused on the bole of the den structure (scent and bait lures were not applied around den trees to avoid attracting other predators). We returned to these structures the day after initial placement of den cameras, and then every 3-5 days
to confirm use for denning based on regular occupancy and images indicating up and down movements on the tree or snag. Trees and snags used ≥3 times in succession and with camera-based evidence of up-down movements were considered denning structures (Zhao et al. 2012). We defined “denning opportunities” as the total number of individual, breeding-age female fishers (≥24 months) either directly monitored in mid-March to June (Matthews et al. 2013a), or measured for teat size during July to January to assess weaning status (Matthews et al. 2013b). We considered kits weaned when denning behavior continued until 31 May or later (Matthews et al. 2013a).

Activities of known-denning female fishers were chronicled for the duration of each denning season by continuous monitoring with cameras and ground checks of den trees. Female fishers typically transfer kits from natal den trees in which they were born to 1-6 other maternal dens during April to June (Matthews et al. 2013a). Each time we had evidence that a denning fisher moved kits to a new maternal den (images of females transporting kits away from den trees, cessation of occupancy over multiple checks), we searched for the female using ground telemetry and repositioned cameras around the next den structure (Zhao et al. 2012). Den cameras were removed in mid-June when females ceased localizing to den structures.

Information on litter size was determined from images from den cameras, or, less frequently, by climbing den trees and using a video camera (Peep-A-Roo Video Probe System, Sandpiper Technologies, Manteca, CA) to count kits inside den cavities (Matthews et al. 2013a). We minimized disturbance to denning females by (1) restricting visits to den structures to service cameras to once every 3-5 days, (2) using deployments of multiple den cameras for obtaining the majority of kit counts,
and (3) by not approaching den trees for climbing until ground-based telemetry indicated the female was well away from the den structure (Zhao et al. 2012).

Maximum reproductive rate was estimated as the sum of the number of adult-age female fishers in the population that localized to den trees during the den season (Mar 21 to Jun 20), plus the number of adult females with enlarged teats that were not monitored but captured and measured before January, divided by the number of adult-age female fishers in the population during mid-March to late January. Weaning rate was estimated as the number of adult-age females known to have survived and localized to den trees through May plus those with enlarged teats that were captured after the den season, divided by the number of adult-age female fishers in the population during mid-March to late January. We note that measurements of teat size were shown to correctly identify over 90% of current year reproducing females that weaned at least 1 kit, and all but 3 adult females for which teat measurements were used to determine reproductive status were part of the Matthews et al. (2013) dataset. Annual estimates of fecundity were calculated as the number of female offspring produced: proportion of adults weaning kits * average weaned litter size * 0.5 (assuming equal sex ratio).

Habitat Characteristics within Fisher Denning Areas. --Denning structures are considered a limiting habitat element for fishers in California and elsewhere (Weir et al. 2012), but site characteristics immediately surrounding the denning structures may also limit fisher use of a site for denning (Zhao et al. 2012). Current forest vegetation management to reduce hazardous fuel levels, improve the vigor of selected trees (pines and oaks), increase spatial heterogeneity, and provide forest products for society may impinge on denning habitats in a variety of ways (Naney et al. 2012, Powell and Zielinski 1994). These management actions can negatively affect fisher habitat (Weir and Corbould 2008), at least in the short term (Thompson et al. 2011), while others may have little impact on fisher habitat suitability (Spencer et al. 2015). Without detailed “local scale” information on habitat characteristics directly associated with fisher denning structures, it will not be possible to adequately manage Sierra Nevada forests in ways that will maintain viable fisher populations.
Habitat characteristics for denning structures.—We developed a protocol for determining the combination of biotic and abiotic characteristics female fishers are likely selecting/using for denning habitats. The protocol was designed to collect similar types of data as those being recorded by the Forest Health Team on the Core Plots in the Sugar Pine area, while also capturing the same types of data being recorded by the USDA Forest Service PSW Kings River Fisher Project at den trees used by fishers in the High Sierra District, Sierra National Forest. Full details on how different habitat data were assessed are provided in Appendix D1. Briefly, we used an 18m radius circular plot centered on the denning structure (Fig. D4) to organize collection of data on (1) canopy cover, (2) litter, duff, and coarse woody debris (associated abundance of fuels), (3) cover and height of herbaceous plants and understory woody shrubs (concealment cover), (4) slope and aspect, and (5) size, number, and height of trees and snags (3 size classes, 4 crown classes). Data on habitat characteristics for den trees were typically collected during late spring or summer, and always when the den trees were no longer in use for denning.

Figure D4: Diagram illustrating the layout of 18m radius circular plots and associated measurement transects for fisher denning structures for the SNAMP Fisher Project in the Bass Lake District, Sierra National Forest.

Fisher Survival

Background.--Understanding survival and the details of cause-specific mortality is fundamental for insight into the population biology of any species, and crucial for understanding the limits to population growth and recovery for rare or endangered species of wildlife. Historical loss and fragmentation of important habitats, combined with overexploitation by hunting and trapping are the
most common drivers of endangerment of wildlife (Lande 1993). Although changes in management may sometimes succeed in reversing problems associated with loss of critical habitats, emergence of new threats that impinge on survival or reproduction can counteract improvements that might otherwise reverse population declines. Emerging threats to survival and population persistence may be obvious such as exposure to novel pathogens and increased occurrence of road-kill deaths (Gaskill 2013, Litvaitis and Tash 2008), or less discernible and linked to changes in community structure or composition that produces an imbalance in predator-prey relations (Roemer et al. 2001).

Factors that contribute to limited growth and expansion of fisher populations in the southern Sierra Nevada are likely linked to a combination of resource and population phenomena. Fishers may be challenged by limited access to suitable resting and denning habitats (Purcell et al. 2009) or insufficient numbers of prey (Zielinski and Duncan 2004), whereas survival may be reduced by high rates of predation (Wengert 2013), wildlife-vehicle collisions (Chow 2009), and exposure to anticoagulant rodenticides (Gabriel et al. 2012a, Thompson et al. 2013). Although habitat requirements of fishers and their responses to forest management are increasingly well known (Aubry et al. 2013, Garner 2013, Zielinski et al. 2013), it has only recently been documented that high numbers of otherwise healthy fishers were succumbing to attacks by other forest carnivores (Wengert et al. 2014) and that illegal use of anticoagulant rodenticides and other toxicants associated with illegal marijuana grow sites on national forests and parks in the southern Sierra Nevada was contributing to both direct mortality and reduced survival of fishers in this region (Gabriel et al. 2012a, 2013, Thompson et al. 2013). Because of heightened concern over the stability of the small population of fishers in the southern Sierra Nevada, our primary objective was to evaluate factors contributing to variation in survival among fishers in the region. Young mammals (particularly males) often experience higher mortality early in life associated with dispersal and establishing independent home ranges (Chepko-Sade and Halpin 1987), and general naiveté with predators and other environmental risks (Farias et al. 2005, Murdoch et al. 2010). Fishers typically disperse before they reach maturity at \( \approx 24 \) months (Arthur et al. 1993), and may suffer lower subadult survival rates as a result. Fishers may also experience lower survival during fall and winter due to the combined effects of higher energetic costs associated with movements in deep snow cover (Powell 1979) and prey limitation when several species of rodent prey utilized by fishers (Zielinski and Duncan 2004) enter into torpor. Therefore, we hypothesized that (1) survival would be lower for juvenile and yearling fishers compared to adults, (2) males would experience lower survival than females related to higher rates of
movement and potentially longer dispersal distances, and (3) survival would be lower during fall and winter than in spring and summer.

**Determination of survival rates.**—We monitored the status (alive, dead, or missing) of radio-collared fishers from time of first capture until death, censorship (due to dropped or failed collars on animals that were not quickly recaptured), or the end of the study. Breakaway devices in the radio-collars occasionally resulted in premature detachment, requiring efforts to re-collar animals that were short-term missing (1-2 months). Because of the relatively common incidences when animals were missing for less than one month, we evaluated survival on monthly intervals rather than weekly or bi-weekly. Overall patterns of survival were determined using the Kaplan-Meier (KM) staggered entry method (Koen et al. 2007, Pollock et al. 1989, Price et al. 2010). KM models were used to produce estimates for annual survival and combined year survival (data pooled by month across all years). The population year was defined as April 1 to March 31 based on the timing of reproduction for female fishers in California with most offspring produced from March 21 to early April (Matthews et al. 2013a). Annual survival can be moderately to highly variable and may result in a negative population growth trajectory that may not be appropriate for a long-lived species with a generation time of two or more years. We therefore smoothed survival estimates by grouping data into 2-year increments (e.g., population years 2 and 3, population years 3 and 4, etc.) and generating KM estimates for each 2-year increment. Live-trapping to capture young-of-the year juveniles was focused during mid-October to February (a few juvenile fishers were occasionally captured before October or in early March). Kaplan-Meier models to estimate “annual” survival for juveniles were typically initiated in December, thereby producing survival estimates for juveniles for a 3-4 month period from December or January to March. When data for juveniles were pooled across population years, however, the dataset allowed for evaluating juvenile survival for the 6 month period from October to March. Z-tests were used to compare estimates for combined year survival for all possible age and sex combinations. Significance levels (α) for multiple comparisons were adjusted for Type I error rates using the Bonferroni procedure (McCann et al. 2010).

**Causes of Mortality**

*Background*—In addition to the challenges described previously, it has been hypothesized that changes to more open canopy forest conditions with an understory of small trees and more shrubs, following either management or wildfire, is contributing to higher rates of predation by bobcats (*Lynx*...
rufous) and coyotes (Canis latrans) (Wengert 2013). Although the habitat requirements for fishers are generally known (Lofroth et al. 2010), details of cause-specific mortality in the southern Sierra Nevada had not been rigorously evaluated until the SNAMP and KRFP studies were initiated in 2007.

Monitoring to detect mortality.-- All radio-collars fitted to fishers on the SNAMP study were equipped with either mortality or activity sensors, internal mercury switches that change the pulse rate when the collar is stationary for over 8 hours (mortality switch) or active (activity sensor). These sensors allowed us to detect inactive signals, investigate fisher mortalities, and recover carcasses soon after death in most cases. When a mortality signal was detected, immediate attempts were made to locate the collar and recover either the carcass or the dropped radiocollar. Carcasses were generally recovered within 24 hours of death.

Radiocollared fishers have been monitored effectively and relocated by intensive ground-based radiotelemetry as part of the KRFP study centered approx. 60 km southeast of the SNAMP Fisher Study Area (Garner 2013). However, most prior studies have been unable to identify causes of death for many deceased study animals because carcasses were not retrieved within 12-48 hours after death (Truex et al. 1998, Aubry and Raley 2006, Jordan 2007). Decomposition begins immediately after death, which can prevent identification of underlying disease processes (Gabriel 2013, Keller et al. 2012), and scavenging can mask both the cause of death and the responsible predator (Wengert 2013). Because of this, our primary rationale for monitoring radiocollared fisher by fixed-wing aircraft up to 6 days/week was to recover carcasses of animals as soon after death as possible. The protocol that was in place from the start of the study until approx. June 2012 was for the biologist in the airplane to use the audio system in the airplane to (1) transmit the estimated location coordinates for any radiocollared fishers detected on inactive pulse to the SNAMP fisher office, whereupon (2) a staff biologist in the vicinity would immediately investigate the location and recover the carcass following an approved forensic protocol, (3) transport the carcass to the SNAMP fisher office where they were placed in -20 C freezer for storage until (4) a necropsy could be scheduled at the UC Davis School of Veterinary Medicine.

Once a radiocollar is activated and deployed, it will typically function (emit a radio signal) for 18-24 months until the battery is expended. In 2011 the SNAMP Fisher project began using radiocollars from Advanced Telemetry Systems (ATS), but discovered that many of the electronic
“mortality switches” built into the ATS radiocollars became defective within 8-10 months of being deployed on study animals. Electronic mortality switches are designed to emit pulses at twice the normal pulse rate when the radiocollar has been stationary for at least 8 hours. When the mortality switches in the ATS brand radiocollars became defective they began to emit intermittent and then consistent false inactive signals. SNAMP Fisher was forced to modify the inactive signal protocol by first plotting locations determined for inactive signals in ArcGIS, whereupon a decision on whether to investigate the location was based on a judgment of the distance of separation between successive aerial radiotelemetry locations (investigation triggered when successive locations were <1000 m apart). The revised protocol was situation specific: the first time a collar was detected emitting an inactive signal, efforts were made to investigate the location immediately. Subsequent inactive signals from that collar were examined carefully prior to on-the-ground investigation. This process may have delayed the recovery of a limited number of carcasses. Efforts were made to correct for the problem of false inactive signals by replacing defective collars with collars of a different manufacture as soon as possible.

When fisher carcasses were discovered we followed a standardized forensic protocol for collecting samples and documenting evidence at mortality sites using photographs and diagrams (Wengert et al. 2013). When predation was suspected as the cause of death (e.g., obvious punctures, partial consumption), we recorded information on the characteristics of the predation event including patterns of consumption and evidence of caching or burying. Samples included swabs of visible bite wounds, clipped fur from near the bite wounds (clipped to avoid fisher DNA in root bulbs), swabs of the claws and teeth, and any non-fisher hairs left on or near the carcass (Wengert et al. 2013). Carcasses were double-bagged in plastic bags, labeled, and transported back to the field offices where they were frozen in a -20°C freezer until being shipped to University of California, Davis for necropsy.

Pathology.--We submitted all carcasses for necropsy and disease and DNA assessment to cooperating pathologists at the University of California Davis, Veterinary Medical Teaching Hospital, and California Animal Health and Food Safety Laboratory in Davis, CA. When possible, the team of pathologists determined cause of death for each fisher using all available information, including necropsy examination, disease and toxicological results, DNA forensics, evidence recovered or identified as important from the mortality site, and habitat characteristics around the carcass. During necropsy, liver samples were collected and subsequently tested for the presence of anticoagulant
rodenticide residues; liquid chromatography-tandem mass spectrometry was used to screen for presence of anticoagulant rodenticides and high-performance liquid chromatography was used to quantify positive samples. When predation was determined to be the cause of death, all lesions attributed to predation were described in detail. To distinguish between ante and post-mortem wounds (i.e., between predation and scavenging), we noted whether the lesions had associated hemorrhage and edema. In 14 cases, too few remains were present to identify hemorrhaging at wound sites, so only molecular analyses were conducted in these cases. Age-class at time of death was estimated as adult (≥24 months), subadult (12-23 months), and juvenile (<12 months) based either on tooth wear or cementum annuli counts.

**Molecular Analyses**—Forensic samples were processed and analyzed for predator (either felid or canid) DNA following the methods of Wengert et al. (2013). Because multiple polymerase chain reaction (PCR) products were occasionally obtained when the products were visualized on an agarose gel, we gel-excised the appropriately sized fragment (200–300bp for felids and 400 for canids) and extracted DNA using Qiagen Qiaquick Gel Extraction kit according to the manufacturer’s instructions. The PCR products were sequenced, then aligned using RidomTraceEdit (Ridom GmbH, Würzburg, Germany). Sequences were cross-referenced on GenBank using Basic Local Alignment Search Tool (BLAST) to match them to the most closely aligned sequence to identify species of predator DNA.

**Population Growth Rates**

*Background.*—Individuals will respond to changes in habitat, food availability, and weather conditions and these factors may cause fluctuations in abundance or density. Fishers are no exception to this general pattern (Jensen et al. 2012). As a rare species, however, the effects of these factors on population stability and viability are a significant conservation concern (Spencer et al. 2011, Reed and McCoy 2014).

Information on the growth trajectory for the fisher population in the SNAMP Fisher study area is uncertain, but there are several competing hypotheses regarding population status in the broader region encompassing the SNAMP Fisher study area. Evidence indicates that fishers in the Sierra Nevada experienced a range contraction of 30-50% over the last 75-100 years (Zielinski et al. 2005, 2013, Spencer et al. 2015), and research conducted between 2002 and 2012 provided no evidence of for an increasing population (Zielinski et al. 2013). Therefore this “no increase” hypothesis suggests
that despite protection from fur trapping and the development of policies to better sustain sensitive birds and mammals (North et al. 2009), fishers in the southern Sierra Nevada are not recovering. An alternative hypothesis is that fishers were very uncommon in our study area prior to 1990, and the current population resulted from a northward expansion from south of the Kings River (Tucker et al. 2014), equivalent to an approx. 30% increase in distribution in the overall southern Sierra Nevada region based on analyses of fisher habitat by Spencer et al. (2014). This alternative view is based primarily on genetic evidence of population subdivision (Tucker et al. 2012, 2014), and potentially supported by increasing fisher detections from track plate and camera monitoring after the mid-1990s (Zielinski et al. 1995, 2005, 2013). Research into the ecological relationships of fishers in the southern Sierra Nevada has increased dramatically since the mid-1990s, and although much insight on fisher ecology has been gained with regards to home range size, population density, and habitat use for resting and other activities (Jordan et al. 2011, Thompson et al. 2012, Purcell et al. 2009), no prior study has reported on the growth rate of any fisher population in this area.

**Population growth rates and Leslie-matrix modeling.**—Intensive investigation as part of the SNAMP Fisher study has generated information on all key vital rates needed to evaluate the population growth rate (λ) in the area, critical for understanding whether the population has the potential to persist, or if it is in decline. We developed an age-structured matrix model to estimate a deterministic population growth rate (λ) for the SNAMP Fisher study population using observed data on denning, fecundity, and survival. We defined 2 “adult” age classes (24 months, ≥36 months) for developing and including estimates of fertility in the matrix model for the population. Fertilities (Fi) were calculated for adult-age female fishers as:

\[
F_i = b(i)P_i
\]

where fecundity, \(b(i)\), was the mean number of female kits weaned per reproductive female (sex ratio at birth assumed = 0.5), and \(P_i\) was the age-specific survival rate (Gotelli 2001). Fertility for juveniles (F1) and subadults (F2) was fixed at 0. Age-specific survival rates were estimated for radiocollared juvenile (P1: 6-11 months), subadult (P2: 12-23 months) and adult-age (P3: ≥24 months) female fishers in the study area using monthly encounter histories in Kaplan-Meier staggered entry model analyses (KM survival). Survival estimates were produced for combined data on numbers of radiocollared fishers in each age class during the six year study period (All-year), given a natural transition of surviving individuals into a subsequent age class. Insufficient data was available to generate accurate annual survival estimates, so data was combined into 2-year increments (e.g., 2008-
Data on numbers of fishers in each age class for each 2-year increment were combined to generate survival estimates, starting with population years 2008-09 and 2009-10, and ending with population years 2012-13 and 2013-14.

Data from radiocollared animals from the study area indicated that female fishers commonly die by 6-8 years of age. We therefore included 8 age classes in our Leslie Matrix (A) formulation, where the numbers of fishers in each age class \( n_1 \) to \( n_8 \) at time \( t+1 \) = \( A \times n \) vector at \( t_0 \) according to equation 2:

\[
\begin{bmatrix}
  n_1 (t + 1) \\
  n_2 (t + 1) \\
  n_3 (t + 1) \\
  n_4 (t + 1) \\
  n_5 (t + 1) \\
  n_6 (t + 1) \\
  n_7 (t + 1) \\
  n_8 (t + 1)
\end{bmatrix}
= \begin{bmatrix}
  F1 & F2 & F3 & F4 & F4 & F4 & F4 & F4 \\
  P1 & 0 & 0 & 0 & 0 & 0 & 0 & 0 \\
  0 & P2 & 0 & 0 & 0 & 0 & 0 & 0 \\
  0 & 0 & P3 & 0 & 0 & 0 & 0 & 0 \\
  0 & 0 & 0 & P3 & 0 & 0 & 0 & 0 \\
  0 & 0 & 0 & 0 & P3 & 0 & 0 & 0 \\
  0 & 0 & 0 & 0 & 0 & P3 & 0 & 0 \\
  0 & 0 & 0 & 0 & 0 & 0 & P3 & 0
\end{bmatrix}
\begin{bmatrix}
  n_1(t_0) \\
  n_2(t_0) \\
  n_3(t_0) \\
  n_4(t_0) \\
  n_5(t_0) \\
  n_6(t_0) \\
  n_7(t_0) \\
  n_8(t_0)
\end{bmatrix}
\]

Equation 2

Estimates for adult fertility were determined from data on weaning rates, fecundity, and survival for young adult (F3; 24 months), and mature adult fishers (F4 ≥36 months) separately.

Numbers of fishers in age classes \( n_2 \), \( n_3 \), and \( n_4 \) for the \( n \) vector at time \( t_0 \) were based on the number of radiocollared female fishers present at the start of population year 3 on April 1, 2009 (\( n_2 = 5 \), \( n_3 = 6 \), \( n_4 = 5 \), \( n_5 = 5 \)), whereas \( n_1 \) was the number of juvenile females in the radiocollared population on Dec 31, 2009 (4 animals). We multiplied the Leslie matrix by the new vector of abundances for \( N_{t+1} \) for 20 successive years, and summed the number of individuals in each age class each year to obtain a total \( N \), and the population growth rate (\( \lambda \)) for year \( t+1 \) was calculated as \( N_{t+1}/N_t \). After several years a stable age distribution was achieved and \( \lambda \) converged to a constant value, which was the estimate of \( \lambda \) for the set of demographic parameters evaluated. We calculated a lower and upper range for \( \lambda \) based on the 95% lower and 95% upper C.I.s for age-specific survival and age-specific fertility. Finally, due to uncertainty in estimates for several demographic parameters related to methodology [small body size prevents radiotelemetry-based monitoring of survival for juveniles until 6 months of age (Facka et al. 2013); teat measures used to identify weaning for a small subset of adult females were less than 100% accurate in assigning reproductive status (Matthews et al. 2013a), estimates of litter size may be biased low if the movements of kits between dens was missed by cameras], we evaluated the sensitivity of the matrix model to 20% reductions in rates of survival and fertility for each age
class. Fertility is linked to age-specific survival according to Equation 1, and changes to fertility associated with reductions in survival were carried into the model when evaluating sensitivities.

**Population Size and Density**

**Background**

Information on population size and demographic parameters are fundamental for managing wildlife populations, especially when declines in abundance or range size have occurred and the species is the focus of conservation management. As previously noted, the overall southern Sierra Nevada fisher population is small (<350 adult fishers; Spencer et al. 2015), but appears to be stable over about the past decade (Zielinski et al. 2013). Research focused on the ecology and habitat use of fishers in the southern Sierra Nevada has been ongoing since the mid-1990s (Jordan 2007, Mazzoni 2002, Truex et al. 1998), but primarily for the area encompassed by the Kings River Fisher Project in the High Sierra District, Sierra National Forest. For that area, Jordan et al. (2011) used a capture-mark-resight/recapture design (CMR) to estimate a density of 0.063-0.109 fishers/km² in 2002-2004, whereas Thompson et al. (2012) used scat detector dogs and genetic detections in a spatially explicit CMR framework to estimate a fisher density of 0.065-0.28 fishers/km². Information on density is needed for other areas of the southern Sierra Nevada as well, because as the area of suitable habitat available to fishers in the southern Sierra Nevada is refined by improved modeling (Spencer et al. 2015), density values can be used to develop more accurate estimates of fisher abundance for conservation planning. Here, we estimated fisher population size and density in the middle four years of the study using mark-resight techniques (McClintock and White 2009) from camera surveys and live trapping.

**General Methods for Camera Surveys and Cameras**

Motion sensing cameras (Silent Image Professional, Rapidfire PC85; RECONYX Inc., Holmen, WI) were systematically deployed near the center of 1–km² grid cells in the study area beginning at the start of each of five “fall-winter” camera survey years (October 15-October 14 the following year). Placement of cameras within 1-km² grid cell cells was determined based on the presence of habitat elements important for fishers (e.g., presence of mature or large diameter trees, moderate to steep slopes, canopy cover ≥60%, proximity to permanent streams; Purcell et al. 2009, Zielinski et al. 2004). Cameras were focused on bait trees upon which we attached baits and applied scent lures as attractants. Baits were small pieces of venison (140-250 grams) in a dark colored sock (reduced consumption by insects), and 8-10 hard-shell pecans strung onto a wire (initial purpose was to
index squirrel abundance, but were also consumed by fishers). Scent lures were Hawbaker’s Fisher Scent Lure (Fort Loudon, PA) dabbed on the bait sock, Caven’s “Gusto” scent lure (Minnesota Trapline Products, Pennock, MN) applied near the base of the bait trees and on several nearby trees, and ~4 grams of peanut butter smeared on the nut ring (Popescu et al. 2014). Camera survey stations were typically visited (checked) every 8-10 days over 32-40 days to refresh scent lures and bait, and to maintain camera units, but the protocol varied depending on whether the survey station was within the Key Watershed part of the study area, or outside that area. Survey cameras within the Key Watersheds were left in place a minimum of 32 days (four 8-10 day checks), whereas cameras outside this area were deployed for a minimum two 8-10 day checks but removed on check two or three if fishers had been detected. We removed survey cameras after four checks unless the unit had been disturbed (most frequently by black bears, *Ursus americanus*) to where the bait tree was out of view or if the unit had been inoperative due to expended batteries or malfunction for more than five days during a check period. In those cases the survey was extended by one or more 8-10 day periods to assure adequate survey effort (Slauson et al. 2009).

**Camera surveys, live trapping, and radiocollar data.**—Camera surveys were done during all months of each camera survey year, but the time frame of interest for this part of the study was October 15 to March 15, related to assumptions for mark-resight analyses of a closed population scenario. There are 145 1-km² grid cells within or overlapping the Key Watersheds boundary; 128 of them are at least 50% USDA Forest Service ownership, and were surveyed in all four survey years. A total 319 1-km² grid cells external to the Key Watersheds and within the study area boundary (Fig. D3) were surveyed in at least one fall-winter camera survey year, and 221 (69%) of those were surveyed in two or more years.

Full details on live-trapping to radiocollar and mark individual fishers were provided above. However, for the purposes of mark-resight analyses, data on captures and recaptures of known fishers were included in the mark-resight dataset. Also, fishers sometimes shed their radiocollars, or collars separated at the breakaways as designed. Dropped radiocollars were retrieved from the field, and the locations of shed radiocollars were included in the resight dataset.

**Monitoring and home ranges.**—Radio collared fishers were monitored for activity status and relocated 4-6 days/week throughout the year by fixed-wing airplane. Standard methods were used to
obtain locations from the airplane as previously detailed, and mean error associated with aerial telemetry locations was estimated at 339 m.

Location records were used to develop home range models for individual fishers using the fixed kernel density method in Home Range Tools for ArcGIS 9.3 (Rodgers et al. 2007). Ninety-five percent fixed kernel home ranges were produced for individual animals for four fall-winter (October 15 to March 16) periods from 2008 to 2012 when ≥ 25 locations were available for an individual fisher. Home range area estimates from fixed kernel utilization distributions are sensitive to the choice of bandwidth as a smoothing parameter (Gitzen and Millspaugh 2003). We used the Ad Hoc method to identify the most appropriate reference bandwidth for smoothing fisher home ranges and minimizing formation of multiple polygons (Kie et al. 2010, Kie 2013). This procedure starts with identifying a reference bandwidth (h_ref), then reducing h_ref in 10% increments. A fixed kernel estimate is plotted for each increment, then visually inspected to determine at what point the home range estimate begins to fracture into multiple polygons. The bandwidth value (h) immediately prior to fracturing is selected as the most appropriate choice for that individual, and the procedure is repeated for each individual (Berger and Gese 2007, Kie 2013).

Resighting and Mark-resight Analyses.—Radiocollared fishers detected by cameras were identified by the pattern of infrared reflective tape bands on the antennae (Popescu et al. 2014). Detections of known fishers were counted once per camera station per calendar day. We were not able to unambiguously identify all radiocollared fishers detected at cameras due to occasional loss of bands and breakage of antennas; these detections were scored as collared unknown. Non-collared animals were counted as unmarked seen.

We considered the population as approximating closure during Oct 15 to Mar 16 because (1) most mortalities in the study site occurred between mid-March and September, (2) natal dispersal by juvenile-age fishers in the population was focused during March to August, and (3) fisher reproduction in California begins the third week in March (Matthews et al. 2013a, this study). Data on individual fisher resightings at camera stations or live traps were scored based on presence within 1-km² grid cells. Individual animal

Illustration D8: Example of infrared tape on collar antenna used to identify individual fishers for mark-resight analyses.
detection histories were developed identifying whether fishers were available for resighting based on
the presence of survey cameras or live traps within the boundaries of their 95% fixed kernel fall-winter
home ranges. Data were also compiled on the numbers of survey cameras and live traps deployed,
survey camera nights, and live trap nights for the fall-winter resight period.

The resighting data were analyzed using robust design mark-resight, log-normal Poisson
models (McClintock and White 2009). The mark-resight robust design is analogous to the mark-
recapture robust design of Kendall et al. (1995) and Kendall et al. (1997), in that it allows for
individual covariates in modeling resighting probabilities, and an open population between primary
sampling occasions. Along with data on marked animals, mark-resight models incorporate sightings of
unmarked animals, while the robust design allows for estimating abundance \( (N) \), apparent survival
between primary intervals \( (\phi) \), mean \( (\alpha) \) and overall resighting probabilities \( (\lambda) \), random individual
heterogeneity \( (\sigma^2) \), and transition probabilities between observable and unobservable states \( (\gamma'' \text{ and } \gamma') \)
(McClintock and White 2009). The parameter of interest, abundance \( (N) \), is a derived parameter, as
Poisson log-normal models estimate the number of unmarked individuals in the population, \( U \)
(McClintock and White 2009).

The Poisson log-normal mark-resight model takes the following form:
\[
[\alpha(.) \sigma(.) U(.) \phi(.) \gamma''(.) \gamma'(.)]
\]
in which \( \phi \) and \( \gamma'' \) (and \( \gamma' \)) were modeled using a sin link, while \( \alpha, \sigma, \) and \( U \) were modeled using a log
link.

The model assumptions are: (1) geographic closure, (2) population closure within primary
intervals, (3) no loss of marks, (4) no error in identifying marked and unmarked animals, (5) equal
resighting probability for both marked and unmarked individuals, and (6) sampling is with replacement
within secondary periods (McClintock and White 2009). We used camera survey years as the primary
sampling intervals and the number of resights and live trap recaptures for marked fishers within each
primary occasion as the resighting histories. Along with capture histories, robust-design Poisson log-
normal models require three other quantities: (1) marked superpopulation, the number of marked
individuals known to be in the population during primary interval \( j \), (2) number of times marked
individuals were sighted, but individual marks could not be identified, and (3) total unmarked
individual sightings during primary interval \( j \).
Because camera and live trapping was unbalanced across the study region among years, we added a grouping variable for subregion, with three subregions defined by the spatial segregation of camera efforts (Fig. D5). Each fisher was assigned to a particular mark-resight subregion based on the position of its 60% fall-winter home range isopleth. In addition, we included area and time (primary sampling interval) covariates,cams (camera effort for each subregion during each primary sampling interval in hours), and live (number of days live trapping was conducted) to account for variation in resighting probabilities, individual covariates weight and sex to account for individual and sex-based resighting probabilities. In the model parameterization, state transition probabilities remained constant \([\gamma(.) \text{ and } \gamma''(.)]\), apparent survival was modeled as function of region \([\phi(area)]\), and different combinations (additive and interactions) of the individual and time and region-based covariates were allowed.

We considered 19 candidate models and used AICc [Akaike Information Criterion adjusted for small sample size; (Burnham and Anderson 2002)] to rank models. We used model averaging for the top ranked models with a cumulative Akaike weight >0.95 to compute parameters and unconditional variances. The Area grouping parameter allowed for estimating population size.

**Figure D5:** Map illustrating the outlines of subregions used to organize camera and live trap data for mark-resight analyses. The study area is within the Bass Lake Ranger District, Sierra National Forest, California, but also included a relatively small portion Yosemite National Park.
and density for each subregion separately. We conducted analyses in program RMark v2.1.7 (Laake 2013) for R 3.0.2 (R_Core_Team 2013), which is an interface for program MARK (White and Burnham 1999). Lastly, the subregion and year-specific abundances were converted to densities by dividing population estimates by the area sampled by cameras and traps for each subregion and year. Areas sampled were estimated from subregion- and year-specific polygons created in ArcGIS 10.2 that encompassed the centroids of all 1-km² grid cells with a survey camera or a live trap with a fisher capture during October 15 to March 16. We then plotted the fall-winter home ranges with the sampling polygons and, based on visual assessment of spatial intersection of the 95% home range isopleths, applied a 1300 m buffer for each polygon. The width of the buffer for the polygons was the radius of the mean 95% fixed kernel fall-winter home range for subadult and adult female fishers in the population (mean = 20.8 km² ± SE 0.89, n = 70; Jordan 2007), which encompassed most areas used by radiocollared fishers resident in each subregion and excluded areas below or above the typical elevation range of fisher camera detections in the study area.

**Dispersal Movements**

**Background**

By simply moving from one habitat patch to another, dispersal of individuals has consequences not only for fitness, but also for population dynamics, population genetics, and species distribution at the landscape scale (Chepko-Sade and Halpin 1987, Lambin 1994, Clobert et al. 2001). For these reasons, processes that foment dispersal behavior have been the focus of research interest in relation to inbreeding avoidance, intraspecific competition for mates and resources (Estes-Zumpf and Rachlow 2009, Wolff et al. 1988), and costs and benefits of dispersal, particularly in relation to gender (Pusey 1987).

Natal dispersal, permanent movement from the natal area to the location where individuals reproduce or would have reproduced depending on survival (Howard 1960), is the most common type of dispersal. Gender bias in which one sex, typically males, disperses more frequently or farther than the other, has been documented in many mammals (Greenwood 1980; Pusey 1987, Sweitzer and Berger 1998). Proximate mechanisms triggering natal dispersal and potentially influencing dispersal distance include population density (Gaines and McClanaghan 1980), habitat quality (Lidicker 1975), and body condition (Dufty and Belthoff 2001, Nunes and Holekamp 1996). Information on dispersal provides insights on how far, and over what sorts of terrain, individuals may move and therefore how
populations may be demographically and genetically interconnected or isolated. Barriers or impediments to dispersal reduce gene flow and may prevent populations from colonizing or recolonizing suitable habitat areas.

Dispersal behavior by fishers is of high management interest in California where the species currently occupies less than half of its historical range as described in the early 1900s (Grinnell et al. 1937). In the southern Sierra Nevada conservation planning area, fishers occupy approx. 4,400 km² of mid-elevation, mixed-coniferous forest between the Merced River in Yosemite National Park in the north to the Greenhorn Mountains in the Sequoia National Forest in the south. The southern Sierra Nevada population does not appear to be expanding geographically (Zielinski et al. 2013), despite changes in management promoting redevelopment of suitable fisher habitat in the Sierra Nevada (North et al. 2009). Dispersal movements by fishers are potentially inhibited by exposure to multiple restrictive habitat and landscape features (Spencer et al. 2015, Tucker 2013). Moreover, Matthews et al. (2013a) suggested that because of their relatively limited vagility, conservation-directed management to promote fisher recovery in formerly occupied portions of their range in California may require translocations, unless population growth rates significantly exceed 1.0 in the future.

We used information on juvenile home ranges, likely maternal home ranges (determined by genetic analyses), and adult home ranges to evaluate patterns in natal dispersal for fishers in the SNAMP Fisher study area. We hypothesized that (1) a greater proportion of the juvenile male population would disperse than the juvenile female population, (2) dispersal distances for males would be longer than for females, and (3) long distance movements would be more frequent for males compared to females. In addition to estimating Euclidean distance between juvenile or maternal home ranges and adult home ranges, we also used a least-cost corridor analyses with an expert opinion-based cost surface to estimate both short and longer distance movement paths associated with natal dispersal. Both methods likely underestimate the actual dispersal distance travelled, because we are unable to plot that actual path followed and must presume based on what we know about the species. At the same time, both methods present critical information for management and conservation planning.

Assessing dispersal using home range models

Location records were used to develop home range models for individual fishers by using the fixed-kernel density method in Home Range Tools for ArcGIS 9.3 (Rodgers et al. 2007; ESRI,
We developed 95% fixed-kernel home range models for juvenile, subadult, and adult-age fishers when ≥ 25 locations were available for the pre-dispersal or post-dispersal period of interest. Approximate center positions (centroids) were estimated for each home range using the XTools extension in ArcGIS (Data East LLC, Novolsibirsk, Russia). Because both area estimates and shapes of fixed kernel home ranges are sensitive to the choice of bandwidth as a smoothing parameter (Gitzen and Millspaugh 2003), we used the Ad Hoc method to identify the most appropriate reference bandwidth for smoothing fisher home ranges and minimizing formation of multiple polygons (Kie 2013). Finally, in some cases radiocollars were shed by juveniles within a few weeks of initial capture, before ≥25 locations records had been acquired. In these cases we used centroids from 100% Minimum Convex Polygons for natal area centroids (Aubry and Raley 2006).

Dispersal distance by Euclidean geometry

Minimum distances moved between natal or maternal home ranges, and subadult or adult home ranges were estimated as the Euclidean distance between each pair of centroids. For juvenile fishers without maternity assignments, we used fall and winter location records to identify a centroid for natal areas, but excluded locations that were associated with initiation of dispersal. Fall and winter location records for juvenile fishers were visually screened in ArcGIS to identify calendar dates associated with initiation of the exploratory, or transitional, period of the dispersal process (Vangen et al. 2001). Location datasets used to develop home ranges for juvenile fishers (natal area home ranges) were truncated by date to exclude transitional movements. Transitional movements were not apparent for all juvenile fishers, however, and in these cases we used the pool of location records from capture to approx. March 31 for the natal area home range.

Microsatellite genetic analyses for identifying maternity

We used microsatellite genotypes to assign maternity for juvenile and subadult fishers, which allowed for estimating natal dispersal from the centroids of denning season home ranges for their mothers. Whole genomic DNA was extracted from fisher tissues and hair using the QIAGEN Dneasy Tissue Kit (Qiagen, Valencia, CA, USA) according to manufacturer's instructions. We analyzed 111 samples at the following thirteen microsatellite loci: Ma1, MP059, MP144, MP175, MP197, MP200, MP247, Ggu101, Ggu216, Lut604, Lut733, Mer022 and Mvis002 (Davis and Strobeck 1998; Flemming et al. 1999, Dallas and Piertney 1998; Jordan et al. 2007). These loci were previously found to be variable in fishers in the Southern Sierra (Jordan et al. 2007; Tucker et al. 2014).
Maternity of kits was evaluated using two approaches; the first was by evaluating allele sharing. However fishers in the southern Sierra Nevada were previously known to be genetically limited (Wisely et al. 2004, Knaus et al. 2011), and we also used insight from field associations (capture positions, home range patterns, denning season data to identify small subsets of 3-6 adult females considered possible mothers for each juvenile/subadult. These subsets of possible mothers were further narrowed to a smaller “candidate set” by excluding those that did not share alleles with the juveniles. We applied the maximum likelihood approach in program CERVUS v3.03 (www.fieldgenetics.com) to evaluate the candidate set of females for maternity assignment. CERVUS is a Windows-based software package for inferring parentage in natural populations wherein laboratory typing error is considered along with data on population allelic frequencies, the number of candidate mothers, and the proportion of potential mothers sampled in Monte Carlo simulations, which produce confidence levels for the candidate set of putative parents (Slate et al. 2000). The confidence measure of CERVUS is based on delta, which is the difference between the likelihood ratio for the most likely candidate and the second most likely candidate (Marshall et al. 1998). We assigned maternal-offspring pairs based on likelihood ratio LOD scores (natural log of the likelihood ratio) using both strict (99%), and relaxed (95%) confidence. In the last step we considered the maternity assignments with field data to verify, or select the next most likely female from the LOD scores based on the known biological status of putative mothers (reproductive or non-reproductive, age as juvenile, subadult or adult in the season juveniles were born). In several cases, the overall analysis was unable to link juveniles to a known, radiocollared female in the population. Developers of CERVUS previously determined that the analytical procedure correctly assigned parentage for ~92% of known fathers in red deer (Cervus elaphus) (Slate et al. 2000). In our study we evaluated the performance of CERVUS to correctly identify mothers using five known mother-offspring pairs.

Dispersal distances for juvenile or subadult-age fishers with maternity assignments were estimated as the Euclidean distance between the geometric centroid of the denning season home range of the mother, and the geometric centroid for the home range where the juvenile settled. In some cases the mother had not been monitored during the denning season when a juvenile was produced. In these cases we used the centroid for the female’s “annual” home range. Annual home ranges were calculated when we had at least 75 location records, with a minimum of five locations in at least three of the four seasons of the year. Seasons were spring (Mar 21 to Jun 20), summer (Jun 21 to Sep 20), fall (Sep 21 to Dec 20), and winter (Dec 21 to Mar 20).
Least-cost paths for natal dispersal

Dispersal is most often reported as the Euclidean, or straight-line distance between the natal area and the subadult or adult home range (Matthews et al. 2013). Fishers in the southern Sierra Nevada inhabit mountainous areas within a limited elevation range and with a mix of forested areas with mild topography, and ridges and deep river canyons with extreme topographic relief. In these types of landscape and habitat conditions opportunities for straight-line movement traversing multiple kilometers will be constrained.

Least-cost modeling is an approach for assessing potential animal routes across the landscape based on the assumed cost of movement between locations or termini (Beir et al. 2008). Least-cost models have previously been used to predict dispersal paths for mammals from empirical data (Driezen et al. 2007), and we took a similar approach in this study. Connectivity analysis was performed between centroids of natal and established juvenile home ranges for 24 female and 20 male fishers with Linkage Mapper (McRae and Kavanagh 2011). Linkage Mapper uses a resistance to movement (cost) surface layer to delineate least cost paths between focal point pairs. A cost surface layer was developed that assigned a resistance score (inverse permeability value) representing the cost to fishers of moving through each land cover type, including potential risk and aversive responses to roads and steep topography in river canyons (Fig. D6).

Expert opinion resistance scores were modified from those developed previously for Sierra Nevada fisher least-cost corridor models (Spencer and Rustigian-Romsos 2012) by (1) simplifying the land cover divisions, (2) expanding the overall cost range, and (3) incorporating recent published and unpublished data on fisher ecology summarized in a conservation assessment developed for fishers in the southern Sierra Nevada by a group of 13 research scientists (Spencer et al. 2015). We used the Polynomial Approximation with Exponential Kernel (PAEK) algorithm in ArcMap 9.3.1 (ESRI 2009) to smooth the movement paths for purpose of display. Length of least cost paths between juvenile or maternal home range centers and subadult or adult home range centers were calculated in ArcGIS 10.2. Basic metrics on least cost paths (means, range, standard error of the mean) were produced and summarized for comparison with mean dispersal distances from Euclidean geometry.

Analysis

Mean dispersal distances were compared between female and male fishers using two-sample $t$-
Figure D6: Illustration of the Expert opinion cost surface used to develop Least Cost movement path for dispersing fishers. The map encompasses the portion of the SNAMP Fisher Study Area including the Key Watersheds and the area to the southeast including Chilkoot Lake and Mammoth Pool Reservoir. Note: Chilkoot Lake is at the northwestern edge of the Chiquito Ridge, a high elevation region including Little Shuteye Peak, Shuteye Peak, and Shuteye Pass; notice the narrowness of restrictive habitat at the Shuteye Pass topographic feature.
tests. We also assessed potential male/female differences in dispersal behavior using Pearson $\chi^2$ or log-linear model analyses. We used dispersal distances to classify each fisher as being either philopatric (dispersal distance $\leq 5.4$ km) or a disperser (dispersal distance $> 5.4$ km), where 5.4 km was the diameter of average 95% fixed kernel home range of adult females fishers in the study population (22.93 km$^2$, $n = 56$; Table D25). We also assessed the overall pattern in dispersal behavior by assessing the proportion of each sex that was very philopatric (dispersal distance $< 2.7$ km; one-half the diameter of the mean 95% fixed-kernel home range for adult female fishers), short distance philopatric ($2.7$ km $\leq$ dispersal distance $< 5.4$ km), a mid-distance disperser ($5.4$ km $\leq$ dispersal distance $< 10.8$ km), or a long distance disperser (dispersal distance greater than $10.8$ km; 2x the diameter of the average adult female home range).

**Home Range Dynamics Methods**

**Background**

Among terrestrial vertebrates, mammalian carnivores have the largest home ranges for their body size of any organism. The fisher, like other mammalian carnivores, occupies a relatively large amount of space for its body mass, with average annual home range areas of 38 km$^2$ for adult males and 15 km$^2$ for adult females across North America (Powell 1994). Studies of the two remnant populations in California have produced home range area estimates generally consistent with North American averages: 22 to 58 km$^2$ for adult males and 5 to 15 km$^2$ for adult females (Boroski et al. 2002, Zielinski et al. 2004). Zielinski et al. (2004) also reported intraspecific variation in home range size between adult females of the northern coastal and southern Sierra Nevada populations.

Intraspecific variation in home range size has been linked to ecological factors such as population density, prey availability, body mass, and latitude (Buskirk and McDonald 1989, Gompper and Gittleman 1991), and to methodological factors such as sampling interval and duration (Buskirk and McDonald 1989, Swihart and Slade 1985). Our review of the literature suggests that little attention has been paid to potential relationships between home range size and field techniques used to obtain animal locations. Further, the choice of an appropriate bandwidth, or smoothing parameter when creating utilization distributions is a critical step during kernel-based home range estimation in need of standardization (Kie et al. 2013).
We present and discuss home range dynamics for fisher in the Sierra National Forest, while also describing seasonal variation in home range movements for female and male fishers. We hypothesized that aerial telemetry would be more likely than ground telemetry to detect animals outside of their core use areas and during dispersal events and sallies, and would therefore produce larger home range estimates. Our primary objective was to compare our fisher home range sizes with those generated by other studies in the southern Sierra Nevada, where ecological factors would be held relatively constant. Additionally, we wished to characterize variation in space use between sexes, among age classes, and across seasons for our study population.

Locations and analyses

Fisher locations from live-trap captures, dropped or shed radiocollars, carcasses, dens and rest trees, camera detections, a small number of GPS radiocollars (Telemetry Solutions, Livermore, CA) and position estimates from fixed-wing aerial radiotelemetry (Table D1) were used to delineate home range

<table>
<thead>
<tr>
<th>Location type</th>
<th>No. of locations</th>
<th>UTM accuracy</th>
<th>Description of methods and details</th>
</tr>
</thead>
<tbody>
<tr>
<td>Aerial telemetry(^a)</td>
<td>31,367</td>
<td>± 339 m</td>
<td>Standard methods by fixed-wing airplane (Sweitzer 2013)</td>
</tr>
<tr>
<td>Camera detections</td>
<td>2,454</td>
<td>± 10 m</td>
<td>Position of camera; individuals fishers identified by infrared tape on radiocollar antennas (Popescu et al. 2014)</td>
</tr>
<tr>
<td>GPS radiocollar(^b)</td>
<td>633</td>
<td>± 15 m</td>
<td>Used on limited number of animals (N = 8) in 2009 and 2010</td>
</tr>
<tr>
<td>Den or rest tree</td>
<td>526</td>
<td>± 10 m</td>
<td>Homing to trees by ground radiotelemetry during spring denning seasons; did not identify rest trees in other seasons</td>
</tr>
<tr>
<td>Live trap capture</td>
<td>277</td>
<td>± 10 m</td>
<td>Trap positions for known ID captures; most live-trapping was in October to March</td>
</tr>
<tr>
<td>Shed radiocollars, fisher carcass</td>
<td>97</td>
<td>± 10 m</td>
<td>Homing to inactive signals by ground radiotelemetry</td>
</tr>
</tbody>
</table>

\(^a\) Accuracy determined as the mean Euclidean distance between aerial telemetry location and fixed position of test collars (n = 501) on the ground. Test locations also included locations to dropped radiocollars, carcass locations, and fishers in live traps (Technicians were "blind" to locations of test collars, or other test locations).

\(^b\) Used for limited duration and primarily on male fishers (596 locations for 6 different males; 37 locations for 2 females). SNAMP Fisher ceased using GPS collars due to poor reliability and bias in fix rates; fix rates were high when GPS collars were left in open areas with mild topography, and low when GPS collars were placed at locations with dense overhead canopy and steep topography.

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ranges. Accuracy of locations obtained by homing to den tree or rest tree locations by ground telemetry, camera detections, capture positions, and carcass and dropped collar positions were generally ± 15 m from a handheld global positioning system device. We addressed and minimized autocorrelation by discarding locations in excess of two per animal per day, or less than 8 hours apart per individual. Location records were used to develop home range models for individual fishers using the fixed kernel density method in Home Range Tools for ArcGIS 9.3 (Rodgers et al. 2007). Ninety-five percent fixed kernel home ranges were produced for individual animals for four seasons, and for “annual” population years. Season-specific home range models were produced when ≥25 locations were available for a fisher. Annual home range models were developed when we had location records in fall, winter, and least one other season, and sample size was ≥75 for all annual home range models. Kernels were smoothed using the minimum proportion of reference bandwidth that produced a contiguous home range polygon (Kie 2013). We defined core use areas using the procedure described by Bingham and Noon (1997), calculating adaptive kernel isopleths at 10% intervals and then identifying at which isopleth actual use exceeds predicted use, assuming a uniform distribution of locations. While more defensible, this approach can yield different core use isopleths for different individuals. We tested for differences in home range areas between males and females, stratified by age and season, using two sample t-tests (P<0.05). Potential differences in home range size among seasons was assessed using repeated measures analysis of variance (ANOVA) (P<0.05).

**SNAMP Fisher Management Indicators**

**Background**

In 2008 there was great interest in new information developing from SNAMP Fisher that might be important for management and management planning. We therefore developed three Indicators for fisher management that would provide insight on the status of the study population of fishers in the Bass Lake District, Sierra National Forest. These management indicators were chosen based on information that could be summarized annually, and that linked to the likely responses of fishers to management and potential habitat change at the local (sub home range scale), home range, and population level (larger landscape scale relevant to District-level forest management; Table D2).

**Mgt. Indicator 1: Occupancy/Activity of fishers within Key Watersheds.**

Beginning October 2007 we implemented regular camera surveys of all 1-km² grid cells that are partly or entirely encompassed by the boundary of the SNAMP Fisher Key Watersheds (Fig. D3). Several
grid cells that were predominantly private lands (e.g., the Fish Camp area), or that were below the typical elevation range of fishers in the region (< 914 m) were not surveyed unless we had permission of access from the landowner. The annual resurvey of the Key Watersheds was a research priority in all years, and camera surveys were initiated at the start of each camera survey year (mid-October), continuing until most grid cells had been surveyed by late winter or early spring. High elevation areas (northeast portion of Key Watersheds) were generally surveyed first, due to more difficult access during mid and late winter. Deep snow conditions in most winters required use of snowmobiles or an all-terrain-vehicle modified with tracks to access high elevation grid cell centers. Results of the Key Watersheds camera surveys were summarized according to (1) fisher presence or absence, and (2) level of fisher activity based on the number of days fishers were detected in each grid cell.

Mgt. Indicator 2: Number of male and female resident fishers using Key Watersheds area. Juvenile fishers exhibit exploratory movements, and sometimes dispersed away from their natal areas where we first captured and monitored them. Dispersal by juvenile fishers often extends into summer when they are 13-15 months old and considered subadults. We considered subadult fishers (12 to 23 months old) to be “settled” after natal dispersal in late August/September. Ninety-five percent fixed kernel home range models were developed from location records during September 1 to March 15 for all radiocollared fishers (Sept-March home range). Analyses were completed in ArcGIS 9.3.1 to estimate the proportion of each Sept-March home range for subadult and adult fishers that were included or “intersected” within the boundary of the Key Watersheds. Management Indicator 2 was calculated as the sum of the proportions of individual subadult and adult Sept-March home ranges within the Key Watersheds focal study area. Management Indicator 2 was calculated for female and male fishers for each of six September to March 15 periods beginning September 2008 and ending March 2013.
Table D2: Overview of potential negative effects of fuel reduction treatments and other forest management activities on the biology and natural history of fishers, organized according to three scales in the SNAMP Fisher study area, Bass Lake District, Sierra National Forest.

<table>
<thead>
<tr>
<th>Scale of effect, Description</th>
<th>Likely response</th>
<th>Data requirements</th>
<th>Management Indicator</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>LOCAL:</strong> SPLATS may cause habitat patches to become less suitable for current use; foraging, refuge/escape cover</td>
<td>Use of treated areas declines or ceases</td>
<td>Before/After and ongoing use of areas altered by management activities</td>
<td>Use of 1-km² grid cells within Key Watersheds (or in other areas), estimated annually using camera surveys</td>
</tr>
<tr>
<td><strong>HOME RANGE:</strong> SPLATS may reduce availability of key resources such as den sites, rest sites, availability of prey</td>
<td>Individual fishers cease use of treated areas</td>
<td>Monitor individual fishers, acquire locations, develop home range models, track dispersal movements</td>
<td>Estimate of the number of individual fishers using the Key Watershed focal study area during population years</td>
</tr>
<tr>
<td><strong>POPULATION/REGION:</strong> Multiple projects implemented every few years may degrade suitable habitat for fishers; population source areas become sink areas</td>
<td>Survival and reproduction decline; population size and density decline over time</td>
<td>Information on survival and reproduction of individual fishers in the overall study area; estimate population growth rate, evaluate population viability in the Sierra National Forest</td>
<td>Survival and reproduction of fishers in the overall study area. Estimate population growth rate, evaluate population viability in the Sierra National Forest</td>
</tr>
</tbody>
</table>

Mgt. Indicator 3: Survival of adult-age female fishers in the SNAMP Fisher Study Area

Survival, and survival of adult females in particular, is an important demographic parameter necessary for understanding the population growth trajectory for most vertebrate wildlife species (Murdoch et al. 2010). All radiocollared fishers were monitored 4-6 days/week by fixed-wing aerial telemetry to assess live/dead status. Information on survival status for radiocollared fishers was organized by month of each population year (Apr 1 to March 31), and analyzed using the Kaplan-Meier (KM) staggered entry method (Koen et al. 2007, Pollock et al. 1989, Price et al. 2010). KM models were used to produce estimates of annual survival and combined year survival for the study population. Annual survival can be moderately to highly variable, thereby suggesting a negative population growth trajectory that may not be appropriate for a long-lived species with a generation time of two or more years. We therefore combined monthly data on survival status for individual fishers for five 2-year periods (population years 2 and 3, population years 3 and 4, population years 4
and 5, population years 5 and 6, population years 6 and 7) beginning April 2008 and ending March 2014. We further extended the inference for this management indicator by estimating survival for juvenile and subadult females, and by compiling data on weaning reproductive rates and weaning litter sizes from data collected on fisher reproduction during April 2008 to June 2013. These data were used to estimate fertility rates, which, along with data on juvenile, subadult, and adult female survival, were used to estimate deterministic population growth rates using a four age class Leslie Matrix population model.

**Fisher response to fuel management**

**Occupancy modeling**

For the purpose of occupancy surveys, we deployed cameras in the 128 1-km² grid cells that were ≥50% public lands and within the 4 focal watersheds. We also deployed cameras in areas with recent histories of extractive or restorative fuel reduction, between 2002 and October 2008, or because forest management projects were planned to occur in the areas before December 2011. Most of these grid cells were repeat surveyed in 7 different camera survey years, as part of our initial plan of using a BACI framework for the occupancy analyses (Popescu et al., 2012). However because we were not aware of all planned or prior forest management activities within the study area when the project was initiated, some of the multi-season grid cells were added to the group that were repeat surveyed several years after the first camera survey year (2007-08).

The distribution of fishers in the southern Sierra Nevada, CA is constrained by elevation, and closely associated with mixed-conifer forest habitats with relatively large trees, and high canopy cover (Davis et al., 2007). We therefore developed local, patch-specific biophysical covariates for use in analytical models of occupancy. We calculated the mean elevation \((elev)\) for each surveyed grid cell, which was always included in occupancy analyses with its quadratic term \((elev^2)\). These covariates were standardized. Habitat covariates included the proportion forest (i.e., total tree) and hardwood cover \((denMD)\) based on land-cover data derived from satellite imagery (CWHR CalVeg; USDA Forest Service 2012). We did not include covariates representing average tree size and slope because of their collinearity with forest cover and elevation.

There were a diversity of forest management activities that occurred on the Sierra NF from 2002 (5 years before the start of our study) until the last camera survey year starting in October 2013 (period
Most of the management activities we used for covariates were developed from the USDA Forest Service FACTs database. FACTs (Forest Service Activity Tracking System; http://www.fs.usda.gov/main/r5/landmanagement/gis), is a tracking system including a geospatial database of forest management activities that occur on national forest service lands in California and elsewhere (FACTs User Guide 2013). Polygon layers included in the FACTS database are associated with attributes detailing management activity codes, and dates for when activities were initiated and completed. There are known uncertainties in FACTS with regards spatial precision, area of treatment polygons, and lack of details on whether a treatment activity was completed for an entire polygon (Garner, 2013). We also know that some entries represent perimeters encompassing smaller subunits treated at the same time as well as some areas unaffected by the management activity (Zielinski et al., 2013). Nevertheless, FACTS data constitute the best available and consistent record of the annual management activities that occurred on national forest lands in our study area.

Two recent studies used FACTS information to assess how fishers respond to disturbances from Forest Projects elsewhere in the southern Sierra Nevada (Garner 2013; Zielinski et al. 2013). We considered FACTS activities that were previously used in those studies, but also reviewed full descriptions of each management activity included in the FACTs User Guide (2013) when identifying a subset of 24 that were considered as potentially influencing local scale habitat use by fishers related to how each altered forest habitat structure or if they represented significant ground-disturbing activities (Zielinski et al. 2013; FACTs User Guide 2013). For example, we included forms of harvest (e.g., code 4152 Group Selection Cut) and vegetation management (e.g., code 4220 Commercial Thinning, code 4580 Mastication/Mowing) that would have direct effects on the basis of their disturbance and alteration of forest structure (Zielinski et al. 2013). We excluded activities that did not meet this criterion, and several that rarely occurred, or that silviculturist Dave Smith with the Sierra NF recommended against using (e.g., code 4290 Administrative Changes; code 4314 Pretreatment Exam for Reforestation; code 4530 Prune; code 4511 Tree Release and Weed; code 4552 Area Fertilizing; code 4980 Other Tree Improvement; code 4540 Control of Understory Vegetation).

There were 4 other activities or events that were not systematically tracked by the FACTS system; hazard tree removals (e.g., hazard tree logging), private timber harvests (THPs), and historical or recent wildfires. Hazard tree logging was the removal of medium and large trees (no DBH restriction) within 91 m of forest roads when they were considered likely to fall during storms, or if they were
decadent or diseased (SNFP 2004). Information on hazard tree logging in the Bass Lake Ranger District was available for 2009, 2010, and 2011, and we were provided with GIS shapefiles identifying road segments along which hazard tree logging occurred. Private timber harvest occasionally occurred on large parcels of private land within or adjacent to the Sierra NF in Madera County and Mariposa County. Harvesting of timber on private lands in California requires preparation of Timber Harvest Plans (THPs) that are reviewed and approved by state agency CAL FIRE, which was our source for geospatial data on private THP activities in Madera County and Mariposa County (ftp://ftp.fire.ca.gov/forest). Basic records on the estimated spatial extent of wildfires that occurred in the national forest portion of study area were maintained by the Sierra NF, and included polygon shapes and ignition dates of wildfires that occurred from 1911 to 2013. We also acquired geospatial data on natural ignition and management fires for Yosemite NP for 1930 to 2008, which was sufficient for our analyses because there were no camera surveys completed in southern Yosemite NP after May 2009. Attribute information included with the various geospatial data were used to assign activities and wildfires to individual camera survey years. For example, if a management activity was identified as completed before October 15, 2009, the disturbance was assigned to camera survey year 2008-09.

We used ArcGIS 10.2 (ESRI, Redlands, CA) to estimate the area of each 1-km² surveyed grid cell with hazard tree logging, private timber harvest, and wildfires, which were merged with the FACTs information for 2002 to 2013. After merging, we reviewed the entries, and removed polygons that were duplicated in several years (e.g., those with the same FACTS code with identical shapes and areas but with different years of completion). We also removed any duplicate wildfire records that were included in both the FACTs data and in the local Sierra NF wildfire database. We then used the detailed descriptions of each FACTs activity type to create 3 composite variables for use as covariates for occupancy analyses. Covariates for extractive fuel reduction (log.5) and restorative fuel reduction (hazfuel.5) included the cumulative areas of these activities in each grid cell in the 5 years immediately preceding each camera survey. For example, the hazfuel.5 covariate for any grid cells that were surveyed in camera survey year 2012-13, was calculated as the sum of the areas (m²) of all restorative fuel reduction activities that occurred in those grid cells during fiscal years 2007-08, 2008-09, 2009-10, 2010-11, and 2011-12, from which we calculated the proportion of the grid cell disturbed by the treatment. Because of the coordinated series of extractive and restorative fuel treatments associated with SPLATs, multiple different treatments could be applied on the same forest stand within a 5 year period (Zielinski et al., 2013). It was therefore possible that the cumulative area of a grid cell that was
treated during a 5-year period could exceed 1-km$^2$. In the few cases where this occurred (hazfuels.5 only), the proportion of the grid cell treated was truncated at 1.0 (100%).

The third composite variable that was related to fisher presence in model analyses was for managed burning and wildfires within each 1-km$^2$ grid cell. When we reviewed the FACTS and Sierra NF and Yosemite NP databases, it was apparent that managed burning was uncommon in the study area during 2002 to 2013. Although managed burns were commonly planned in the Sierra NF portion of the study area as part of SPLAT-based fuel reduction, many managed burns were cancelled and not rescheduled because weather conditions were not suitable, or because burning was prohibited by the San Joaquin Valley Air District (D. Martin, personal communication). Also, a late summer managed burn in Yosemite NP in 2009 escaped containment and burned 7,425 ha (Big Meadow Fire), which discouraged other managed burns in the region for several years thereafter. We therefore combined information on managed burning and the longer time-series of wildfires in the study area into a single composite variable representing managed burn+wildfires within 50 years of a survey (burn.1.50).

We used multi-season occupancy models to evaluate the importance of forest management covariates to explain the persistence of fishers at occupied grid cells and colonization of unoccupied grid cells (Zielinski et al., 2013). We defined colonization ($\gamma$) as the probability that a grid cell unoccupied in year $t$ would be occupied in year $t+1$, and modeled it as: logit($\gamma$) = $\beta_0 + \beta_1 x_1 + \beta_2 x_2 + \ldots$. We defined persistence as 1- extinction where extinction ($\phi$) was the probability that a grid cell occupied in year $t$ would be unoccupied in year $t+1$, and modeled it as: logit($\phi$) = $\beta_0 + \beta_1 x_1 + \beta_2 x_2 + \ldots$. The multi-season models also included a component for occupancy in the initial year a site was surveyed: logit($\psi_{\text{initial}}$) = $\beta_{\psi_0} + \beta_{\psi_1} x_1 + \beta_{\psi_2} x_2 + \ldots$.

We created a detection history of whether a fisher was observed by a camera within each grid cell during each consecutive survey period after set-up or re-baiting for up to 5 8-10 day periods during a survey year. This was repeated for up to 6 consecutive years (e.g., 00101 00000 01110 00010 01101 00000) for every grid cell. If surveys did not occur during any of the 5 periods and 6 seasons at any of the grid cells these data were treated as missing data. Models were solved by maximum likelihood estimation (MLE) via R statistical software (Version 3.0.1, www.r-project.org) using the unmarked package and the coexist function (Fiske and Chandler 2011). We followed an information-theoretic approach for evaluating the relative importance of different candidate models, and for assessing the
relative importance of individual covariates [sum of AIC weights ($AIC_{wi}$) for candidate models including each covariate; Burnham and Anderson 2002].

Covariates for potentially explaining detection probability included a categorical, first order Markov process reflecting whether a fisher was detected in the previous survey period in a season ($auto.y$; Hines et al., 2010; Slauson et al., 2012), the number of functional camera days in a survey period divided by 10 ($camdays$), $denMD$, and a categorical variable representing whether the survey was conducted in summer ($summer$) instead of in fall to spring.

Due to the smaller sample size of sites available for fitting multi-season models ($n = 361$), we only evaluated the role of forest management covariates ($log.5$, $hazfuels.5$, and $burn.1.50$) in explaining annual transitions in occupancy state (colonization and extinction). For the initial occupancy component of the multi-season models, we restricted potential explanatory covariates to $denMD$ and $elev + elev^2$. For multi-model evaluations of multi-season models, we first fit models including all 8 combinations of the forest management covariates on the colonization component and an intercept-only extinction component. We considered any covariate with a relative importance value $> 0.65$ to be predictive and important for colonization. Next, we fit models including all 8 combinations of the forest management covariates on the extinction component multiplied by all combinations of colonization covariates identified as important. We deemed any covariate in the extinction models with a relative importance value $> 0.65$ as predictive for explaining local extinction. Finally, we computed model-averaged parameter estimates for the colonization and extinction covariates identified as important. Model averaging was based on only those models summing to the top 0.95 of model weights.

**Integration**

Development of vegetation map

We refer the reader to Appendix C for more complete details because we used the same mapping procedure at Sugar Pine as was done at Last Chance. In summary, we developed a pre-treatment vegetation map using a combination of LiDAR, high-resolution digital color-infrared (CIR) aerial imagery, and an intensive network of field plots. First, we used LiDAR and CIR data to create an initial polygon-based map where the polygons represented areas of homogeneous vegetation in terms of species, vertical structure, basal area, and canopy cover. We collected the LiDAR and CIR data...
data before the SPLAT implementation, and we sampled vegetation at the field plots before and after treatment. We then used the field-plot data to impute detailed attributes (e.g., tree lists and fuels models) for each polygon. Thus, we derived two different maps (with and without treatment), which we used in fire and forest-growth modeling.

Modeling fire and forest dynamics

We again refer the reader to Appendix C for more complete details of the fire and forest-growth simulations because we followed the same general procedure at Sugar Pine as we did at Last Chance. We used FARSITE (Finney 1998) to simulate a likely wildfire scenario based on the weather conditions during the 2014 French Fire, which burned 13,837 ac (5,602 ha) 12.5 mi (20 km) southeast of the study area. We obtained weather information from the Batterson Remote Automatic Weather Station, limited to the active burning period of the French Fire (August-September 2014), which served as the basis of our fire modeling. Moisture content for live and dead woody fuels and live herbaceous fuels used in the model were equivalent to 97th percentile weather conditions. Our ignition location was established using fire-origin point data supplied by the Bass Lake Ranger District of the Sierra National Forest. Based on the mapped data, we identified an area with the highest ignition frequency, which was located on the west ridge of the Cedar Valley watershed. The simulation duration was set to allow the fire perimeter to expand through the entire study area.

For all four scenarios (treated/fire, untreated/fire, treated/no fire, untreated/no fire), we then simulated 30 years of forest growth on the study area in 10-year time steps using the Forest Vegetation Simulator (FVS; Dixon 2002) with the Fire and Fuels Extension (FFE; Reinhardt and Crookston 2003). The simulations were performed using the integrated platform ArcFuels (Ager et al. 2006, Vaillant et al. 2011), which runs FVS-FFE to produce the forest structure inputs needed for FARSITE.

Assessing the effects of fire and SPLATs on fisher habitat

We identified canopy cover and large trees as the most important elements of forest structure for fisher habitat because fisher den and resting locations in the southern Sierra Nevada were associated with high canopy cover and large trees (Zielinski et al. 2004, Purcell et al. 2009, Thompson et al. 2011). We defined fisher habitat as forest stands where the canopy cover was ≥60% and the density of large trees (≥24 in [61.0 cm] dbh) was ≥15.4 trees/ac (38 trees/ha).
We defined the canopy cover threshold for fisher habitat as ≥60% because 95% fixed kernel home ranges for 16 adult female fishers in the Kings River Project area in the Sierra National Forest averaged 63% (Thompson et al. 2011). Furthermore, fisher resting habitat sites are characterized by high canopy cover that is typically >60% (Purcell et al. 2009, Thompson et al. 2011), and the California Wildlife Habitat Relationships database uses a 60% canopy cover threshold as one of the criteria in its definition of high-quality fisher reproductive habitat (California Department of Fish and Game 2008).

We defined a large tree as ≥24 in (61.0 cm) diameter at breast height (dbh) because resting trees at the lower end of the size distribution (i.e., mean minus the standard error) in two different studies were of a similar size (Zielinski et al. 2004, Purcell et al. 2009). Thus, any tree ≥24 in dbh was potentially suitable as a fisher resting site. Next, we determined the threshold density of large trees (i.e., 24 in dbh) by examining stand-level tree lists surrounding den locations of 28 female fishers in the Kings River study area from 2008-2013 (Rebecca Green, unpublished data). When there were multiple dens per female, we randomly chose a single den for that individual. Data for natal dens were used preferentially; natal dens were where the young were born. We used data at maternal den locations for 7 females for which natal den locations were not available; fisher young were moved to maternal dens when conditions were no longer suitable at the natal dens. We defined the threshold density for large trees as ≥15.4 trees/ac (38 trees/ha) because this was the median density of large trees surrounding the 28 den locations.

Results

Basic Fisher Population

A total of 110 individual fishers were captured in live traps as part of the SNAMP Fisher Project from Dec 2007 to Dec 2013 (62 females, 48 males). In the first 3.5 months of trapping in population year 1 we captured 3.6 noncollared (“new”) fishers/100 trap nights, and 6.8 total fishers/100 traps nights. During population years 2008-09 to 2011-12 a mean of 0.94 previously unknown individual fishers were captured per 100 trap nights, and there were an average 2.43 total captures per 100 trap nights. Data on traps nights were not available for 2012-13 or for March to December 2013, when a total of 9 new fishers and 19 total captures occurred (Table D3). No fishers died during capture and handling in the study. However, one adult female fisher captured in October 2009 did not fully recover. The female fisher was held in the trap cubby overnight for additional time
to recover, but died the next morning while in transit to the Fresno Chaffee Zoo for treatment by a wildlife zoo veterinarian. A necropsy completed for the fisher identified her cause of death as septicemia from a previously fractured jaw, which led to emaciation and starvation.

An overarching goal of the study was to monitor a minimum of 20 radicollared fishers at all times, which was considered a requirement for producing reliable estimates of survival and reproduction for the population. The study achieved that milestone in mid July 2008, about six months after live trapping was initiated in late December 2007 (Fig. D10, Table D4). There were brief periods in several years when the radiocollared population declined below 20 individuals (Fig. D10). The annual oscillation in numbers of radiocollared fishers was related to the combination of dropped or shed radiocollars (breakaway units built into radiocollars parted as designed), and mortality which was focused during spring and summer in all years of the study. After the end of our annual pause in live trapping during the spring denning season, the number of radiocollared fishers gradually or rapidly

<table>
<thead>
<tr>
<th>Population Year</th>
<th>Trap nights</th>
<th>New individuals</th>
<th>Recaptures</th>
<th>Total captures</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08</td>
<td>280</td>
<td>10</td>
<td>9</td>
<td>19</td>
</tr>
<tr>
<td>2008-09</td>
<td>2793</td>
<td>34</td>
<td>40</td>
<td>74</td>
</tr>
<tr>
<td>2009-10</td>
<td>2898</td>
<td>20</td>
<td>52</td>
<td>73</td>
</tr>
<tr>
<td>2010-11</td>
<td>2173</td>
<td>15</td>
<td>30</td>
<td>44</td>
</tr>
<tr>
<td>2011-12</td>
<td>1914</td>
<td>22</td>
<td>27</td>
<td>48</td>
</tr>
<tr>
<td>2012-13</td>
<td>No datac</td>
<td>8</td>
<td>11</td>
<td>19</td>
</tr>
<tr>
<td>2013d</td>
<td>No data</td>
<td>1</td>
<td>8</td>
<td>9</td>
</tr>
</tbody>
</table>

a Number of traps set for capture during an overnight period.

b Includes one orphan fisher kit captured in a live trap in 2010-11, and one orphan fisher kit captured in a live trap in 2011-12.

c PSW Forest Service trapping, no data on trap nights.

d Apr 1 to Dec 31; end of SNAMP Fisher.
Figure D7: Number of radio-collared fishers that were monitored for survival and reproduction (females) during the period of SNAMP Fisher from December 2007 to December 2013.
increased when trapping resumed, and as young-of-the-year juvenile fishers were recruiting into the study population in the fall and winter (Fig. D10). With the exception to the first and last year of the study, we were able to monitor survival for at least 40 different fishers in each population year. Notably, in 2011-12 we were monitoring more than 40 individual fishers for several successive months (Fig. D10, Table D4).

**Basic Camera Survey Results**

Camera surveys were a major aspect of SNAMP Fisher in all years. In the overall period of the study we surveyed for fisher presence in 905 unique 1-km² grid cells. The distribution of camera surveys extended from Yosemite Valley in the north, to the slopes above the San Joaquin River canyon to the south and southeast (Fig. D8). Surveys occurred within Yosemite National Park in winter 2009 only, research that was part of a companion study organized by Reginald Barrett and funded by the California Department of Fish and Wildlife. We also obtained data from camera surveys in 24 grid cells located north of the Merced River in Yosemite Valley (not displayed) that were completed by cooperating biologists from Yosemite National Park or the Central Sierra Nevada Environmental Research Center (CSERC). Fishers were not detected in any of the 24 grid cells, reinforcing that the Merced River is the northern edge of the range of fishers in the southern Sierra Nevada.

Fisher activity was identified in 448 of the 905 unique grid cells surveyed (Fig. D8). We used and 6500 feet elevation (1372 and 1981 m elevation). Fisher detections were uncommon above 7500 feet (2286 m) elevation, but the pattern suggested that fishers occasionally use private lands outside of the Sierra National Forest as low as 3000 feet (914 m) elevation (Fig. D9).

Camera effort was focused in the Key Watershed focal study area. The number of 1-km² grid cells surveyed ranged from 122 in 2007-08 and 133 in 2012-13 (Table D5). Across the larger overall SNAMP Fisher study area we surveyed 204 1-km² grid cells in 2012-13 and 409 grid cells in camera

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**Table D4**: Number of radiocollared fishers being monitored for the SNAMP Fisher Project at the start and end of six different population years.

<table>
<thead>
<tr>
<th>Population Year</th>
<th>Start N</th>
<th>End N</th>
<th>Individual fishers N&lt;sup&gt;a&lt;/sup&gt;</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08</td>
<td>--</td>
<td>7</td>
<td>11</td>
</tr>
<tr>
<td>2008-09</td>
<td>6</td>
<td>30</td>
<td>41</td>
</tr>
<tr>
<td>2009-10</td>
<td>30</td>
<td>32</td>
<td>51</td>
</tr>
<tr>
<td>2010-11</td>
<td>32</td>
<td>32</td>
<td>55</td>
</tr>
<tr>
<td>2011-12</td>
<td>32</td>
<td>44</td>
<td>59</td>
</tr>
<tr>
<td>2012-13</td>
<td>44</td>
<td>33</td>
<td>50</td>
</tr>
<tr>
<td>2013&lt;sup&gt;b&lt;/sup&gt;</td>
<td>33</td>
<td>14</td>
<td>33</td>
</tr>
</tbody>
</table>

<sup>a</sup> Number of individual fishers radio-collared and monitored for ≥ 1 day

<sup>b</sup> Apr 1 to Dec 31; end of SNAMP Fisher
Naïve occupancy for all grid cells surveyed varied from ≈ 0.60 in 2008-09 to ≈ 0.40 in 2009-10 
(Table D5). Occupancy for multi-year surveyed grid cells (corrected for probability of detection <1.0) 
oscillated from ≈ 0.80 in 2007-08 to 0.62 in 2009-10 and then increased back to ≈ 0.80 in 2011-12 
(Fig. D10).

In addition to basic naïve occupancy (presence/absence), we assessed fisher activity based on 

**Figure D8**: Distribution of survey grid cells and fisher detections from camera surveys in the overall 
SNAMP Fisher study area in the period from October 2007 to October 2013.
the number of occasions that fishers visited camera stations. Visit occasions were defined as distinct event periods when fishers activated the motion sensors with at least a 15 minute break between successive visits. Review of images suggested this was an appropriate period of time separating distinct visit periods. We scored a total 4727 fisher visits to camera stations during the study (range 583 to 951; Table D6). Fisher visits ranged from 11.6 (2010-11) to 20.4 (2012-13) per 100 trap nights (Table D6). However, and in accordance with our finding of lower probability of detection for fishers during summer season compared to fall and winter seasons (Popescu et al. 2014), fisher visits/100 camera survey days was very low during summer (3.6), and highest during winter (33.3). Higher probability of detection during winter.

**Figure D9:** Elevation range of fishers in the SNAMP Fisher study area based on the proportion of grid cells surveyed with fisher detections in 500 foot (152 m) elevation bins.

**Table D5:** Number of 1km² grid cells surveyed with cameras by camera survey year (Oct 15 to Oct 14).

<table>
<thead>
<tr>
<th>Camera survey year</th>
<th>Key Watersheds</th>
<th>Outside Key Watersheds</th>
<th>Entire study area</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Grid cells</td>
<td>Fisher detected</td>
<td>Naïve occupancy</td>
</tr>
<tr>
<td>2007-08</td>
<td>122</td>
<td>71</td>
<td>0.582</td>
</tr>
<tr>
<td>2008-09</td>
<td>129</td>
<td>75</td>
<td>0.581</td>
</tr>
<tr>
<td>2009-10</td>
<td>127</td>
<td>75</td>
<td>0.591</td>
</tr>
<tr>
<td>2010-11</td>
<td>125</td>
<td>82</td>
<td>0.656</td>
</tr>
<tr>
<td>2011-12</td>
<td>128</td>
<td>98</td>
<td>0.766</td>
</tr>
<tr>
<td>2012-13</td>
<td>133</td>
<td>70</td>
<td>0.526</td>
</tr>
</tbody>
</table>

All years unique grid cells surveyed: \(N = 905\)

*Some grid cells were surveyed twice during a camera survey year; those grid cells were counted once for this summary.*
is likely due to reduced prey availability compared to summer. For example, California ground squirrels (*Otospermophilus beecheyi*) and long-eared chipmunks (*Tamias quadrimaculatus*) enter into torpor (hibernation) during winter, and data on alligator lizards (*Elgaria multicarinata*) and other summer season prey are not available.

**Illustration D9:** Winter period and summer period fisher detections at camera survey stations.

**Table D6:** Summary data on the number of camera days for all cameras used to survey for fishers (effort), and the number of fisher visits during each camera survey year (~Oct 15 to Oct 14).

<table>
<thead>
<tr>
<th>Camera survey year</th>
<th>Camera days (all cameras)</th>
<th>Camera days (Fisher grid cells)</th>
<th>Fisher visits</th>
<th>Visits per 100 camera days</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08</td>
<td>7914</td>
<td>4328</td>
<td>583</td>
<td>13.5</td>
</tr>
<tr>
<td>2008-09</td>
<td>10605</td>
<td>5550</td>
<td>794</td>
<td>14.3</td>
</tr>
<tr>
<td>2009-10</td>
<td>14955</td>
<td>5990</td>
<td>951</td>
<td>15.9</td>
</tr>
<tr>
<td>2010-11</td>
<td>16457</td>
<td>5614</td>
<td>649</td>
<td>11.6</td>
</tr>
<tr>
<td>2011-12</td>
<td>14059</td>
<td>7149</td>
<td>949</td>
<td>13.3</td>
</tr>
<tr>
<td>2012-13</td>
<td>7584</td>
<td>3926</td>
<td>801</td>
<td>20.4</td>
</tr>
</tbody>
</table>

*a* Estimated days that cameras were functioning and focused on the bait tree  
*b* Functional camera days for grid cells with fisher detections  
*c* Based on images sequences with fishers (fisher detections) that were separated by a minimum of 15 minutes.  
*d* Fisher visits divided by functional camera days for grid cells with fisher detections x 100

**Figure D10:** Estimated fisher occupancy (95% CI) for multi-year surveyed (*n* = 292) during six camera survey years for the SNAMP Fisher study. Occupancy is corrected for imperfect probability of detection.
Fisher Denning and Reproduction

Denning period

Den cameras provided detailed information on the activities of 32 adult female fishers during six spring denning seasons. Based on information from the spatial clustering of aerial telemetry locations, ground-based telemetry, and den cameras, denning was initiated in the last week of March in most years (earliest date was March 22), and females typically ceased regular use of den trees in the first week of June (Table D7). The latest known regular use of a den tree was June 20 in spring 2012. It is likely that females continued to use trees as short term den/rest structures during summer when their dependent kits were trailing them, but we did not attempt to systematically identify those types of short duration use structures.

In the spring 2008 den season SNAMP Fisher monitored a single female fisher, but in all other years we monitored at least nine individual females (Table D8). The mean number of dens used per female per season was 2.4 (range 1 to 5), and the mean number of cameras used to monitor each den structure was 3.1. On average each denning female was monitored with den cameras 34.3 days/season (range 28.9 to 37; Table D8), excluding days or periods when successive use maternal den trees were yet to be identified. Fifteen female fishers were monitored with den cameras in one den season, 11 were monitored in two seasons, three were monitored in three seasons, two were monitored in four seasons, and one was monitored in five denning seasons.

Denning activity, litter size and weaning rates

Denning status was determined for 89 of 93 total denning opportunities for breeding-age (≥24 months) females in 6 denning seasons from 2008 to 2013 (Table D9). We were unable to adequately monitor 4 breeding-age females for determining denning status when radiocollars were shed (n = 3) or ceased functioning (n = 1) within the first 31 days of a denning season. The average date that females

Table D7: Estimated dates for the initiation and end of denning by female fishers in the Sierra National Forest, California. Data are from March 2008 to June 2013.

<table>
<thead>
<tr>
<th>Season</th>
<th>Mean start of denning</th>
<th>Mean end of denning</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td># dens</td>
<td>Estimate</td>
</tr>
<tr>
<td>2008</td>
<td>1</td>
<td>27-Mar-08</td>
</tr>
<tr>
<td>2009</td>
<td>12</td>
<td>27-Mar-09</td>
</tr>
<tr>
<td>2010</td>
<td>13</td>
<td>25-Mar-10</td>
</tr>
<tr>
<td>2011</td>
<td>8</td>
<td>1-Apr-11</td>
</tr>
<tr>
<td>2012</td>
<td>11</td>
<td>31-Mar-12</td>
</tr>
<tr>
<td>2013</td>
<td>12</td>
<td>27-Mar-13</td>
</tr>
</tbody>
</table>

*aEstimated from spatial clustering of sequential aerial radiotelemetry locations (Zhao et al 2012).
initiated denning behavior was March 28 (range March 22 to April 9). The average date that females ceased localizing to den trees was June 9 (range May 30 to June 22).

Seventy-six (85%) breeding-age female fishers either exhibited denning behavior \((n = 63)\) or were determined to have denned and weaned at least 1 kit based on size of teats \((n = 13; \text{ Table D9})\). Among 76 breeding-age females that initiated denning, 64 (84%) were identified as weaning kits. Overall, 72% of 89 known status, adequately monitored denning opportunities for breeding-age females produced at least one weaned kit (Table D9).

Eleven (17.5 %) of 63 cases of denning for females that were monitored during spring periods failed prior to kits being weaned (Table D10). Three of the 11 denning failures were females that initiated denning but ceased localizing to natal den trees 17, 35, and 41 days later, potentially related to the death of kits. Eight den failures were due to death of the denning female; seven deaths were by attacks by predators, and one was the result of a denning female either dying of internal bleeding induced by exposure to rodenticides, or from the combination of trauma from being struck by a vehicle on a highway and internal bleeding related to exposure to rodenticides. One of the seven females that died from predator attack was infected with canine distemper virus, which may have contributed to her vulnerability (Keller et al. 2012).
Six of eight deaths of denning females occurred when the locations of den trees were known and were being monitored. In one case den camera images included a bobcat with a dead kit in its mouth, and the partial carcass of the denning female was recovered nearby. In a second case the den structure was a large, unstable snag, and we did not attempt to climb the tree to determine litter size due to safety considerations. In each of the other four cases we climbed the den trees to assess litter size, and recover kits in accordance with California Department of Fish and Wildlife policy. A total five live kits were recovered from two of the den trees (litter size 2, 3), two deceased kits were found in a den cavity of the third tree, and we failed to find kits in the fourth tree.

The five orphan kits that were rescued were raised in captivity by a local wildlife rehabilitation organization licensed by the California Department of Fish and Wildlife, and under the care and supervision of a professional zoo veterinarian. One of the orphan kits died in captivity by urinary tract blockage attributed to a parasitic nematode, whereas the other 4 survived captive rearing. Two kits from one litter were released within their mother’s home range, and the two kits from the second litter were released into an area with suitable fisher habitat abutting the south margin of the study site.

We used a combination of images from den cameras ($n = 43$) and den cavity investigations with a video camera ($n = 4$) to determine litter size for 48 of 59 denning females that were monitored (Table D10). A total 73 kits were known produced, and average litter size was 1.5 (Table D10). After accounting for known mortalities of denning females, we estimated that 64 of the 73 kits produced were weaned from den trees, whereas seven kits died or would have died had they not been rescued (Table D10).
Table D9: Summary of female fisher (≥2 years old) denning and weaning rates by age class and year on the Bass Lake Ranger District in the Sierra National Forest, California, 2008-2013.

<table>
<thead>
<tr>
<th>Age class (Years)</th>
<th>Pop Year</th>
<th>No. Adult Females&lt;sup&gt;a&lt;/sup&gt;</th>
<th>Monitored mid-Mar to May 31&lt;sup&gt;b&lt;/sup&gt;</th>
<th>Teats measured (Jul to Jan)&lt;sup&gt;c&lt;/sup&gt;</th>
<th>Denning&lt;sup&gt;d&lt;/sup&gt;</th>
<th>Proportion denned&lt;sup&gt;e&lt;/sup&gt;</th>
<th>Unknown status&lt;sup&gt;f&lt;/sup&gt;</th>
<th>Failed&lt;sup&gt;g&lt;/sup&gt;</th>
<th>Died while denning</th>
<th>Weaned&lt;sup&gt;h&lt;/sup&gt;</th>
<th>Proportion weaned&lt;sup&gt;i&lt;/sup&gt;</th>
</tr>
</thead>
<tbody>
<tr>
<td>2</td>
<td>2008</td>
<td>11</td>
<td>2</td>
<td>9</td>
<td>9</td>
<td>0.82</td>
<td>9</td>
<td></td>
<td>9</td>
<td>0.82</td>
<td></td>
</tr>
<tr>
<td></td>
<td>2009</td>
<td>17</td>
<td>14</td>
<td>3</td>
<td>15</td>
<td>0.88</td>
<td>1</td>
<td>1</td>
<td>13</td>
<td>0.76</td>
<td></td>
</tr>
<tr>
<td></td>
<td>2010</td>
<td>17</td>
<td>15</td>
<td>1</td>
<td>14</td>
<td>0.88</td>
<td>1</td>
<td>3</td>
<td>11</td>
<td>0.69</td>
<td></td>
</tr>
<tr>
<td></td>
<td>2011</td>
<td>16</td>
<td>11</td>
<td>3</td>
<td>12</td>
<td>0.86</td>
<td>2</td>
<td>1</td>
<td>1</td>
<td>0.71</td>
<td></td>
</tr>
<tr>
<td></td>
<td>2012</td>
<td>17</td>
<td>17</td>
<td>14</td>
<td>1</td>
<td>0.82</td>
<td>3</td>
<td>11</td>
<td>10</td>
<td>0.71</td>
<td></td>
</tr>
<tr>
<td></td>
<td>2013</td>
<td>15</td>
<td>15</td>
<td>12</td>
<td>0.86</td>
<td>1</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

<sup>a</sup> All females ≥ 24 months of age that were known in the population during the year. Includes females that were captured after the end of the denning season in mid-June.

<sup>b</sup> Number of females monitored by radio telemetry during all of part of the period before they died or had a dropped/failed collar after denning status had been determined.

<sup>c</sup> Number of females that were uncollared during the denning period, but were captured during July to January when teat measurements were taken and used to determine weaning status as described by Matthews et al. (2013).

<sup>d</sup> Number of females that exhibited denning behavior, or that were determined to have weaned at least one kit based on teat measurements.

<sup>e</sup> Number of denning females divided by the number of adult females minus the number of females of unknown status.

<sup>f</sup> Number of females (≥ 2 years old) for which denning was unknown or suspected, but dropped or failed radiocollars prevented determination of denning status.

<sup>g</sup> Number of females (≥2 years old) that exhibited denning behavior and ceased denning behavior prior to weaning.

<sup>h</sup> Number of denning females that were known alive and exhibited denning behavior until after May 31.

<sup>i</sup> Number of females that weaned kits, divided by the number of adult females minus the number of females with unknown status.
### Table D10: Information on female fisher kit production for six spring denning seasons (March 21 to June 31) in the Sierra National Forest, California, October 2008 to June 2013.

<table>
<thead>
<tr>
<th>Age class (Years)</th>
<th>Denning females&lt;sup&gt;a&lt;/sup&gt;</th>
<th>Denning Females with kit counts&lt;sup&gt;b&lt;/sup&gt;</th>
<th>Kits&lt;sup&gt;c&lt;/sup&gt;</th>
<th>Litter size&lt;sup&gt;d&lt;/sup&gt;</th>
<th>Denning females deaths&lt;sup&gt;e&lt;/sup&gt;</th>
<th>Known kit deaths&lt;sup&gt;f&lt;/sup&gt;</th>
<th>Denned to Weaning&lt;sup&gt;g&lt;/sup&gt;</th>
<th>Kits weaned&lt;sup&gt;h&lt;/sup&gt;</th>
<th>Kits weaned per litter (fecundity)</th>
</tr>
</thead>
<tbody>
<tr>
<td>2</td>
<td>19</td>
<td>15</td>
<td>21</td>
<td>1.4</td>
<td>1</td>
<td>14</td>
<td>19</td>
<td>1.3</td>
<td></td>
</tr>
<tr>
<td>3-5</td>
<td>32</td>
<td>26</td>
<td>40</td>
<td>1.6</td>
<td>3</td>
<td>7</td>
<td>22</td>
<td>34</td>
<td>1.3</td>
</tr>
<tr>
<td>≥6</td>
<td>8</td>
<td>7</td>
<td>12</td>
<td>1.7</td>
<td></td>
<td>7</td>
<td>12</td>
<td>1.7</td>
<td></td>
</tr>
<tr>
<td>Year</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2008</td>
<td>2</td>
<td>1</td>
<td>1</td>
<td></td>
<td>1</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2009</td>
<td>12</td>
<td>9</td>
<td>15</td>
<td>1.5</td>
<td>1</td>
<td>10</td>
<td>15</td>
<td>1.5</td>
<td></td>
</tr>
<tr>
<td>2010</td>
<td>13</td>
<td>11</td>
<td>20</td>
<td>1.8</td>
<td>3</td>
<td>7</td>
<td>7</td>
<td>13</td>
<td>1.9</td>
</tr>
<tr>
<td>2011</td>
<td>8</td>
<td>7</td>
<td>11</td>
<td>1.6</td>
<td>1</td>
<td>7</td>
<td>11</td>
<td>1.6</td>
<td></td>
</tr>
<tr>
<td>2012</td>
<td>14</td>
<td>11</td>
<td>16</td>
<td>1.5</td>
<td>3</td>
<td>2</td>
<td>10</td>
<td>14</td>
<td>1.4</td>
</tr>
<tr>
<td>2013</td>
<td>10</td>
<td>8</td>
<td>10</td>
<td>1.3</td>
<td></td>
<td>8</td>
<td>10</td>
<td>1.3</td>
<td></td>
</tr>
</tbody>
</table>

<sup>a</sup> Number of females (≥2 years old) that exhibited denning behavior and were monitored by radiotelemetry, den cameras, or both. Excludes females whose reproductive status was not known and those that initiated denning behavior but ceased denning before May.

<sup>b</sup> Number of denning females for which kit counts were determined by images from den cameras, den cavity video camera, or both.

<sup>c</sup> Total number of kits counted.

<sup>d</sup> Number of denning females with kit counts divided by the number of kits counted.

<sup>e</sup> Number of denning females known to have died during the denning season while provisioning kits in den trees. Numbers of kits in litters were not known for all of the denning females that died.

<sup>f</sup> Kits that were known present in den trees when the mother died, or those that were found dead inside den cavities. This estimate assumes that 5 orphan kits that were removed from den cavities would have perished if they had not been rescued.

<sup>h</sup> Number of monitored denning female fishers exhibiting denning behavior that continued to weaning.
Denning structures

We identified 125 unique structures used as natal or maternal dens, including 54 black oak, 41 incense cedar, 19 white fir, 10 sugar pine or ponderosa pine, and one canyon oak (*Quercus chrysolepsis*) (Table D11).

Repeat use of den trees was not uncommon. Sixteen individual den trees were used more than once; 15 trees were used in two years, and one tree was used in four different den seasons. In all but two cases of repeat den tree use the same individual reused one or several den trees between successive years. In two cases a female used a den that had been used by a different female in a previous year. Successive dens of females that used more than 1 den structure were located an average of 413 m apart (*n* = 52, range 75-1398). The distance between the natal den tree and the first maternal den tree averaged 419 m, whereas successive use maternal den trees were in closer proximity (mean = 287 m, *t*<sub>69</sub> =1.75, *P*=0.04).

Fifty-six percent of the

<table>
<thead>
<tr>
<th>Tree type, Species</th>
<th>Denning events&lt;sup&gt;a&lt;/sup&gt;</th>
<th>Percent within group</th>
<th>Unique structures&lt;sup&gt;b&lt;/sup&gt;</th>
<th>Repeat use structures&lt;sup&gt;c&lt;/sup&gt;</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Live trees</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Black oak</td>
<td>34</td>
<td>43</td>
<td>31</td>
<td>3</td>
</tr>
<tr>
<td>Incense cedar</td>
<td>25</td>
<td>32</td>
<td>19</td>
<td>4</td>
</tr>
<tr>
<td>White fir</td>
<td>14</td>
<td>18</td>
<td>14</td>
<td></td>
</tr>
<tr>
<td>Sugar pine</td>
<td>3</td>
<td>4</td>
<td>3</td>
<td></td>
</tr>
<tr>
<td>Ponderosa pine</td>
<td>2</td>
<td>3</td>
<td>2</td>
<td></td>
</tr>
<tr>
<td>Canyon oak</td>
<td>1</td>
<td>1</td>
<td>1</td>
<td></td>
</tr>
<tr>
<td><strong>Snags</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Black oak</td>
<td>27</td>
<td>42</td>
<td>23</td>
<td></td>
</tr>
<tr>
<td>Incense cedar</td>
<td>27</td>
<td>42</td>
<td>22</td>
<td>4</td>
</tr>
<tr>
<td>White fir</td>
<td>5</td>
<td>8</td>
<td>5</td>
<td>5</td>
</tr>
<tr>
<td>Pine species&lt;sup&gt;d&lt;/sup&gt;</td>
<td>5</td>
<td>8</td>
<td>5</td>
<td></td>
</tr>
<tr>
<td><strong>Live tree or snag</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Black oak</td>
<td>61</td>
<td>43</td>
<td>54</td>
<td>3</td>
</tr>
<tr>
<td>Incense cedar</td>
<td>52</td>
<td>36</td>
<td>41</td>
<td>8</td>
</tr>
<tr>
<td>White fir</td>
<td>19</td>
<td>13</td>
<td>19</td>
<td>5</td>
</tr>
<tr>
<td>Pine species</td>
<td>10</td>
<td>7</td>
<td>10</td>
<td></td>
</tr>
<tr>
<td>Canyon oak</td>
<td>1</td>
<td>1</td>
<td>1</td>
<td></td>
</tr>
<tr>
<td><strong>Total den structures</strong></td>
<td>143</td>
<td>125</td>
<td>16</td>
<td></td>
</tr>
</tbody>
</table>

<sup>a</sup> Count of all known denning events for each species of tree.
<sup>b</sup> Count of individual trees; those used in multiple seasons counted once.
<sup>c</sup> Number of individual trees used ≥ two times for denning; one live cedar tree was used by the same female in four successive denning seasons, but all other repeat use trees were known used in two den seasons only.
<sup>d</sup> Pine snags could not always be identified as sugar pine or ponderosa.
unique individual trees used for denning in the SNAMP area were live trees \((n = 70)\), whereas 44\% \((n = 55)\) were snags (Table D11). Black oak was the most common live tree used for denning, followed by incense cedar (Table D11). Among snags used as denning structures, black oak and incense cedar were both commonly used, whereas white fir and pines (sugar pine or ponderosa pine) were less common as snag-type den trees (Table D11). Overall, black oaks and incense cedar were the two most common tree species used for denning (Table D11).

Habitat characteristics of den structures

Mean diameter at breast height (DBH) of black oak denning structures was smaller than that for other tree species used (Table D12). Mean heights of live trees were taller than snags of the same species (Fig. D11), reflecting that many of the snags used for denning were at advanced stages of decay.

**Table D12:** Basic information on the size (DBH) and height of trees (live or snag) used as denning structures by female fishers in the SNAMP Fisher study from March 2008 to June 2013.

| Tree species | Live trees | | Snags or dead trees | | |
|--------------|------------|----------------|-------------------|----------------|
|              | \(n\)      | Mean DBH (cm) | Mean height (m) \(a\) | \(n\) | Mean DBH (cm) | Mean height (m) |
| Black oak    | 30         | 74.2          | 21.7              | 5     | 69.5          | 8.8             |
| Incense cedar| 18         | 127.2         | 32.5              | 22    | 105.1         | 16.4            |
| White fir    | 14         | 110.8         | 33.9              | 22    | 103.7         | 27.4            |
| Pines        | 5          | 112.8         | 37.4              | 5     | 109.6         | 27.6            |

\(a\) Data on mean tree height are for the subset of den trees for which detailed data on habitat measurements were completed (\(n = 84\)).

**Figure D11:** Summary information on the (a) mean height of denning structures (unique trees), and the (b) elevation range for fisher den trees for the SNAMP Fisher Study during 2008 to 2013 (6 denning seasons).
The majority of denning structures used by fishers in the SNAMP Fisher study area were in the elevation range from 4500 (1371 m) to 6000 feet (1829 m; 83%, \( n = 104 \); Fig. D11b). Additional information obtained from circular habitat plots assessments included indications of high canopy cover, limited herbaceous cover, and relatively low shrub cover near most den trees (Table D13). Concealment cover was 64% low ground cover, 46% high ground cover, and 38% and 36% low shrub and high shrub cover, respectively. On average, belt transects within the circular habitat plots around den trees included an average of 6.5 down logs (coarse woody debris, CWD; logs/branches with a minimum large end diameter of 15 cm, ≥1 m total length). Many denning structures were on steep slopes (Table D13) but there was no obvious preference for aspect (Fig. D12).

<table>
<thead>
<tr>
<th>Attribute</th>
<th>Mean</th>
<th>Range</th>
</tr>
</thead>
<tbody>
<tr>
<td>Canopy cover</td>
<td>72%</td>
<td>30-94%</td>
</tr>
<tr>
<td>Shrub cover</td>
<td>19%</td>
<td>0-82.5%</td>
</tr>
<tr>
<td>Herbaceous cover</td>
<td>6%</td>
<td>0-29%</td>
</tr>
<tr>
<td>Prevailing slope</td>
<td>37%</td>
<td>3-75%</td>
</tr>
</tbody>
</table>

Table D13: Basic habitat attributes around fisher den trees for the SNAMP Fisher Study area in spring 2008 to spring 2012.

Northerly (Range: 316° - 45°) N = 23 trees
Westerly (Range: 226° - 315°) N = 21 trees
Easterly (Range: 46° - 135°) N = 20 trees
Southerly (Range: 136° - 225°) N = 15 trees

Figure D12: Aspect of fisher den structures on the SNAMP Fisher Study. Data are for 79 den trees.
Activity patterns of denning females

Additional insight on denning activities by adult female fishers was provided by analyses of den camera images. Adult females were detected by den cameras at known active dens an average of 0.64 times/day (range 0.57 to 0.73) (Table D8) and the mean number of detections of up and down movements ranged from 1.1 to 1.3 per day (Table D8), indicating that fishers do not typically leave and return to den trees multiple times a day. In addition to information on male visits to den trees (we obtained image sequences of eight mating, or copulation events at the base of den trees), den cameras identified three occasions when a female fisher briefly returned to a den tree at least one day after she had already moved kits to another tree nearby (Table D14). On eight occasions den cameras detected other female fishers (non-collared or different collared fisher) at den trees of female fishers (Table D14).

Information from the 83 occasions when females were detected moving kits was used to estimate fecundity. A total of 1295 detections were identified as female fishers departing from, or returning to the den tree, whereas there were 99 detections of females at or near the base of den trees that could not be unequivocally classified except as active outside the den cavity (Table D14). We were able to identify 316 image sequences consistent with either continuous den attendance, or
continuous time away from the den when denning females were likely foraging. Den attendance bouts were shortest late in the den season and longest in the middle of the den season (Table D15). Forage bouts away from den trees were shortest early in the den season, and approximately equal thereafter (Table D15).

<table>
<thead>
<tr>
<th>Table D14: Summary data on denning activities by female fishers determined from monitoring den trees with remote cameras. Data are from the Sierra National Forest, California from April 2008 to June 2012.</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Spring</strong></td>
</tr>
<tr>
<td>------------</td>
</tr>
<tr>
<td>2008</td>
</tr>
<tr>
<td>2009</td>
</tr>
<tr>
<td>2010</td>
</tr>
<tr>
<td>2011</td>
</tr>
<tr>
<td>2012</td>
</tr>
<tr>
<td>2013&lt;sup&gt;c&lt;/sup&gt;</td>
</tr>
</tbody>
</table>

<sup>a</sup> Detections at base of tree, or on the tree for which directionality or activity was uncertain  
<sup>b</sup> Detections when the female was carrying objects as they returned and ascended the den tree  
<sup>c</sup> General information only available for 2013.

<table>
<thead>
<tr>
<th>Table D15: Information on den attendance and foraging excursions, developed from analyses of data from cameras used to monitor fisher den trees during five denning seasons. Data are from the Sierra National Forest, California from April 2008 to June 2012.</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Den attendance bouts (minutes)</strong></td>
</tr>
<tr>
<td>Season&lt;sup&gt;a&lt;/sup&gt;</td>
</tr>
<tr>
<td>Early</td>
</tr>
<tr>
<td>Middle</td>
</tr>
<tr>
<td>Late</td>
</tr>
<tr>
<td>Overall</td>
</tr>
</tbody>
</table>

<sup>a</sup> Seasons were Early (March 26 to April 20), Middle (April 21 to May 15), and Late, (May 16 to June 11), identified by dividing the overall den season into three 25 day periods from late March to mid-June.  
<sup>b</sup> Shortest duration bout.  
<sup>c</sup> Longest duration bout.
Fisher Survival

Sixty-six (60%) of the 110 individual fishers radiocollared during the study were known to have died, including 32 females and 34 males (Table D16). Excluding population year 2007-08, an average of 10.5 radiocollared fishers perished each population year (Fig. D13). The mean number of deaths by sex for population year 2008-09 through population year 2013-14 was 5.3 for females and 5.2 for males. Fisher survival with population data combined into 2-year periods was generally higher for adult and juvenile fishers than for subadults (Table D17). Ninety-five percent confidence intervals overlapped for females and males in all two-year period with the possible exception of subadults in year group 3. Two-year survival rates among females ranged from a low of 47% to a high of 89% for subadult females (Table D17). Two year survival for juvenile females was always ≥ 74%, whereas among adult females it ranged from a low of 0.69% to a high of 0.86 (Table D17). Fisher survival for all years combined was highest for juvenile females and lowest for subadult males (Table D17). Also, although not significantly different, survival was consistently higher for females compared to males (all age classes; Table D17).
Table D16: Review of all known deaths of radiocollared fishers in seven population years (Apr 1 to Mar 31), summarized by sex, and cause-specific mortality from necropsy examinations by pathologists at the UC Davis School of Veterinary Medicine (Davis, CA).

<table>
<thead>
<tr>
<th>Year, Sex</th>
<th>Predation&lt;sup&gt;a&lt;/sup&gt;</th>
<th>Disease&lt;sup&gt;b&lt;/sup&gt;</th>
<th>Starvation-related injury, septicemia&lt;sup&gt;c&lt;/sup&gt;</th>
<th>Roadkill</th>
<th>Rodenticide toxicosis</th>
<th>Indeterminate, unknown&lt;sup&gt;d&lt;/sup&gt;</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
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</tr>
<tr>
<td>Female</td>
<td>1</td>
<td>1</td>
<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>Male</td>
<td>1</td>
<td>1</td>
<td></td>
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<td></td>
</tr>
<tr>
<td>2008-09</td>
<td></td>
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</tr>
<tr>
<td>Female</td>
<td>4</td>
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<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Male</td>
<td>1</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2009-10</td>
<td></td>
<td></td>
<td></td>
<td></td>
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<td></td>
</tr>
<tr>
<td>Female</td>
<td>5</td>
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<tr>
<td>Male</td>
<td>3</td>
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<td>2010-11</td>
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<td></td>
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<tr>
<td>Female</td>
<td>5</td>
<td>1</td>
<td></td>
<td>1</td>
<td>1</td>
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</tr>
<tr>
<td>Male</td>
<td>4</td>
<td>1</td>
<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>2011-12</td>
<td></td>
<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>3</td>
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<td></td>
</tr>
<tr>
<td>Male</td>
<td>5</td>
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</tr>
<tr>
<td>2012-13</td>
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</tr>
<tr>
<td>Female</td>
<td>3</td>
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<td></td>
<td></td>
</tr>
<tr>
<td>Male</td>
<td>1</td>
<td></td>
<td></td>
<td>1</td>
<td>1</td>
<td>1</td>
</tr>
<tr>
<td>2013&lt;sup&gt;e&lt;/sup&gt;</td>
<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
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<td></td>
<td></td>
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<tr>
<td>Male</td>
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<td></td>
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<td></td>
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<tr>
<td>All years</td>
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</tr>
<tr>
<td>Female</td>
<td>20</td>
<td>3</td>
<td></td>
<td>1</td>
<td>1</td>
<td>1</td>
</tr>
<tr>
<td>Male</td>
<td>12</td>
<td>5</td>
<td></td>
<td>3</td>
<td>2</td>
<td>2</td>
</tr>
</tbody>
</table>

<sup>a</sup> One female death by predation in 2009-10 may have been related to the animal being weakened/sick from CDV when it encountered a coyote (*Canis latrans*); further discussed by Keller et al. (2012).

<sup>b</sup> Three disease deaths were associated with canine-distemper virus, one was considered by Toxoplasmosis, and one was due to pleruritus+pneumonia.

<sup>c</sup> Most deaths in this category were associated with prior injury that contributed to starvation and septicemia.

<sup>d</sup> Necropsies were completed, but cause of death could not be determined.

<sup>e</sup> Includes deaths of two male fishers that died January 1, 2014 and March 31, 2014. Although this was after the end of SNAMP Fisher, both of the animals were radiocollared as part of SNAMP.
Causes of Mortality

Necropsies were completed for 50 of the 66 radiocollared fishers that died during the SNAMP Fisher study. Assignment of cause-specific mortality was possible for 47 of the 50 animals with necropsy reports (94%). Three necropsy reports were indeterminate with regards cause of death for the fisher (Table D16). To date, a known cause of death has been determined for 71% of the 66 mortalities. Among known-cause mortalities predation was the primary cause of death, accounting for 68% of 47 known-cause deaths (Fig. D14). Deaths by disease, injury-related starvation or septicemia, and human-linked factors such as vehicle strike or rodenticide poisoning combined to account for 32% of known-cause mortalities (Fig. D14).

Table D17: Estimates of survival (s(t)), for radiocollared fishers using population data combined for analysis into a series of five 2-year groups beginning in population year 2 (2008-09) and ending in population year 7 (2013-14), and for all years of data combined. Survival was assessed using Kaplan-Meier staggered entry analyses. Population years were from April 1 to March 31, and ages were defined as juvenile [≤ 12 months], subadults [12 to 23 months], and adults [≥ 24 months].

<table>
<thead>
<tr>
<th>Year group, Sex</th>
<th>Juveniles</th>
<th>Subadults</th>
<th>Adults</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>s(t)</td>
<td>95% CI</td>
<td>s(t)</td>
</tr>
<tr>
<td>2008-09, 2009-10</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.80</td>
<td>0.58-1.02</td>
<td>0.47</td>
</tr>
<tr>
<td>Male</td>
<td>0.83</td>
<td>0.50-1.17</td>
<td>0.40</td>
</tr>
<tr>
<td>2009-10, 2010-11</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.80</td>
<td>0.59-1.01</td>
<td>0.67</td>
</tr>
<tr>
<td>Male</td>
<td>0.60</td>
<td>0.30-0.90</td>
<td>0.43</td>
</tr>
<tr>
<td>2010-11, 2011-12</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.74</td>
<td>0.54-0.94</td>
<td>0.89</td>
</tr>
<tr>
<td>Male</td>
<td>0.67</td>
<td>0.42-0.92</td>
<td>0.50</td>
</tr>
<tr>
<td>2011-12, 2012-13</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.80</td>
<td>0.55-1.05</td>
<td>0.73</td>
</tr>
<tr>
<td>Male</td>
<td>0.83</td>
<td>0.56-1.11</td>
<td>0.92</td>
</tr>
<tr>
<td>2012-13, 2013-14 a</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.73</td>
<td>0.48-0.98</td>
<td>0.74</td>
</tr>
<tr>
<td>Male</td>
<td>0.75</td>
<td>0.33-1.17</td>
<td>0.66</td>
</tr>
<tr>
<td>All Years; Dec 07-Mar-14</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>0.75</td>
<td>0.60-0.89</td>
<td>0.71</td>
</tr>
<tr>
<td>Male</td>
<td>0.60</td>
<td>0.42-0.78</td>
<td>0.57</td>
</tr>
</tbody>
</table>

a Insufficient data for estimating survival for juveniles in this Year group.
Predation accounted for nearly twice as many known cause deaths for females (43%) than for males (26%), whereas all of the disease and roadkill deaths were males (Fig. D14).

Serological testing of blood samples collected at captures revealed low levels of exposure to canine distemper virus in the study population (Gabriel 2013). However, in spring 2009 a relatively small scale epizootic of CDV occurred in the study population, contributing to the deaths of four fishers; three by direct infection, and one that was killed by a coyote attack, but was likely weakened due to presence of CDV infection (Table D16, Fig. D14; Keller et al. 2012).

In Spring 2009, the SNAMP Fisher Team recovered the first fisher known to have died by toxicosis after exposure to rodenticides. In total, three fishers were known to have died after exposure to rodenticides as of June 2014, including two males and one female. The discovery of death associated with rodenticides led to two peer-reviewed papers. One detailed issues with anticoagulant rodenticides on public lands (Gabriel et al. 2012) and a second paper revealed that female fishers with larger numbers of marijuana grow sites within their home ranges experience reduced survival (Thompson et al. 2013).

**Illustration D11:** Remains of a female fisher killed by a predator (left), and a male fisher that was determined to have died by infectious disease (right).
Population Growth Rates

Empirically developed estimates of key demographic parameters needed to estimate a deterministic growth rate for the population ($\lambda$) were developed during the study (Tables D22, D23). Estimates for $\lambda$ were below 1.0 (population decline) in two 2-year groups, equal to 1.0 in one 2-year group (stable), and slightly positive in two 2-year groups (increasing population) (Table D18). The All Years $\lambda$ was 0.90, which was suggestive of population decline, however, the range for all results overlapped 1.0.

Table D18: Demographic parameters and deterministic population growth rates (range) for five two-year groups$^a$, and for population data for all years of the study combined (All years).

<table>
<thead>
<tr>
<th>Parameter, Age class</th>
<th>Year group 1</th>
<th>Year group 2</th>
<th>Year group 3</th>
<th>Year group 4</th>
<th>Year group 5</th>
<th>All Years</th>
</tr>
</thead>
<tbody>
<tr>
<td>Weaning reproduction</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Young adult</td>
<td>0.67</td>
<td>0.89</td>
<td>1.00</td>
<td>0.83</td>
<td>0.70</td>
<td>0.68</td>
</tr>
<tr>
<td>Adult</td>
<td>0.75</td>
<td>0.67</td>
<td>0.83</td>
<td>0.82</td>
<td>0.87</td>
<td>0.74</td>
</tr>
<tr>
<td>Weaning litter size</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Young adult</td>
<td>1.27</td>
<td>1.57</td>
<td>1.50</td>
<td>1.20</td>
<td>1.20</td>
<td>1.19</td>
</tr>
<tr>
<td>Adult</td>
<td>1.31</td>
<td>1.31</td>
<td>1.60</td>
<td>1.55</td>
<td>1.40</td>
<td>1.45</td>
</tr>
<tr>
<td>Weaning fecundity ($b_i$)$^b$</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Young adult</td>
<td>0.42</td>
<td>0.70</td>
<td>0.75</td>
<td>0.50</td>
<td>0.42</td>
<td>0.41</td>
</tr>
<tr>
<td>Adult</td>
<td>0.49</td>
<td>0.44</td>
<td>0.67</td>
<td>0.64</td>
<td>0.61</td>
<td>0.53</td>
</tr>
<tr>
<td>Survival ($P_i$)</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
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</tr>
<tr>
<td>Juvenile</td>
<td>0.76</td>
<td>0.80</td>
<td>0.74</td>
<td>0.80</td>
<td>1.00</td>
<td>0.75</td>
</tr>
<tr>
<td>Subadult</td>
<td>0.47</td>
<td>0.67</td>
<td>0.89</td>
<td>0.73</td>
<td>0.73</td>
<td>0.71</td>
</tr>
<tr>
<td>Adult</td>
<td>0.81</td>
<td>0.70</td>
<td>0.69</td>
<td>0.86</td>
<td>0.74</td>
<td>0.73</td>
</tr>
<tr>
<td>Fertility ($b_iP_i$)</td>
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<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Young adult</td>
<td>0.20</td>
<td>0.47</td>
<td>0.67</td>
<td>0.36</td>
<td>0.31</td>
<td>0.29</td>
</tr>
<tr>
<td>Adult</td>
<td>0.40</td>
<td>0.30</td>
<td>0.46</td>
<td>0.55</td>
<td>0.45</td>
<td>0.39</td>
</tr>
<tr>
<td>Leslie Matrix $\lambda^c$</td>
<td>0.87 (0.65-1.08)</td>
<td>0.88 (0.63-1.12)</td>
<td>1.00 (0.77-1.22)</td>
<td>1.04 (0.81-1.26)</td>
<td>1.03 (0.77-1.22)</td>
<td>0.90 (0.71-1.12)</td>
</tr>
</tbody>
</table>

$^a$ Two-year groups were 2008-09 and 2009-10 (1), 2009-10 and 2010-11 (2), 2010-11 and 2011-12 (3), 2011-12 and 2012-13 (4), and 2012-13 and 2013-14 (5).

$^b$ Fecundity is the number of female offspring produced, calculated as weaning reproduction*weaning litter size*0.5 (assumes equal sex ratio at birth).

$^c$ The range for $\lambda$ was based on the 95% confidence intervals for the survival rates for the five two-year groups (Table D17). The range for $\lambda$ for the All years data was based on the 95% CIs for the means for weaning reproductive rate and litter size, and for the 95% CIs for age-specific survival (Table D17).
Population Size and Density

Population size was estimated for the middle four population years of the six year study. In population year 1 (2007-08), we had only a small number of fishers radiocollared during the last few months of that period (Tables D9, Fig. D10), and camera images for the entire population year 2012-2013 were not available due to the conclusion of SNAMP Fisher field work. During the central four year period we captured and radiocollared 101 individual fishers (57 females and 44 males) on 258 occasions during 9732 trap-nights between December 2007 and March 2012. Resighting efforts, both by camera and live traps, varied by subregion and, to a lesser extent, year (Table D19). Cameras accounted for 86% of 1421 total radio-marked fisher detections, with live trap recaptures providing 201 sightings.

Mean overall abundance across all subregions ranged from 48.2 individuals in Year 2 to 61.8 individuals in Year 4. Variation was at least partly related to differences in area surveyed among years (Table D20). Estimates of areas sampled were generally consistent within subregions among years (Table D20). The increase in area surveyed in Subregion 1 in fall-winter 2009-10 was due to a program that extended camera surveys north into the Yosemite South region of Yosemite National Park (Fig. D2) in winter 2010. In fall-

| Table D19: Summary data on camera and live trap activities within 4 fall-winter camera survey years (October 16 to March 15) in the Bass Lake District, Sierra National Forest Study area, October 2008 to March 2012. (see Figure D6 for subregion map). |
|------------------|------------------|------------------|------------------|------------------|
| Subregion, Year | Camera surveys   | Live traps       | Estimated area surveyed (km^2)^a |
|                  | Grid cells       | Grid cells       |                               |
|                  | Nights           | Nights           |                               |
| Subregion 1. Nelder Grove, Sugar Pine, Miami Mountain |
| 2008-09          | 147              | 121              | 223.2                        |
| 2009-10          | 160              | 161              | 307.2                        |
| 2010-11          | 132              | 72               | 214.3                        |
| 2011-12          | 141              | 147              | 224.6                        |
| Subregion 2. Central Camp, Whisky, Grizzly, Jackass |
| 2008-09          | 48               | 17               | 267.6                        |
| 2009-10          | 12               | 56               | 248.0                        |
| 2010-11          | 20               | 47               | 244.2                        |
| 2011-12          | 65               | 80               | 305.5                        |
| Subregion 3. Chowchilla Mountain, Rush Creek, Sweetwater |
| 2008-09          | 16               | 25               | 128.8                        |
| 2009-10          | 2                | 39               | 111.8                        |
| 2010-11          | 1                | 22               | 136.2                        |
| 2011-12          | 14               | 32               | 132.8                        |

^a Based on a 1300 m buffer applied to polygons encompassing grid cells surveyed by cameras and grid cells with live trap captures.
winter 2011-12 search effort was expanded in the Grizzly and Jackass subregion when non-collared fishers were detected on camera in areas that had not been surveyed previously. Mean annual population density for the three subregions ranged from 0.072 to 0.097 fishers/km² (Fig. D15).

Subregion 1 had consistently high average densities (0.073-0.125 individuals/km²), with an increasing trend across the last three years of the period (Table D20, Fig. D15). Subregion 3 had initial low density (0.056 ± 0.005 individuals/km²), but gradually increased by the end of the period (0.106 ± 0.005 individuals/km²). Subregion 2 showed no particular trend, and average densities varied across seasons between 0.066 (fall-winter 2009-10) and 0.092 individuals/km² (fall-winter 2010-11). Temporally, mean population density was lowest in fall-winter 2009-10 at 0.075 ± SE 0.006 individuals/km², and increased thereafter to a high of 0.097 ± SE 0.008 in fall-winter 2011-12 (Fig. D15). Mean population density was consistently high in the last 2 years of the study across all subregions (0.089 – 0.106 individuals/km²).

### Table D20: Mark-resight estimates of population size for three subregions in 4 Fall-Winter survey years (October 16 to March 15) in the Bass Lake District, Sierra National Forest, October 2008 to March 2012.

<table>
<thead>
<tr>
<th>Subregion, Year</th>
<th>n</th>
<th>95% C.I.</th>
<th>Density(^a)</th>
<th>Density range(^b)</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Subregion 1: Nelder Grove, Sugar Pine, Miami Mtn</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2008-09</td>
<td>27.9</td>
<td>23.6-32.2</td>
<td>0.125</td>
<td>0.106-0.144</td>
</tr>
<tr>
<td>2009-10</td>
<td>22.3</td>
<td>19.0-25.6</td>
<td>0.073</td>
<td>0.062-0.083</td>
</tr>
<tr>
<td>2010-11</td>
<td>19.1</td>
<td>16.3-22.0</td>
<td>0.089</td>
<td>0.076-0.103</td>
</tr>
<tr>
<td>2011-12</td>
<td>23.2</td>
<td>20.2-26.2</td>
<td>0.103</td>
<td>0.090-0.117</td>
</tr>
<tr>
<td><strong>Subregion 2: Central Camp, Whisky Ridge, Grizzly, Jackass</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2008-09</td>
<td>18.8</td>
<td>10.5-21.2</td>
<td>0.070</td>
<td>0.044-0.097</td>
</tr>
<tr>
<td>2009-10</td>
<td>16.3</td>
<td>10.4-21.4</td>
<td>0.066</td>
<td>0.037-0.094</td>
</tr>
<tr>
<td>2010-11</td>
<td>22.5</td>
<td>15.4-24.5</td>
<td>0.092</td>
<td>0.066-0.118</td>
</tr>
<tr>
<td>2011-12</td>
<td>24.6</td>
<td>17.8-26.5</td>
<td>0.080</td>
<td>0.062-0.099</td>
</tr>
<tr>
<td><strong>Subregion 3: Chowchilla Mtn, Rush Creek, Sweetwater</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2008-09</td>
<td>7.2</td>
<td>5.8-8.6</td>
<td>0.056</td>
<td>0.045-0.067</td>
</tr>
<tr>
<td>2009-10</td>
<td>9.7</td>
<td>8.7-10.6</td>
<td>0.086</td>
<td>0.078-0.095</td>
</tr>
<tr>
<td>2010-11</td>
<td>10.0</td>
<td>8.8-11.3</td>
<td>0.074</td>
<td>0.065-0.083</td>
</tr>
<tr>
<td>2011-12</td>
<td>14.0</td>
<td>12.8-15.3</td>
<td>0.106</td>
<td>0.096-0.115</td>
</tr>
</tbody>
</table>

\(^a\) Population size \((n)\) divided by the estimated sample area for the subregion in the Fall-Winter camera survey year, included in Table D1.

\(^b\) Calculated based on the lower and upper values of the 95% C.I., divided by the estimate of the sampled area provided in Table D1.
Dispersal Behavior and Movements

The combination of field data and genetic data allowed for the possibility of assessing dispersal for 33 female and 25 male fishers that were captured as juveniles ($n = 53$), or young subadults ($n = 8$; ≤18 months old) (Table D21). Fifteen of those fishers (25.8%) died, disappeared, or were caught too late in the year to define a juvenile home range (Table D21). Dispersal was assessed for 43 (74%) of the 58 animals, based on identification of likely natal areas from either field data or genetic-based maternity assignments (Table D21).

Considering data for dispersal using either field or genetic-based natal area determination and based on Euclidean distances, male fishers tended to disperse longer distances than females, but the difference was not significant (Table D28). The longest Euclidean distance dispersal for a female fisher was 24.53 km, compared to 36.17 km for a male fisher; however the large range of dispersal distances for both sexes precludes precise statistical comparison.

Euclidean dispersal movements often originated from within the Key Watershed focal study area, but other Subregions of the study area produced dispersing animals as well (Fig. D16). There was no clear patterning with regards directionality of dispersal, except perhaps the general northwest to southeasterly orientation associated with the Sierra Nevada range (Fig. D16).

One male fisher immigrated into the SNAMP Fisher Study area in the Bass Lake Ranger District from south of Shaver Lake within the High Sierra District. This fisher, KRFP ID M38 (SNAMP ID M47), was originally captured and marked with a PIT tag on the Kings River Fisher Project in December 2010. M38 was recaptured by the KRFP researchers in February 2012, when he was released without a radiocollar due to a neck injury. M38 was captured 13 months later in March 2013 within the SNAMP study area. Although his Euclidean distance-based dispersal track was
estimated at ≈ 36 km, it is more likely that his dispersal track was more circuituous, and in the range of 67-69 km (Fig. D17).

Dispersal movements predicted by Least Cost Movement (LCP) analyses over landscape features considered restrictive to fishers produced longer mean dispersal distances than Euclidean paths (Table D23, Fig. D18). Nevertheless, and in accordance with data from Euclidean distances, there was no evidence for a significant sex-bias in LCP predicted dispersal tracks (Table D23).

Table D21: Review of information on juvenile or subadult fishers captured on the SNAMP Fisher study for which dispersal assessments were possible from field data, maternal assignments from genetic analyses, or from either source.

<table>
<thead>
<tr>
<th>Maternal year, Sex</th>
<th>n</th>
<th>Died</th>
<th>Missing, disappear</th>
<th>Late capture</th>
<th>Dispersal not assessed</th>
<th>Field</th>
<th>Genetics</th>
<th>Both</th>
<th>Total</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007 Female</td>
<td>3</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>2</td>
<td>2</td>
<td>2</td>
<td>2</td>
</tr>
<tr>
<td>2007 Male</td>
<td>3</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>2</td>
<td>1</td>
<td>1</td>
<td>2</td>
</tr>
<tr>
<td>2008 Female</td>
<td>8</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>4</td>
<td>5</td>
<td>3</td>
<td>6</td>
</tr>
<tr>
<td>2008 Male</td>
<td>4</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>3</td>
<td>3</td>
<td>3</td>
<td>3</td>
</tr>
<tr>
<td>2009 Female</td>
<td>7</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>5</td>
<td>3</td>
<td>3</td>
<td>5</td>
</tr>
<tr>
<td>2009 Male</td>
<td>4</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>3</td>
<td>3</td>
<td>3</td>
<td>3</td>
</tr>
<tr>
<td>2010 Female</td>
<td>6</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>5</td>
<td>4</td>
<td>4</td>
<td>5</td>
</tr>
<tr>
<td>2010 Male</td>
<td>8</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>5</td>
<td>6</td>
<td>4</td>
<td>7</td>
</tr>
<tr>
<td>2011 Female</td>
<td>7</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>4</td>
<td>6</td>
<td>4</td>
<td>6</td>
</tr>
<tr>
<td>2011 Male</td>
<td>4</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>3</td>
<td>3</td>
<td>3</td>
<td>3</td>
</tr>
<tr>
<td>2012 Female</td>
<td>2</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>2</td>
<td>1</td>
<td>1</td>
<td>1</td>
</tr>
<tr>
<td>2012 Male</td>
<td>2</td>
<td>1</td>
<td></td>
<td></td>
<td></td>
<td>1</td>
<td>1</td>
<td>1</td>
<td>1</td>
</tr>
<tr>
<td>All years Female</td>
<td>33</td>
<td>7</td>
<td>1</td>
<td>1</td>
<td>20</td>
<td>20</td>
<td>16</td>
<td>24</td>
<td></td>
</tr>
<tr>
<td>All years Male</td>
<td>25</td>
<td>3</td>
<td></td>
<td></td>
<td>3</td>
<td>17</td>
<td>16</td>
<td>14</td>
<td>19</td>
</tr>
</tbody>
</table>

*a Dispersal was not assessed if the animals died before <18 months old, when they were missing and not recaptured, or if they were captured after mid-January (<10 months old).

*b Animals for which home ranges allowed identification of likely natal areas (juvenile home ranges), as well as post dispersal home ranges as subadults or adults.

*c Animals for which maternal assignments were made using DNA analyses; natal areas were based on maternal home ranges.

*d Animals for which dispersal could be assessed using both field data (juvenile home ranges) and maternal assignments from genetic analyses.
Table D22: Estimates of mean Euclidean distances moved by dispersing fishers ≤18 months old on the SNAMP Fisher study. Dispersal was estimated by (1) distance between centroids for juvenile home ranges and subadult or adult home ranges, (2) distance between centroids for maternal home ranges (based on genetic-based maternity assignments) and adult or last known home ranges, or (3) distance between either juvenile home range centroids (fishers without maternity assignments) or maternal home range centroids and adult or last known home ranges.

<table>
<thead>
<tr>
<th>Dispersal, Sex</th>
<th>n</th>
<th>Mean distance (SE)</th>
<th>Range</th>
<th>t-test contrasts$^a$</th>
</tr>
</thead>
<tbody>
<tr>
<td>1. Juvenile to adult home range (field data)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>20</td>
<td>4.89 (1.36)</td>
<td>0.24-22.26</td>
<td>$t_{35}=1.35, P = 0.19$</td>
</tr>
<tr>
<td>Male</td>
<td>17</td>
<td>8.48 (2.39)</td>
<td>0.94-36.17</td>
<td></td>
</tr>
<tr>
<td>2. Maternal to adult home range (genetics)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>20</td>
<td>5.00 (1.21)</td>
<td>0.46-24.53</td>
<td>$t_{34}=1.32, P = 0.20$</td>
</tr>
<tr>
<td>Male</td>
<td>16</td>
<td>7.44 (1.41)</td>
<td>1.82-21.20</td>
<td></td>
</tr>
<tr>
<td>3. Juvenile or Maternal to adult home range (combined field and genetics)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>24</td>
<td>5.76 (1.26)</td>
<td>0.52-24.53</td>
<td>$t_{41}=1.67, P = 0.10$</td>
</tr>
<tr>
<td>Male</td>
<td>19</td>
<td>9.81 (2.22)</td>
<td>0.94-36.17</td>
<td></td>
</tr>
</tbody>
</table>

$^a$ Unequal variance t-tests.

Table D23: Mean Least Cost Movement paths (LCP) developed to evaluate dispersal by fishers ≤ 18 months old in the SNAMP Fisher Study area. LCP tracks were estimated for (1) dispersal between centroids for juvenile home ranges and subadult or adult home ranges, (2) for dispersal between centroids for maternal home ranges (based on genetic-based maternity assignments) and adult or last known home ranges, and for (3) dispersal between either juvenile home range centroids (fishers without maternity assignments) or maternal home range centroids and adult or last known home ranges.

<table>
<thead>
<tr>
<th>Dispersal, Sex</th>
<th>N</th>
<th>Mean Least Cost path (SE)</th>
<th>Range</th>
<th>t-test contrasts$^a$</th>
</tr>
</thead>
<tbody>
<tr>
<td>1. Juvenile to adult home range</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>20</td>
<td>7.53 (2.39)</td>
<td>0.47-44.09</td>
<td>$t_{35}=0.90, P = 0.38$</td>
</tr>
<tr>
<td>Male</td>
<td>17</td>
<td>11.63 (4.11)</td>
<td>1.03-69.82</td>
<td></td>
</tr>
<tr>
<td>2. Maternal to Adult home range</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>20</td>
<td>6.95 (1.62)</td>
<td>0.47-34.06</td>
<td>$t_{34}=1.07, P = 0.29$</td>
</tr>
<tr>
<td>Male</td>
<td>16</td>
<td>9.52 (1.77)</td>
<td>1.85-26.15</td>
<td></td>
</tr>
<tr>
<td>3. Juvenile or Maternal to Adult home range</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Female</td>
<td>24</td>
<td>8.76 (2.11)</td>
<td>0.47-44.09</td>
<td>$t_{41}=1.16, P = 0.25$</td>
</tr>
<tr>
<td>Male</td>
<td>19</td>
<td>13.48 (3.71)</td>
<td>1.03-69.82</td>
<td></td>
</tr>
</tbody>
</table>

$^a$ Unequal variance t-tests.
Figure D16: Plot of Euclidean distance dispersal movements for juvenile and young subadult female and male fishers within the SNAMP Fisher Project study area. Note: plot excludes the dispersal track for fisher M47 (KRFP fisher that dispersed north from south of Shaver Lake (High Sierra District) to near “Central Camp” in the Bass Lake District.
Figure D17: Plot of the potential dispersal tracks for KRFP Fisher M38 from his last live-trap position in February 2012 to his post-dispersal home range centroid near Central Camp within the SNAMP Fisher Study area 13 months later. The plot includes Euclidian distance as well as the estimated Least Cost Movement path, which we consider more realistic given the very steep and vertical cliffs typical of the San Joaquin River canyon east of Redinger Lake.
Figure D18: Estimated Least Cost Movement paths for young fishers (≤ 18 months) that were assessed for dispersal in the SNAMP Fisher study area from 2008 to 2013. Least cost movement path were developed as a more realistic way to assess fisher movements given that a number of landscape and habitat features are known avoided or restrictive to fishers as part of their overall natural history.
Young female fishers appeared somewhat more philopatric than male fishers, based on the proportion that moved less than the mean diameter of the annual home range for adult females in the study population (Fig. D19). The pattern was not significantly different however (Pearson $\chi^2 = 1.12$, $P = 0.29$). Also, there was no statistical evidence that male fishers dispersed farther than female fishers when dispersal distances were scored based on two levels of philopatry and two levels of dispersal (Euclidean distance Likelihood ratio $\chi^2 = 3.89$, $P = 0.27$; Fig. D20). The same analysis using LCP distances visualized in Fig. D18 was also nonsignificant (LCP Likelihood ratio $\chi^2 = 1.87$, $P = 0.60$; Fig. D18). However, it was noteworthy from a genetic perspective that 67% of females were philopatric, compared to about 45% of young males (Fig. D20; Table D24).

Figure D19: Proportion of female and male fishers that dispersed less than the diameter of the mean annual home range for adult female fishers (22.99 km; diameter = 5.401 km) in the SNAMP Fisher study area. Fishers that dispersed <5.4 km were considered as exhibiting philopatry, whereas those that moved >5.4 km were considered dispersers.

Figure D20: Plot of four different categories of dispersal distances for male and female fishers. Categories are based on dispersal distances of less than 0.5, 1.0, 1.5 and >2 times the mean diameter of the annual adult female home range in the study area. Illustration D12 (right): female fisher on the move.
Information on timing of dispersal is important for understanding whether juveniles captured in fall and winter were resident (born near the area of capture and initial locations), or if they originated elsewhere. Five dispersal events (20.8%) were initiated by juvenile fishers during fall to mid-winter (Table D24). Fourteen (58.3%) were initiated during the late winter to mid-spring time frame, and five started in late spring or summer (Table D24). Thus, nearly 80% of natal dispersal events occurred after February 5 when fishers were 11-13 months old.

### Table D24: Information on periods of the year when juvenile fishers initiated transitional movements as part of natal dispersal, and numbers of young fishers (<18 months old) that were philopatric, or that dispersed more than 1 diameter of the mean adult female home range (22.99 km; diameter = 5.41).

<table>
<thead>
<tr>
<th>Dispersal parameter</th>
<th>Female</th>
<th>Male</th>
<th>Total</th>
</tr>
</thead>
<tbody>
<tr>
<td>Timing of dispersal initiation</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Fall to mid-winter (Oct 15 - Feb 4)</td>
<td>2</td>
<td>3</td>
<td>5</td>
</tr>
<tr>
<td>Late winter to mid-spring (Feb 5 - May 5)</td>
<td>7</td>
<td>7</td>
<td>14</td>
</tr>
<tr>
<td>Late spring or summer (May 6 - Sep 20)</td>
<td>2</td>
<td>3</td>
<td>5</td>
</tr>
<tr>
<td>Dispersal distance</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Short distance philopatric (&lt; 2.7 km)</td>
<td>10</td>
<td>3</td>
<td>13</td>
</tr>
<tr>
<td>Philopatric (2.7 km to 5.4 km)</td>
<td>6</td>
<td>6</td>
<td>12</td>
</tr>
<tr>
<td>Medium distance dispersal (5.4-10.8 km)</td>
<td>5</td>
<td>6</td>
<td>11</td>
</tr>
<tr>
<td>Long distance dispersal (&gt;10.8 km)</td>
<td>3</td>
<td>5</td>
<td>8</td>
</tr>
</tbody>
</table>

a Data on initiation of dispersal were for a smaller subset of juveniles (n = 22) that made transitional movement that were apparent based on aerial telemetry locations and home range models
b <0.5X diameter of mean adult female home range
c 0.5-1X diameter of mean adult female home range
d 1-2X diameter of mean adult female home range
e >2X diameter of mean adult female home range

**Home Range Dynamics**

We obtained and processed ≈ 35,365 location records from all sources (Table D3; Fig. D21) from October 2007 to December 2013. The location dataset was screened for errors and duplicates (same day, same animal, <8 hrs apart in time), after which approx. 32,370 of the locations were retained for detailed analyses of movements (home ranges) for 109 different fishers. Most of the location records were from aerial radiotelemetry (88%), which were less accurate than other types of locations in the database (Table D3).

Annual 95% fixed kernel home range areas differed by sex for all age classes (Fig. D22), with mean values ranging from 20.98 km² for juvenile females to 86.18 km² for adult males (Table D25).
Male fishers are larger in body mass and morphological size than females (Powell 1993), and size dimorphism was already evident between sexes when juvenile fishers were captured and measured in October and November (7-8 months old) (Table D30). Body size is closely related to home range size in mammals (Swihart et al. 1988), which helps explain the larger size of annual home ranges for all age classes of male fishers in this study (Table D25, Figure D17).

Although fishers have previously
been described as exhibiting intrasexual territoriality (Powell et al. 2003), we noted considerable overlap between the annual home ranges of adults of the same sex (Fig. D23). Annual home ranges overlapped extensively among neighboring females, but overlap declined at the 70 or 60 percent fixed-kernel isopleths. These results suggest that female fishers maintain exclusive intra-sexual territories in their core use areas. Adult males move widely during the breeding season, resulting in widely overlapping use areas during spring (Popescu et al. 2014).

Figure D23: Annual home ranges for female and male fishers. The plot illustrates space use behavior where (1) the larger home ranges of males overlap home ranges of all females in the population, and (2) high overlap in space use among resident females at the 95% home range isopleth.

Home range sizes for fishers varied seasonally (Table D26; Fig. D24). Adult female home ranges were smallest during the spring, and reproducing females have smaller home ranges than non-
reproducing females during this time when mothers are constrained to the den area and provisioning kits at den structures. Home ranges of denning females were smaller than non-reproductive female home ranges through the summer, before offspring become independent. Size of seasonal home ranges among adult male fishers was smallest during the summer and largest during the spring, reflecting wide movement associated with mating during March and April (Table D26). In contrast, seasonal home ranges of subadult males (likely non-reproductive) were largest during winter and relatively stable during spring, summer, and fall (Table D26). Excluding the spring season home range for adult males, home range size was largest for all age and sex classes of fishers during winter, likely due to scarcity of prey.

<table>
<thead>
<tr>
<th>Table D25: Mean annual and core use home range sizes (km² ± SE) for radio-tracked fishers at the SNAMP site, December 2007 to March 2013</th>
</tr>
</thead>
<tbody>
<tr>
<td>Age/Sex</td>
</tr>
<tr>
<td>---</td>
</tr>
<tr>
<td>Juvenile (&lt;12 months)</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
<tr>
<td>Subadult (12 to 23 months)</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
<tr>
<td>Adult (≥24 months)</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>Table 26: Mean home range sizes (95% Fixed Kernel; km² ± SE) for fishers during four seasons of the year. Data for animals radio-collared on the Sierra National Forest, CA from December 2007 to March 2013.</th>
</tr>
</thead>
<tbody>
<tr>
<td>Age</td>
</tr>
<tr>
<td>Juvenile</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
<tr>
<td>Subadult</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
<tr>
<td>Adult</td>
</tr>
<tr>
<td>Female</td>
</tr>
<tr>
<td>Male</td>
</tr>
</tbody>
</table>

| Seasons were Spring: 21 Mar to 20 Jun; Summer: 21 Jun to 20 Sep; Fall 21: 21 Sep to 20 Dec; Winter: 21 Dec to 20 Mar. |
| Excludes home ranges for fishers that exhibited movements associated with dispersal |
| Includes home ranges for adult females that denned during the spring period of each year and excludes non-denning adults. |
**Figure D24:** Plot illustrating size of the mean seasonal home range size (SE bars) for female (a) and male (b) fishers from the SNAMP Fisher Project. NOTE: the scale is different for the two plots, which helps to illustrate similarities in habitat use patterns for the different age and sex groups. More details are provided in Table 26. (Illustration D14: adult female fisher departing a black oak den tree in spring 2011).

**SNAMP Fisher Management Indicators**

Management indicator 1 (occupancy/presence of fisher detections in 1-km² grid cells within the Key Watersheds) ranged from a low of 53% in 2012-13 to a high of 76% in 2011-12 (Table D27). The index of fisher activity developed for Management Indicator 1 indicated that the estimated detection rate (detections/100 camera survey days) was highest in 2012-13 and lowest in 2010-11. It was unusual that the detection rate was highest in the same year that naïve occupancy was lowest (Table D27). Camera survey year 2012-13 was atypical in that many grid cells in the Key Watershed were surveyed during summer when detection rates are significantly lower (Popescu et al. 2014). It was therefore possible that the low occupancy for 2012-13 compared to most other years was related to timing of surveys.

Spatially, the distribution of fisher active grid cells changed among years (Fig. D25). Visually, there was the appearance that fisher detections were somewhat reduced in the Cedar Valley Project region of the Key Watersheds (center-south; Figs. D4, D29) immediately after project implementation.
There were also changes in fisher detections in the northeast region of the Key Watersheds, which may have been associated with mastication and other activities associated with the Fish Camp Project (Figs. D4, D29). Visual comparisons of presence/absence are not appropriate for detecting patterns or trend in occupancy (persistence, extinction, recolonization) related to forest management projects, however. Detailed, multi-year occupancy modeling analyses are underway, which include the proportion of each grid cell treated in each of six years by different forest management activities. Models also include other covariates potentially important for understanding detection histories and habitat use (e.g., season, elevation).

<table>
<thead>
<tr>
<th>Camera survey year</th>
<th>Grid cells surveyed</th>
<th>Grid cells with fisher detections</th>
<th>Naïve occupancy</th>
<th>Fisher detections per 100 survey days</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08</td>
<td>122</td>
<td>71</td>
<td>0.582</td>
<td>11.4</td>
</tr>
<tr>
<td>2008-09</td>
<td>129</td>
<td>75</td>
<td>0.581</td>
<td>13.2</td>
</tr>
<tr>
<td>2009-10</td>
<td>127</td>
<td>75</td>
<td>0.591</td>
<td>15.2</td>
</tr>
<tr>
<td>2010-11</td>
<td>125</td>
<td>82</td>
<td>0.656</td>
<td>10.5</td>
</tr>
<tr>
<td>2011-12</td>
<td>128</td>
<td>98</td>
<td>0.766</td>
<td>14.2</td>
</tr>
<tr>
<td>2012-13</td>
<td>133</td>
<td>70</td>
<td>0.526</td>
<td>18.6</td>
</tr>
</tbody>
</table>

a Camera surveys were completed in each of six camera survey years (≈ Oct 15 to Oct 14) using a standard protocol.

b Number grid cells with fisher detections divided by the total number of grid cells surveyed; occupancy rate is not corrected for a survey-specific probability of detection < 1.0.

c Estimated as the number of functional camera survey days with fisher detections, but excluded camera days for grid cells with no fisher detections.
Figure D25: GIS plots illustrating change in patterns of occupancy for Management Indicator 1 for six camera survey years. Ratios indicate the number of grid cells with detections versus total grid cells surveyed. (Table D27 for details).
As an extension to Management Indicator 1, we also created an overall index of fisher activity for each grid cell based on mean number days in each camera survey year with fisher detections and the proportion of survey years with fisher detections (Fig. D26). The index illustrates that fisher activity was consistently high in the center and northwest region of the Key Watersheds and lowest from Cedar Valley southward (Fig. D26).

Management Indicator 2 identified an average of 5.0 subadult or adult females and 2.0 subadult or adult males using the Key Watershed focal study area across all years, assuming all animals were identified and collared (Table D28). For both sexes combined, the number of resident fishers using the focal study area ranged from 6.2 to 7.7, and the variation among years was small (Table D28, Fig. D27).

**Table D28:** Management indicator for the number of resident subadult and adult fishers using the Key Watershed focal study area for their various home range activities during Sep 1 to Mar 15 of each year.

<table>
<thead>
<tr>
<th>Year</th>
<th>Females</th>
<th>Males</th>
<th>Both sexes</th>
</tr>
</thead>
<tbody>
<tr>
<td>2007-08&lt;sup&gt;b&lt;/sup&gt;</td>
<td>5.6</td>
<td>2.1</td>
<td>7.7</td>
</tr>
<tr>
<td>2008-09</td>
<td>6.1</td>
<td>1.4</td>
<td>7.5</td>
</tr>
<tr>
<td>2010-11</td>
<td>4.1</td>
<td>2.1</td>
<td>6.2</td>
</tr>
<tr>
<td>2011-12</td>
<td>4.0</td>
<td>2.9</td>
<td>6.9</td>
</tr>
<tr>
<td>2012-13</td>
<td>5.0</td>
<td>1.7</td>
<td>6.7</td>
</tr>
</tbody>
</table>

<sup>a</sup> Numbers are based on the sum of the proportion of each individual fishers’ 95% fixed kernel home range included within the Key Watershed region.

<sup>b</sup> Because of the limited number of fishers radiocollared during the first project year (n = 7, before March 31, 2008) it was not informative to calculate this Management Indicator in that year.

**Figure D26:** Index of fisher activity from repeat camera surveys completed in the Key Watersheds focal study area. The Index was calculated as the mean no. of days with fisher activity for years that the grid cell was surveyed/1 + proportion of surveyed years with fishers detections in the grid cell.
Population Year 2: 5.6 resident females
Population Year 3: 6.1 resident females
Population Year 4: 4.1 resident females
Population Year 5: 4.0 resident females
Population Year 6: 5.0 resident females

Figure D27: Estimated number of subadult and adult female fishers (resident females) with home ranges including portions of the Key Watersheds focal study area. Polygons are individual 95% fixed kernel home ranges based on location records during the Sep 1 to Mar 15 period during each population year (Apr 1 to Mar 31).
The original Management Indicator 3 was recast to estimate survival for adult female fishers for a sequence of 2-year groups of demographic data (data combined for Kaplan-Meier models of survival). For the first 2-year group, we included data for the small number of fishers \((n=7)\) that were captured and radiocollared from mid December 2007 to March 31, 2008. We further summarized data on Juvenile and subadult female survival, and calculated point estimates of weaning reproduction and weaning litter size for each of the five 2-year groups (Table D18). Expanded Management Indicator 3 identified that adult female survival ranged from a low of 0.69 in Year group 3 to a high of 0.86 in Year group 4 (Table D29). Corresponding data on survival for juvenile and subadult females and data on reproduction identified that relatively low levels of survival and reproduction suggested the population was in decline \((\lambda < 1.0)\) between 2008 and 2010, stable between 2010 and 2012, and increasing by 3-4%/year during 2012 to 2014 (Table D29). However the fact that 95% CI for \(\lambda\) overlapped 1.0 in all years indicates that these values should be interpreted carefully.

**Table D29**: Expanded Management Indicator 3 for adult female survival, including Leslie Matrix population growth rates for two year running average starting in 2008 and ending in spring 2014. Population years start April 1 and end March 31. Numbers in parentheses for survival are the 95% CIs based on Kaplan-Meier staggered entry survival analyses for the group of years identified.

<table>
<thead>
<tr>
<th>Year group, Demographic rate</th>
<th>Juvenile</th>
<th>Subadult</th>
<th>Adult</th>
<th>(\lambda^a)</th>
</tr>
</thead>
<tbody>
<tr>
<td>1. 2007-08, 2008-09, 2009-10</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Survival, (s(t))</td>
<td>0.76 (0.53-0.99)</td>
<td>0.47 (0.28-0.67)</td>
<td>0.81 (0.66-0.96)</td>
<td>0.87 (0.65-1.08)</td>
</tr>
<tr>
<td>2. 2009-10, 2010-11</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Survival, (s(t))</td>
<td>0.8 (0.59-1.01)</td>
<td>0.67 (0.42-0.92)</td>
<td>0.70 (0.54-0.86)</td>
<td>0.88 (0.63-1.12)</td>
</tr>
<tr>
<td>3. 2010-11, 2011-12</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Survival, (s(t))</td>
<td>0.74 (0.54-0.94)</td>
<td>0.89 (0.71-1.07)</td>
<td>0.69 (0.53-0.86)</td>
<td>1.00 (0.77-1.22)</td>
</tr>
<tr>
<td>4. 2011-12, 2012-13</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Survival, (s(t))</td>
<td>0.80 (0.55-1.05)</td>
<td>0.73 (0.52-0.94)</td>
<td>0.86 (0.71-1.00)</td>
<td>1.04 (0.81-1.26)</td>
</tr>
<tr>
<td>5. 2012-13, 2013-14</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Survival, (s(t))</td>
<td>1.0</td>
<td>0.73 (0.48-0.98)</td>
<td>0.74 (0.56-0.93)</td>
<td>1.03 (0.77-1.22)</td>
</tr>
</tbody>
</table>

\(^a\) Population growth rate was estimated using the demographic parameters developed for each Year group in a Leslie-Matrix model formulation described previously. The range in values for \(\lambda\) was based on the 95% CIs for survival for each age class when producing fertility \((F_i)\) rates using the equation \(F_i = b_iP_i\), where \(b(i)\) was fecundity, and \(P_i\) was the age-specific survival rate (see Table D17).

\(^b\) Year group 1 includes information for a small number fishers monitored for survival from late December to March 2008. All other year groups include two population years of data.
**Fisher response to fuel management**

Management disturbances and wildfire

Our analyses of FACTS and other extractive and restorative management activities revealed that the estimated area of forest disturbing activities that occurred in the study area was highest for restorative fuel reduction, moderate for logging, and lowest for managed burning and natural or human caused wildfires (Table D30). We estimated that there was an annual average 1.9% (SD 0.70) of the study area treated for restorative fuel reduction each year from 2002-03 to 2012-13, and 20.6% of the study area was disturbed by these activities in all 11 years. We estimated that there was an annual average of 1.1% (SD 0.70) of the study area with extractive fuel reduction each year from 2002-03 to 2012-13, and an estimated 12.1% of the study area was disturbed by logging in all 11 years. We estimated that there was an annual average of 0.25% (SD 0.28) of the study area with managed burning each year from 2002-03 to 2012-13, and an estimated 2.8% of the study area was disturbed by managed burns in all 11 years. Also, the combined area disturbed by all 3 management activities averaged 36.3 km²/year from 2002-03 to 2012-13, which represented an annual disturbance of 3.2%/year from SPLATS in the overall study area. Our fire variables included managed burns+forest fires, and we estimated that the annual average portion of the study area with managed burns+wildfires was 0.56%/year (SD, 0.83) from 2002-03 to 2012-13, and 6.2% of the overall study area was exposed to those disturbances in the 11 years. Also, in the 44 years from 1957 to 2001, we estimated that 130.2 km² (11.6%) of the overall study area was burned by wildfires.

**Multi-season occupancy**

The mean detection probability for fishers per 8-10 day survey period in the 361 multi-season survey grid cells was 0.31 (95% CI: 0.28, 0.37). Naïve initial occupancy among the multi-season grid cells was 0.66, whereas our modeled estimate for initial occupancy averaged across survey sites was 0.75 (95% CI: 0.59, 0.87). Mean annual persistence (1-extinction) was 0.87 (95% CI: 0.82, 0.91), whereas the annual colonization rate was 0.34 (95% CI 0.28, 0.42).

Our multi-season occupancy modeling identified a single best model for local colonization that included the intercept only (Table D31). Covariates hazfuels.5, log.5, and burn.1.50 were included in 3 lower ranking colonization models with support, but the relative importance for...
each individual variable was \leq 0.35. We therefore fit an intercept-only colonization component in our subsequent evaluation of extinction covariates.

Table D30: Estimates of the areas (km\(^2\)) disturbed by logging activities, fuel reduction treatments, and managed burns in the Bass Lake District, Sierra National Forest, and southwestern Yosemite National Park in 11 camera survey years (Oct 15 to Oct 14) from 2002 to 2013 as well as wildfire activity in 5-year periods from 1957 through 2001.

<table>
<thead>
<tr>
<th>5 yr period or survey year</th>
<th>Restorative fuel reduction</th>
<th>Extractive fuel reduction</th>
<th>Managed burns + forest fire</th>
</tr>
</thead>
<tbody>
<tr>
<td>Area</td>
<td>Study area (%)</td>
<td>Area</td>
<td>Study area (%)</td>
</tr>
<tr>
<td>1957 to 1961</td>
<td>36.40</td>
<td>7.28</td>
<td></td>
</tr>
<tr>
<td>1962 to 1966</td>
<td>5.30</td>
<td>1.06</td>
<td></td>
</tr>
<tr>
<td>1967 to 1971</td>
<td>6.05</td>
<td>1.21</td>
<td></td>
</tr>
<tr>
<td>1972 to 1976</td>
<td>3.43</td>
<td>0.69</td>
<td></td>
</tr>
<tr>
<td>1977 to 1981</td>
<td>4.65</td>
<td>0.93</td>
<td></td>
</tr>
<tr>
<td>1982 to 1986</td>
<td>11.46</td>
<td>2.29</td>
<td></td>
</tr>
<tr>
<td>1987 to 1991</td>
<td>41.91</td>
<td>8.38</td>
<td></td>
</tr>
<tr>
<td>1992 to 1996</td>
<td>0.99</td>
<td>0.20</td>
<td></td>
</tr>
<tr>
<td>1997 to 2001</td>
<td>20.05</td>
<td>4.01</td>
<td></td>
</tr>
<tr>
<td>2002-03</td>
<td>23.7</td>
<td>2.10</td>
<td>11.6</td>
</tr>
<tr>
<td>2003-04</td>
<td>13.7</td>
<td>1.21</td>
<td>2.9</td>
</tr>
<tr>
<td>2004-05</td>
<td>13</td>
<td>1.15</td>
<td>7</td>
</tr>
<tr>
<td>2005-06</td>
<td>26</td>
<td>2.31</td>
<td>6.8</td>
</tr>
<tr>
<td>2006-07</td>
<td>34.5</td>
<td>3.06</td>
<td>13.1</td>
</tr>
<tr>
<td>2007-08</td>
<td>15.8</td>
<td>1.40</td>
<td>2.1</td>
</tr>
<tr>
<td>2008-09</td>
<td>29.1</td>
<td>2.58</td>
<td>11.4</td>
</tr>
<tr>
<td>2009-10</td>
<td>27.4</td>
<td>2.43</td>
<td>27</td>
</tr>
<tr>
<td>2010-11</td>
<td>12.4</td>
<td>1.10</td>
<td>13.8</td>
</tr>
<tr>
<td>2011-12</td>
<td>12.6</td>
<td>1.12</td>
<td>24.3</td>
</tr>
<tr>
<td>2012-13</td>
<td>23.9</td>
<td>2.12</td>
<td>16.1</td>
</tr>
<tr>
<td><strong>Total area</strong></td>
<td><strong>232.1</strong></td>
<td><strong>136.1</strong></td>
<td><strong>69.87</strong></td>
</tr>
</tbody>
</table>

Our multi-season models evaluating local extinction identified a single top model including covariate hazfuels.5 only (hazfuel.5 relative importance = 0.98) (Table D31). There were 2 models with support that included the covariates log.5 and burn.1.50, but the individual relative importance metrics for both were low. We found that fisher persistence (1 - extinction)
was negatively associated with *hazfuels.5*; probability of persistence decreased by 27% as the proportion of the grid cell treated for cumulative restorative fuel reduction increased from 0 (occupancy = 0.89, 95%CI 0.85, 0.92) to 1.0 (occupancy = 0.65, 95%CI 0.46, 0.81).

**Table D31:** Candidate models for multi-season occupancy evaluations of colonization and local patch extinction for camera surveys for fishers in the Bass Lake District, and southwestern Yosemite National Park, California from Oct 2007 to Oct 2014.

<table>
<thead>
<tr>
<th>Model, covariate</th>
<th>AIC</th>
<th>ΔAIC</th>
<th>AIC&lt;sub&gt;wt&lt;/sub&gt;</th>
<th>Cumulative AIC&lt;sub&gt;wt&lt;/sub&gt;</th>
<th>Covariate importance</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Colonization</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>intercept only</td>
<td>4211.96</td>
<td>0.00</td>
<td>0.34</td>
<td>0.34</td>
<td></td>
</tr>
<tr>
<td><em>hazfuels.5</em></td>
<td>4213.15</td>
<td>1.19</td>
<td>0.19</td>
<td>0.53</td>
<td>0.35</td>
</tr>
<tr>
<td>log.5</td>
<td>4213.87</td>
<td>1.91</td>
<td>0.13</td>
<td>0.66</td>
<td>0.27</td>
</tr>
<tr>
<td>burn.1.50</td>
<td>4213.95</td>
<td>2.00</td>
<td>0.13</td>
<td>0.79</td>
<td>0.27</td>
</tr>
<tr>
<td><em>hazfuels.5</em> + log.5</td>
<td>4215.14</td>
<td>3.19</td>
<td>0.07</td>
<td>0.86</td>
<td>0.27</td>
</tr>
<tr>
<td><em>hazfuels.5</em> + burn.1.50</td>
<td>4215.15</td>
<td>3.19</td>
<td>0.07</td>
<td>0.93</td>
<td></td>
</tr>
<tr>
<td>burn.1.50 + log.5</td>
<td>4215.87</td>
<td>3.91</td>
<td>0.05</td>
<td>0.97</td>
<td></td>
</tr>
<tr>
<td><em>hazfuels.5</em> + burn.1.50 + log.5</td>
<td>4217.14</td>
<td>5.19</td>
<td>0.03</td>
<td>1.00</td>
<td></td>
</tr>
<tr>
<td><strong>Extinction</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>hazfuels.5</em></td>
<td>4205.26</td>
<td>0.00</td>
<td>0.50</td>
<td>0.50</td>
<td>0.98</td>
</tr>
<tr>
<td><em>hazfuels.5</em> + log5</td>
<td>4207.08</td>
<td>1.82</td>
<td>0.20</td>
<td>0.70</td>
<td></td>
</tr>
<tr>
<td><em>hazfuels.5</em> + burn.1.50</td>
<td>4207.11</td>
<td>1.85</td>
<td>0.20</td>
<td>0.90</td>
<td></td>
</tr>
<tr>
<td><em>hazfuels.5</em> + burn.1.50 + log.5</td>
<td>4208.96</td>
<td>3.70</td>
<td>0.08</td>
<td>0.98</td>
<td></td>
</tr>
<tr>
<td>log.5</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>intercept only</td>
<td>4212.67</td>
<td>7.42</td>
<td>0.01</td>
<td>0.99</td>
<td></td>
</tr>
<tr>
<td>log.5</td>
<td>4214.16</td>
<td>8.90</td>
<td>0.01</td>
<td>0.99</td>
<td>0.29</td>
</tr>
<tr>
<td>burn.1.50</td>
<td>4214.61</td>
<td>9.36</td>
<td>0.00</td>
<td>1.00</td>
<td>0.28</td>
</tr>
<tr>
<td>burn.1.50 + log.5</td>
<td>4216.05</td>
<td>10.80</td>
<td>0.00</td>
<td>1.00</td>
<td></td>
</tr>
</tbody>
</table>
Integration

Fire modeling

Fuels treatments reduced the intensity of the simulated fire, as evidenced by the predicted flame lengths (Fig. D28). On the untreated landscape, 68.6%, 18.4%, 11.2%, and 1.8% of the study area experienced flame lengths of <2, 2-4, 4-8, and >8 m, respectively. In comparison, on the treated landscape, 75.1%, 16.4%, 7.5%, and 1.0% of the study area burned at these flame lengths. Collins et al. (2011) noted that flame lengths >2 m often corresponded to areas with crown fire initiation (i.e., torching). Thus, a greater proportion of the untreated landscape was exposed to potential crown fire (31.4%) than for the untreated landscape (24.9%).

Figure D28: Flame lengths (m) of simulated fires at Sugar Pine on (a) untreated and (b) treated landscapes. We show the location of treatment polygons in (a) for ease of comparison, but treatments were not implemented in (a).

Assessing the effects of fire and SPLATs on fisher habitat

We found that SPLATs caused an immediate, slight reduction in potential fisher habitat. The entire area of the four watersheds was 35,103 ac (14,206 ha), and in year 0, there were 16,013 ac (6,480 ha) of potential fisher habitat on the untreated landscape compared to 13,938 ac
(5,641 ha) on the treated landscape (Fig. D29). In the absence of simulated fire, the amount of habitat steadily increased over time and was actually slightly greater on the treated landscape in years 10 and 30 (Fig. D2). When fire was simulated, SPLATs had a slight, positive effect on the amount of potential fisher habitat up to 30 years later. In year 30, there were 14,653 ac (5,930 ha) of potential fisher habitat on the untreated landscape compared to 15,254 ac (6,173 ha) on the treated landscape (Fig. D2).

**Figure D29:** The amount of potential fisher habitat on the 35,103-ac Sugar Pine study area under four scenarios: 1) no SPLATs and no wildfire; 2) SPLATs and no wildfire; 3) no SPLATs and wildfire; and 4) SPLATs and wildfire. Year 0 for the “no treatment” scenarios was 2008, and year 0 for the “treatment” scenarios was 2013 (i.e., after SPLATs were implemented). Simulated fires occurred in year 0 for both the “no treatment” and “treatment” scenarios, and post-fire effects were first assessed in year 10.
Discussion

Reproduction and Basic Demography

Empirical data on reproductive rates and litter sizes are important for understanding the ability of a population to withstand challenges to survival, and to produce realistic estimates of population size in landscape level population models being developed for conservation planning (Lofroth et al. 2010, Spencer et al. 2011). The basic life history of fishers with regards reproduction is generally well known. Fishers have a long gestation period due to the reproductive strategy of delayed implantation (Powell 1993). Once the blastocyst implants in the uterine wall 10-11 months after fertilization of the egg, embryonic development resumes and ≈ 36 days later 1-4 kits are typically born.

Parturition for fishers in northern California typically occurs in mid to late March (Matthews et al. 2013a), and female fishers in the SNAMP Fisher population were no exception based on initiation of denning around March 22-31 (Table D7). Also, duration of denning for fishers in our study (≈ 70-75 days; Table D7), and cessation of localization to den trees in early to mid-June was typical of elsewhere in the western United States (Matthews et al 2013a, Aubry and Raley 2006). The mean denning rate for female fishers in the SNAMP Fisher study was 0.85 (Table D9), which was slightly lower than for fishers in the Hoopa Fisher Study in northern California (0.88; Matthews et al. 2013a), similar to in the Kings River Fisher Project area (0.86; R. Green, unpublished), and higher than in southern Oregon (0.59; Aubry and Raley 2006). The average weaning rate from SNAMP females in our study area (0.74; Table D9) was higher compared to the Hoopa Fisher Study (0.65; Matthews et al. 2013a), and in southern Oregon (0.44; Aubry and Raley 2006). However, as Matthews et al. (2013a) noted, reports of low weaning rates from some studies may be due to the difficulty in differentiating between subadult and adult females at first capture. We closely tracked most animals in the population from when they were juveniles until death, and ages for nine female fishers that were captured early in the study were determined by cementum annuli (Mattson’s Laboratory, Milltown, MT; Poole et al. 1994).
Average litter sizes are larger for female fishers in eastern North America (2-4 kits/litter; Paragi et al. 1994, York 1996) compared to in the western United States where litter sizes are most commonly 1 or 2 kits (Aubry and Raley 2006, Matthews et al. 2013). The mean litter size from SNAMP Fisher (1.5 kits/litter, Table D10) was similar to reports from the Kings River Fisher Project (1.6; R. Green, unpublished), but lower than 1.8 kits/litter reported for the Sequoia National Forest, California (Truex et al 1988), 1.9 kits/litter from the Hoopa Fisher Study in northern California (Matthews et al. 2013a), or 1.8 kits/litter in southern Oregon (Aubry and Raley 2006).

Close monitoring of denning behavior by several current studies including SNAMP Fisher is providing insight on the risks female fishers encounter while attempting to reproduce. In the course of five denning seasons, we documented eight cases when females died or were killed with dependent kits in den cavities (Tables D15 & D16). Death of denning female fishers appears fairly common, based on reports from the Hoopa Fisher Study (n = 5; Matthews et al. 2013a), the USFS Kings River Study (n = 3; C. Thompson, unpublished data), the ongoing California Department of Fish and Wildlife, U.S. Fish and Wildlife, and Sierra Pacific Industries “Stirling Fisher Reintroduction Project” (n = 5; Powell et al. 2013), and the Olympic Fisher Reintroduction Project (n = 2; Lewis et al. 2012). Evidence that a significant number of females exhibiting denning behavior may die before weaning is important because weaning rates may be biased high unless estimates are based on complete monitoring through the duration of the denning period (Facka et al. 2013).

**Denning Structures and Denning Habitats**

Across their range in North America female fishers give birth to kits in cavities in live trees or snags (Paragi et al. 1996, York 1996, Weir et al. 2012, Zhao et al. 2012, Matthews et al. 2013a). Cavities provide protection from predators and inclement weather during the early spring to late spring when females are rearing their young (Weir et al. 2012). Most female fishers use more than one denning structure during a den season (range 1-6; Matthews et al. 2013a), and female fishers on the SNAMP Fisher study used an average of 2.4 different denning structures per den season (range 1-5), compared to 3.4 on the Kings River Fisher Study (R. Green,
unpublished data), and 3.1 in northwestern California (Matthews et al. 2013). Female fishers may use more than one denning structure in a season for several reasons: to accommodate kit growth by moving to larger cavities, to reduce predation risk, as bobcats and mountain lions may discover a den location due to odors from the accumulation of urine and feces, to move closer to unexploited foraging areas, and to avoid exposure to feces and parasites that may accumulate in den cavities.

The lower mean number of den trees used by female fishers on the SNAMP Fisher study area compared to on the KRFP or the Hoopa Fisher Study may be related to disturbance by researchers. Biologists on both the KRFP and Hoopa Fisher studies climb den structures of most known denning female fishers to obtain kit counts, and they also attempt to extract kits from den cavities to measure body size, collect tissue samples, and to insert PIT tags for later identification (Thompson et al. 2011, Matthews et al. 2013a,b). This process requires presence of multiple biologists at the den tree for periods of 60 to 180 minutes. Although we occasionally ascended den trees in the SNAMP study area to obtain kit counts (\(n = 9\) total den tree climbs during six denning seasons), most kit counts (\(\approx 90\%\)) were obtained using images from 2-4 motion-sensing cameras placed around denning structures to monitor and chronicle denning activities remotely. Moreover, we noticed that some individual female fishers were sensitive to presence of technicians setting up den cameras (the process requires 30 to 60 min), based on short duration use of their denning structures after the first visit. Our den camera protocol was adjusted to minimize time needed to setup and service den cameras by quickly switching out memory cards and reviewing images away from the denning structure. Also, whenever possible, we did not approach den trees to service cameras when radiotelemetry identified presence of the denning female.

On the nearby Kings River Fisher Project where habitats are similar to the SNAMP Fisher study area, denning structures used by female fishers were most commonly black oaks (54%: 50% live, 4% snags). Overall, 91% of denning structures used on the Kings River Fisher Project were live trees. When repeat use den trees were counted just once, 43% of the unique denning structures used in the SNAMP Fisher study area were black oak trees (Table D11; 25%
live, 18% snags), and the remaining unique den trees were primarily incense cedar (33%) or white fir (15%). Only 56% of the denning structures used by female fishers in the SNAMP study area were live trees.

Weir et al. (2012) noted that trees need to have two very specific features for female fishers to use them for denning; some form of physical damage to the tree bole to provide access for decay organisms, and the damage must be of particular dimensions to provide females access to the interior of the tree while excluding predators (Lofroth et al. 2010). McDonald (1990) noted that live black oaks are susceptible to internal decay and probably last longer on the landscape than conifer snags. However, 26% of 125 unique den structures used by fishers on the SNAMP study site were in conifer snags (Table D11), suggesting they are not especially uncommon on the landscape in our area. Also, our observations of incense cedars and white fir suggested these two tree types were susceptible to the types of damage identified by Weir et al. (2012), particularly with regard to fire scars for cedar trees. Many of the cedar trees selected for use as den trees had basal fire scars, and the actual den cavities in both cedar trees and white fir were commonly associated with large branch break points.

Habitat and site characteristics immediately surrounding the denning structures are likely important for appropriate thermal conditions, availability of prey, and avoidance of predators (escape cover and concealment cover). Denning structures used in the SNAMP Fisher study area were generally larger than available trees and snags; mean DBH was relatively large (larger for conifers than the hardwoods) and mean tree heights were taller for live conifers compared to conifer snags or oaks in general (Table D12, Fig. D11). Canopy cover was greater than 80% in the vicinity of many den trees (Table D13). Shrub cover near den trees was variable, as was aspect (Table D13, Fig. D12). Most den trees had multiple large down trees/logs nearby, and concealment cover to the base of den trees averaged more than 45%. Although detailed analyses of data from fixed radius habitat plots have not been completed, habitat characteristics were developed from high resolution Lidar data for many den trees within the Key Watersheds. As part of collaborative work with the Spatial Team, Zhao et al. (2012) identified that fishers selected den sites with tall trees and steep slopes within a 10-m radius of the den tree, high forest
structural complexity within 20 m, large tree clusters within 30 m, and high canopy cover and larger mature trees within 50 m. Finally, at the larger landscape scale, the mean elevation for denning structures used in the SNAMP Fisher study was 1,591 m (Fig. D11).

**Fisher Survival and Cause-specific Mortality**

The SNAMP Fisher study uncovered a wider diversity of causes of mortality for fishers in the region than anticipated (Table D16). In the first five years of the study many newly deceased fishers were recovered before the pilot and biologist in the airplane landed back at the Mariposa Airport, and almost always within six hours of the first indication that an animal’s radiocollar was pulsing inactive. Although the fixed-wing aerial telemetry effort was expensive, the SNAMP study identified the first known death for the species caused by active infection with Canine Distemper Virus (CDV). We also recovered the fresh carcass of a fisher in spring 2009 that was subsequently determined to have died from exposure to anticoagulant rodenticides. This discovery prompted testing of archived tissue samples of dead fishers throughout California and in the western United States, leading to two peer-reviewed papers focused on the problem of rodenticides and other poisons used at clandestine marijuana grow sites on California public lands (Gabriel et al. 2012, Thompson et al. 2013). Moreover, an important and very real benefit from the investment in an aviation program in support of SNAMP Fisher has been the discovery that survival and reproduction of fishers in the Sierra National Forest is challenged by multiple factors external to, and not directly linked to current forest management activities.

Well over half of the individual fishers captured and radiocollared during the study had perished as of April 2014. Known sources of mortality in the SNAMP Fisher study population included high numbers of attacks by predators (interspecific killing; Wengert et al. 2014), roadkill deaths on Highway 41, infection by canine distemper virus (CDV; Keller et al. 2012) and *Toxoplasma gondii*, injury-induced starvation or septicemia, entrapment in a water tank, and acute toxicosis and hemorrhaging caused by exposure to rodenticides (Gabriel et al. 2013, Thompson et al. 2013). Males made up the large majority of fishers that died of infectious disease, roadkill, and rodenticide exposure. On other hand, a greater proportion of females than males were killed by predators (Fig. D8), and work with collaborators from UC Davis indicated
that males were less susceptible to bobcat predation than females (Wengert et al. 2014).

The diverse threats to survival that impinge on population growth in our study population are not unique to the southern Sierra Nevada region. Fishers in the Hoopa Valley in northern California area also died as a result of predation (Wengert et al. 2014), disease (Gabriel 2013), and rodenticides and other toxicants (Gabriel et al. 2013). Also, fishers that were reintroduced in northern California as part of the Stirling Fisher Reintroduction Project site have succumbed to predation, disease, and trauma from collisions with vehicles (Powell et al. 2013).

Survival estimators generally assume that live-trapping and radiocollars do not influence survival of study animals. Based on necropsies and extensive pathological tests completed on carcass remains of 47 dead fishers, no mortalities on the SNAMP Fisher study site were directly attributable to capture-related injury or radiocollars (e.g., strangulation or infection from chafing on the neck). However, one adult female fisher failed to survive the capture process to recovery and release. We acknowledge that the stress of capture and anesthesia contributed to the death of this female, though detailed pathological examination revealed that she was extremely emaciated and suffering from systemic infection from serious injury (laceration to the rostrum, fractured mandible, partially disarticulated lower jaw) prior to capture, and was unlikely to survive regardless of capture and handling (Gabriel 2013).

Analyses of live/dead status of individual fishers from SNAMP Fisher indicated that survival rates for adult females were within the range observed for other areas in the western United States. Overall survival for adult female fishers was 0.74, compared to 0.77 at the KRFP site (Sweitzer et al., In revision), which was higher than the 0.61 rate reported for a smaller sample of radio-collared female fishers on the Sequoia National Forest south of our study region (Truex et al. 1998). Aubry and Raley (2006) reported an adult female survival rate of 0.78 from a study in southwestern Oregon, whereas Higley et al. (2012) estimated adult female survival at 0.77 to 0.79 in northwestern California based on two different analytical methods (Known-fate models and Capture-Mark-Recapture, respectively). Jordan et al. (2011) reported a combined
male-female adult survival rate of 0.94 for research completed in the KRFP study area in 2002-2004, however, their survival estimate was based on camera detections rather than radio-collared individuals, and the values reported were considered to have low precision related to tag loss and other factors.

In general, survival of male fishers on the SNAMP Fisher study area was consistently lower among all age and sex classes compared to females (Table D16). All year survival for SNAMP Fisher males ranged from 0.57 to 0.64, which was lower than adult male survival for fishers in southwest Oregon (0.85; Aubry and Raley 2006), in the Sequoia National Forest, California (0.73; Truex et al. 1996) and in northwest California (0.75 to 0.72; J. M. Higley unpublished report).

At the outset of the study we anticipated that survival would be lower for juvenile and subadult fishers compared to adults, as is typical for several species of mesocarnivores (Farias et al. 2005, Murdoch et al. 2010). Although survival among subadults trended lower than for adults, juvenile survival by both male and female fishers was often very similar or trended higher than adult survival (Table D16). We believe this is unlikely and an artifact of our inability to monitor juvenile survival during their first six months of life. Juvenile fishers are small in size and body mass during summer, and in our study and for most prior studies attempting to ascertain survival, juveniles were not fitted with radiocollars until fall or winter when many individuals within the cohort may have already perished (Facka et al. 2013). Even less is known about survival of kits when they are being provisioned inside den cavities (Lofroth et al. 2010). This is important because modeling efforts using empirically derived demographic parameters identify that population size and likelihood of persistence are relatively insensitive to juvenile survival (Buskirk et al. 2012, Spencer et al. 2011), potentially because juvenile survival is biased high.

The all year estimate of female survival on the SNAMP Fisher site was higher for juveniles, similar for subadults, and lower for adults compared to parameter values used by
Spencer et al. (2011) to simulate fisher population dynamics under different management scenarios for the southern Sierra Nevada region. Our combined year estimates of survival for juvenile females was 0.75 (value used by Spencer et al. = 0.50), 0.71 for subadult females (value used by Spencer et al. = 0.70), and 0.74 for adult females (value used by Spencer et al. = 0.90). Spencer et al. (2011) reported their model was relatively insensitive to juvenile and subadult survival (and other demographic parameters), but highly sensitive to adult female survival. Our empirically derived estimate for adult female survival (Table D17) was 15% lower than 0.90, which is important because Spencer et al. (2011) noted that a 5% decrease in female survival produced an approximate 18% reduction in the ending population size 40 years after model initiation. Similarly, 10% and 25% reductions in female survival resulted in 37% to 72% reductions in the ending population size. The significantly higher survival rate we estimated for juvenile females might ameliorate the reduced end year population size associated with 15% lower adult female survival. A new modeling effort is underway that will integrate new information on demographic rates from the SNAMP and KRFP study sites (Spencer et al. 2015).

Wildlife populations are exposed to a variety of mortality factors, which vary in importance towards limiting or impinging on population growth. Predation was clearly identified as the most important source of mortality on the SNAMP Fisher study area (Table D15, Fig. D8). Data on percent deviation in survival described by Sweitzer et al. (In revision) indicated that predation was more important than disease processes and human-linked factors for limiting fisher survival.

Disease in the form of canine distemper, toxoplasmosis, or pleururitus+pneumonia caused death of five radiocollared fishers during the SNAMP Fisher study, and an additional four fishers died of septicemia or starvation due to puncture wounds or other injury (Table D16; Gabriel 2013). The death of four fishers on our study by infection or starvation after suffering wounds or debilitating injury was not unusual or surprising for an animal as active as the fisher. Other long term studies of radio-collared fishers have reported similar circumstances (Aubry and Raley 2006, Weir and Corbould 2008).
Infectious disease has been a conservation concern for the two isolated populations of fishers in California since exposure to CDV and other pathogens was first documented in northern California in the early 2000s based on serological testing (Brown et al. 2008). Canine distemper is of special concern because of the abundance and widespread extent of mortalities among multiple species of rare and endangered carnivores has been reported (Timm et al. 2009, Williams et al. 1998, Woodroffe 1999). An outbreak, or localized epizootic of CDV that likely originated on the SNAMP site in spring 2009, and then spread south into KRFP during summer 2009 resulted in death of four fishers (Keller et al. 2012, Table D2). This disease-related mortality event confirmed that exposure by fishers to CDV and other agents of disease is of conservation concern for fishers in the western United States in general (Gabriel et al. 2012b), but particularly for the small, isolated population of fishers in the southern Sierra Nevada (Gabriel 2013). One fisher on the SNAMP study site was also confirmed to have died by complications after parasitic infection by *Toxoplasma gondii* (Gabriel 2013). Although exposure of fishers to *Toxoplasma gondii* was previously documented for fishers in North America (Larkin et al. 2011), this was the first case where complications from toxoplasmosis resulted in death (Gabriel 2013).

Wildlife-vehicle collisions may be a locally-critical mortality factor. Highway 41 is a very busy road locally referred to as the Wawona Road once it enters Yosemite National Park near the small community of Fish Camp. During the study period six non-collared fishers were also known to have been killed by vehicle strikes on Highway 41. Nine uncollared fishers were known to have been killed by vehicles along a 42 km stretch of Highway 41 between January 2008 to March 2013 in Yosemite National Park. Chow (2009) previously reported 4 fisher roadkill deaths between 1992 and 2004 along the same section of Highway 41, identifying this roadway as problematic for fisher survival in the region. Roadkill deaths of fishers have been reported in northern California as well, including two near Trinity Lake, (Truex et al. 1998), eight along paved highways in Humboldt and Siskiyou County (Gabriel 2013), and one in Butte County (Powell et al. 2012). In total, we are aware of 34 documented cases of fisher mortality by vehicle-strikes in California from 1992 to 2013 (Table D15). Moreover, seven fisher deaths were reported in western Washington state in association with the Olympic Fisher Reintroduction Project (Lewis 2014).
Our original prediction was that survival would be lowest during winter among each sex and age class. Sweitzer et al. (In revision) found that this prediction was not supported by the data, and that a disproportionate number of fisher deaths occurred during spring and summer. Increased mortality of fishers in this period is potentially related to exposure to second generation anticoagulant rodenticides, which is typically applied most heavily in the spring growing season. Expanded testing for anticoagulant rodenticides in archived tissues for fishers that died on our study before 2009, and for fishers that died on the Hoopa fisher study in northern California revealed that the majority of the animals had been exposed to anticoagulant rodenticides (>80%) and other toxicants being used in and around illegal marijuana grow sites on California public and tribal lands (Gabriel et al. 2012a). Ongoing investigations indicate that use of anticoagulant rodenticides at illegal grow sites is focused during spring and early summer when the marijuana plants are small and vulnerable to herbivory by rodents and insects (Gabriel et al. 2012a, 2013, Thompson et al. 2013). A total of eight fishers (three from SNAMP Fisher, five from the Hoopa fisher study in northern California) have now been documented as dying from exposure to rodenticides or other toxicants associated with marijuana grow sites (Gabriel 2013).

Another human-linked source of death for fishers in our study was entrapment or drowning in water tank. At the SNAMP site in spring 2008 we recovered the carcass of a non-collared fisher on the ground next to an open water tank (the cover had been ajar) where maintenance crews servicing the tank deposited the animal. Truex et al. (1998) and Powell et al. (2012) both reported deaths of single radio-collared fishers in abandoned water tanks at research sites in north central California, whereas Folliard (1997) recovered skeletal remains of eight fishers from an abandoned water tank on private timberlands in northwestern California. Finally, L. Davis (personal communication, Sept 7, 2013) reported the death of a radio-collared fisher that became trapped in a relatively short section of an upright culvert during a study of fishers in the Cariboo-Chilcotin region of British Columbia, Canada (Davis 2008). It appears that death of fishers by entrapment in water tanks and other human structures may not be uncommon. Folliard’s (1997) 15 year old recommendation that abandoned water tanks on private and public forests in California be covered, or modified by inserting branches or poles so that fishers and other wildlife can self-rescue should be applied whenever possible.
**Population Size and Density**

Prior to this study there was limited information on the distribution and abundance of fishers at the north margin of their extant range in the southern Sierra Nevada. Despite many years of surveys with cameras and track plates, the lack of evidence of fishers north of Yosemite Valley suggested that the population in the SNAMP Fisher study area was likely sparse (low density). Also, there had been no indication that surplus animals were dispersing northward into suitable, but unoccupied habitat north of the Merced River (Spencer et al. 2011, Spencer et al. 2015). Moreover, reports of multiple roadkill fishers along Highway 41/Wawona Road between the south boundary of the park and the tunnel just north of Yosemite Valley suggested that dispersal and the overall population was being limited by deaths on that highway (Chow 2009).

Federal and state agencies are currently developing strategies to manage for long term viable populations of fishers in the southern Sierra Nevada, and six years of intensive investigation as part of the SNAMP Fisher study has recently produced the first estimates of abundance for the region. We estimated the size of the fisher population in the overall SNAMP study population at 48 to 62 individuals (Table D2). Narrow confidence intervals for the population estimates were likely due to the combination of a relatively high probability of detection (0.4 to 0.75) for our camera protocol when cameras were within the home ranges of radiocollared fishers (Popescu et al. 2014) (Table D2).

Mean annual population density for the three Subregions of the overall study area ranged from 0.072 to 0.097 fishers fishers/km², which was consistent with data from two previous studies of fishers in the High Sierra District of the Sierra National Forest, located 50 km south of our study site. Jordan et al. (2011) used a similar CMR design to estimate a density of 0.063-0.109 fishers/km² for the Kings River study area in 2002-2004. Thompson et al. (2013) used scat detector dogs and genetic detections in a spatially explicit CMR framework modified for variable search intensity to estimate a fisher density of 0.065-0.28 fishers/km² for the Kings River Fisher Project area in fall 2007. Thompson et al. (2013) emphasized that a modal density of 0.104 fishers/km² was the most appropriate point estimate developed from their research. At a research site on the Hoopa Valley Indian Reservation (Hoopa Fisher Study) in northern California, Higley...
et al. (2013) used CMR methods to determine that the density of fishers increased from 0.12-0.29 fishers/km² over 9 years from 2005-2013. In central Massachusetts, USA, Fuller et al. (2001) applied CMR models to camera sightings and determined fisher densities of 0.19-0.25 fishers/km². Considering the subset of studies that used CMR methods, the densities we estimated for the SNAMP Fisher study area are the lowest reported (Table D2).

As previously detailed, conservation planning is underway for fishers in the southern Sierra Nevada, including new modeling to estimate areas of suitable habitat for fishers in occupied “core” regions within the southern Sierra Nevada (Spencer et al. 2015). Our study area is within Habitat Core and Connectivity Area 5, for which the area of suitable habitat was estimated as 1,096 km² (Table D2, Spencer et al. 2015). We calculated the mean density and 95% C.I. for 12 area- and year-specific densities developed by our CMR modeling (Table D2; 0.085 fishers/km², 95% C.I. 0.073-0.097), and estimated that there were 93 (range 80-107) fishers in the Southern Sierra Nevada Habitat Core and Connectivity area 5.

In the context of similar data from other studies, the population of fishers in the Bass Lake Ranger District extending into southern Yosemite National Park is small, genetically limited (Tucker et al. 2014), and exists at a density that is lower than has been reported for any part of California or North America with the exception of boreal forest regions of northern British Columbia, Canada (Weir and Corbould 2006). Moreover, there are important challenges to the long term viability of fishers in the southern Sierra Nevada region as a whole, including periodic epizootics of canine distemper (Keller et al. 2012), exposure to poisons and other toxicants that directly and indirectly increase mortality (Thompson et al. 2013), and large, catastrophic wildfires capable of eliminating thousands of hectares of foraging and denning habitat in short periods of time (days or weeks; Final Update on 2013 Rim Fire: http://inciweb.nwcg.gov/incident/article/3660/21586/).
Dispersal and Home Range Movements

Information on dispersal provides important insight on how far individuals of a species may move on their own, which is valuable for understanding the potential that unoccupied but otherwise suitable habitat will be colonized or recolonized by the species without management intervention. For their body size, fishers appear to be relatively poor dispersers and large scale genetic substructure analysis supports this observation (Kyle et al. 2001). Fisher movement behavior appears to vary by age, sex, season, and habitat characteristics. Juvenile dispersal may vary widely, depending on habitat availability and landscape permeability.

Intensive monitoring of individual fishers by fixed-wing aircraft, in combination with an expansive trapping effort across the entire SNAMP Fisher study area provided insight on dispersal that would have been difficult to acquire otherwise. Microsatellite DNA analyses to identify maternity for many juveniles and some subadults further extended our inference to larger numbers of potential dispersers (24 females, 19 males).

We found limited evidence that natal dispersal was male-biased according to any of the typical metrics reported in the literature for this life history phenomenon, however the small samples size and wide range in dispersal distances precluded any robust statistical comparisons. Dispersal distances were not significantly longer for males (mean = 8.46 km) compared to females (4.89 km) based on either Euclidean distances or for more realistic Least Cost movement paths (Table D23, Figs. D17). There was no significant difference in the proportion of each sex that dispersed, or that remained philopatric (Fig. D13, Table D23), and, similar numbers of males ($n = 5$) and females ($n = 3$) undertook long distance dispersal movements from their likely natal areas (Fig. D14). Timing of dispersal in the SNAMP Fisher study population was focused during mid-February to July, and the longest distance dispersal event a female fisher in the population undertook was 22.3 km (44.1 by the Least Cost Path), compared to 36.2 km for a male (69.8 by the Least Cost Path)(Tables D28, D29). We did document dispersal by several fishers across landscape features previously identified as restrictive based on population genetics (Tucker et al. 2012, Wisely et al. 2004). Four fishers regularly moved across the Chiquito Ridge (via Shuteye Pass), and two male fishers transitioned across the San Joaquin River canyon.
Our data on dispersal differed from reports from southern Oregon and northwestern California. Aubry and Raley (2006) reported that mean juvenile male dispersal distance was 29 km, while the mean dispersal for females was 6 km. Dispersal distance in the Hoopa area of Northern California averaged 4.0 km (range = 0.8-18.0 km) for 7 females, and was 1.3 km for one male (Matthews et al. 2013a), however the authors noted that their focus on capturing adult females limited their ability to estimate male dispersal.

The maximum known dispersal distance for fishers from the literature was 100 km (York 1996), while the maximum observed movement of a translocated individual in unoccupied habitat was 163 km (Roy 1991). The relatively small number of long distance dispersal events noted during the six year SNAMP Fisher study suggests that long distance movements are uncommon and that the effective dispersal distance may be less than maximum dispersal capacity (Tucker et al. 2013).

**Population Growth and Threats to Population Persistence**

Estimates of $\lambda$ for fishers derived from empirical data specific to the area of inference are rare for California, and absent for the southern Sierra Nevada. The All Year survival and empirically derived demographic rates produced a $\lambda$ of 0.90 (range 0.77-1.22). While this point estimate suggests a negative growth rate, it was encouraging that the range for the all year population growth rate extended above 1.0 (Table D28). Elsewhere in California, Higley et al. (2013) integrated data on apparent survival from CMR models and data on reproduction in a series of random effects models to evaluate $\lambda$ for fishers in the Hoopa Fisher Study. Two models produced $\lambda$ estimates close to or greater than 1 (Both sexes, Females only; see Higley et al. 2013). Swiers (2013) used Robust Design models, software program POPAN, and Pradel models to develop information on demographic rates and population size for assessing whether removal of adult fishers from a population in northern California/southern Oregon for translocation elsewhere negatively affected population growth. Swiers’ (2013) top ranked Pradel model produced a population growth rate of 1.06 (95% CI = 0.97-1.15), suggesting a stable or slightly increasing population after nine ‘prime breeding adult’ fishers had been live-trapped and removed from the population for translocation.
We identified several sources of mortality in the study population, and the indication of an overall negative population growth rate was in accordance with the fact that 60% of the 110 fishers that were radiocollared died (Table D16). The matrix model we developed was realistic and based on current knowledge of fisher life histories in California, but some demographic parameters were less well known than others. Survival of juvenile fishers during the three month period from mid-June to October is poorly known for our study, and for all other detailed studies of fishers in California (Facka et al. 2013). The estimate for juvenile female survival used in the matrix model was based on the 6-7 month period from October to March, which likely overestimated the number of juveniles recruited into the population. However, a basic sensitivity analysis indicated that the population growth rate was insensitive to variation in fertility for all age classes, and least sensitive to juvenile survival compared to subadult and adult survival.

SNAMP Fisher Management Indicators

Three management indicators we developed in 2008-09 as a mechanism for interim reporting on the status of fishers in the study area appeared useful when considered in relation to data on population growth rates and population density. Naïve occupancy in the Key Watershed was lowest in Camera survey years 2007-08, 2008-09, and 2009-10 when population growth rates were negative, but then increased in the later years when the growth rate was stable or positive (Tables D33, D35). The number of resident female fishers using the Key Watersheds did not track changes in population growth rates as closely, but the difference in the metrics was smallest in Population years 4 and 5 when the growth rate was negative or at approximate stasis. Adult female survival tracked change in population growth rate closely, declining from 2-year group 1 to 3, and then increasing afterwards (Table D28). Also, population density was in decline from 2007 to 2009, but then increased during Camera survey years 3 (2010-11) and 4 (2011-12) (Fig. D9), coincident with improved survival among juvenile and adult female survival (Table D28). We recommend that future long term studies consider developing similar metrics as a monitoring tool, and for interim reporting to interested stakeholders.
Fisher response to fuel management

Concerns that initiation of focused management to reduce fuel levels in Sierra Nevada mixed-conifer forests to correct for 90 to 100 years of fire suppression might have negative effects on habitat use by fishers were only partly supported by results from our study. Fisher occupancy was not negatively associated with either extractive or restorative fuel reduction, though disturbances from restorative fuel reduction had a negative effect on local scale persistence. We believe that the lack of a relationship between extractive fuel reduction and occupancy by fishers was most likely due to the combination of related factors. First, the overall extent of logging in our study in the 11 years from 2002 to 2013 was likely much lower than historically, and was likely further diminished by poor market conditions for wood products when a severe recession began in 2008. Second, estimates of annual disturbance from extractive fuel reduction among occupancy survey grid cells was equivalent to levels known “tolerated” by fishers elsewhere in the Sierra NF (Zielinski et al. 2013). Among the 361 multi-season survey grid cells, 172 of them encompassed 51.9 km$^2$ of disturbance from extractive fuel reduction, representing disturbances of 2.7%/year to grid cells with disturbance, and 1.3%/year among all grid cells. Zielinski et al. (2013) investigated tolerance of fishers to forest management in the High Sierra District, Sierra NF, and reported that 14 km$^2$ patches of forest habitat with high use by fishers typically had 2.6% of the areas disturbed by forest management annually, whereas 14 km$^2$ patches of forest with low use by fishers averaged 3.5% disturbance/year. Thus, the areas of extractive fuel reduction in our study were comparable to the 2.6% disturbance in high fisher use forest patches in the High Sierra District, Sierra NF, and below some threshold of ≥ 3.5% management disturbance/year that would likely cause fishers to use a different area (Zielinski et al. 2013).

Our occupancy modeling supported the hypothesis that fishers would reduce their use of local patches of forest exposed to proportionally higher levels of cumulative restorative fuel reduction. Nevertheless, an important prediction from our multi-season model was that small patches of forest with 100% of the area treated for hazardous fuels over 5 years would maintain an occupancy rate between 0.46 and 0.81, assuming no threshold effect. Thus, even at what would be considered a very high level of disturbance, fishers were not predicted to completely
cease using those areas. For context, an occupancy rate of 0.65 for fishers elsewhere in the southern Sierra Nevada would be considered high, and a positive observation with regard to long term continuation of occupancy (Zielinski et al. 2013).

Ladder fuels, surface fuels, and thick layers of duff targeted under SPLAT-based management provide important habitat for squirrels and rodents preyed on by fishers, owls, and other forest carnivores (Kelt et al. 2013). Therefore, if forest patches that were extensively treated for restorative fuel reduction harbored less abundant prey, fishers may have shifted to nearby less disturbed forest patches to forage. The possibility that thinning of trees and shrubs, and reduction in understory surface fuels (coarse woody debris) has a negative effect on rodent populations has been considered by several recent studies. Meyer et al. (2007) reported reduced captures of northern flying squirrels in forest stands that were thinned and underburned in the High Sierra District, Sierra NF. Treated stands had reduced canopy cover and relatively shallow litter depth, and Meyer et al. (2007) considered that reduced abundance of flying squirrels may have been due to reduced abundance of truffles (fruiting bodies of hypogeous fungi) when duff was removed or reduced in depth after fuel reduction. Amacher et al. (2008) reported a negative effect of fuel reduction treatments (without follow-on burning) on abundance of deer mice, a positive effect of managed burning for deer mice, but no detectable effects of thinning or burning treatments on long-eared chipmunks, California ground squirrel, or brush mouse (*Peromyscus boylei*) at a research site in the north-central Sierra Nevada. Amacher et al. (2008) suggested that scattered debris and wood shards from rotary mastication was associated with the negative treatment effect for deer mice, whereas follow-on burning removed residual woody debris and thinned the understory, thereby improving conditions for deer mice. Converse et al. (2006) reported lower density or a trend for lower density for gray-collared chipmunks (*Neotamias canipes*) and Mexican woodrats (*Neotoma mexicana*) in thinned+burned forest stands in Arizona, which was linked to reduced coarse woody debris and reduced density of shrubs. In that same study abundance of deer mice increased after thinning+burning, and there was no treatment-linked change in abundance for golden-mantled ground squirrel (*Spermophilus lateralis*) (Converse et al., 2006). In restoration-treated ponderosa pine forests in Arizona, Lobeerger et al. (2011) found that winter season home ranges of tassel-eared squirrels (*Sciurus aberti*) disproportionally encompassed areas that had not been treated, whereas in other seasons their home ranges included a subset of the treated stands that retained relatively high canopy cover.
Bull and Blumton (1999) indexed presence of small mammals from track surveys in lodgepole pine (*Pinus contorta*) and mixed-conifer forest stands treated for fuel reduction in northeastern Oregon. We were unable to identify studies that reported responses of Douglas squirrels or dusky footed woodrats (*Neotoma fuscipes*) to fuel reduction treatments, but, based on habitat associations for *Neotoma* (Innes et al., 2007; Kelt et al., 2013), understory thinning and removal of surface fuels and coarse woody debris may reduce habitat suitability for woodrats (Lehmkuhl et al., 2006), whereas Douglas squirrels are a habitat generalist and less likely to be negatively impacted by fuel reduction (Coppeto et al., 2006, Herbers and Klenner, 2007; Kelt et al., 2013). Kelt et al. (2013) suggested that small mammal assemblages in the Sierra Nevada showed relatively limited responses to canopy thinning under current forest management. Abundance of small mammals in the Sierra Nevada has been linked to variation in production of cones or hard mast by pines and oaks (Coppeto et al., 2006; Wilson et al., 2008), which is important because a general pattern in many studies we reviewed was that inter-annual variation in abundance of small mammals was evident, and either masked or was much more important than the smaller effects introduced by fuel reduction-induced change to habitats (Converse et al., 2006; Coppeto et al., 2006; Amacher et al., 2008, Wilson et al., 2008; Kelt et al., 2013). We therefore conclude that reduced local scale habitat use by fishers in grid cells with larger areas treated for restorative fuel reduction was not likely to have been caused by changes in abundance of rodent prey from the associated disturbance to their habitats.

We consider it likely that the predicted 27% decline in persistence of local scale habitat use when cumulative restorative fuel reduction in a 1-km² grid cell approached 1.0 (100%) was associated with fishers shifting to forage in adjacent areas with less disturbance. A 27% decline in persistence of occupancy coupled with an annual colonization rate of 34%, suggests that fishers are flexible with regards local scale habitat use, and they might resume use of treated areas after several years of ecological recovery. Modeling analyses by Thompson et al. (2011) applied to a fisher occupied area of the High Sierra District, Sierra NF (Bear Fen) suggested that tree thinning (≤ 89 cm DHB) in mixed-conifer forest did not significantly reduce habitat suitability or “displace” habitat components from reference conditions in home ranges of resident female fishers. Based on these results from a nearby area in the Sierra NF, we believe it likely that fishers in our study area are likely to resume using forest patches treated for restorative fuel.
reduction within a few years of extensive disturbance. Also, fishers are known to adjust space use to avoid disturbed areas within their home ranges. Garner (2013) reported that resident fishers included areas treated for extractive+restorative fuel reduction in their overall and core home ranges in proportion to availability on the overall landscape. At the finer scale of individual locations, Garner (2013) found that those same resident fishers avoided using areas within ≈ 200 m of fuel treatments. We interpret this result as consistent with ours; fishers were predicted to continue using 1-km² patches of forest with more extensive cumulative disturbance by fuel treatments, but at a reduced level compared to areas with less disturbance. Finally, our assessment of how fishers responded to forest management was at the scale of 1-km² patches of forest, which was small in relation to resident adult female (≈ 23 km²) and resident adult male (≈ 86 km²) home ranges in our study area. If a 1-km² patch of habitat within the home range of a resident female fisher was 100% treated for fuel reduction of any type, 95.7% of that animal’s home range could remain available for normal levels of foraging, contingent on SPLATs being dispersed on the landscape and not locally concentrated as appears typical (Modhaddas et al., 2010).

**Integration**

We found that the SPLATs at Sugar Pine slightly reduced simulated fire behavior and resulted in greater amounts of projected fisher habitat up to 30 years after the fire. In the absence of simulated fire, we found that the SPLATs had an immediate, negative effect on the amount of fisher habitat, but SPLATs did not generally have a negative effect on fisher habitat when we modeled future forest growth for 30 years. In all scenarios, the differences between the treated and untreated landscapes were small.

Our results were in general agreement with prior findings. Thompson et al. (2011) performed an analogous study to ours, in which they modeled fire and forest growth under treatment and no treatment scenarios and assessed fisher habitat suitability in the southern Sierra Nevada. They projected that fuels treatments had slight, short-term negative effects on fisher habitat, but provided significant protection in the event of fire and also extended the lifespan of current functional habitat. Truex et al. (2013) suggested that less fisher resting habitat was
present immediately after mechanical fuels treatments were implemented in the Sierra Nevada. However, fishers consistently used areas in the southern Sierra Nevada where some timber harvest had occurred, so it may be possible to implement fuels-reduction treatments at an extent and rate that achieves fire-hazard-reduction goals (Zielinski et al. 2013).

As we noted in Appendix C for the California spotted owl, the net benefits of SPLATs for the Pacific fisher will depend upon the true, but unknown, probability that high-severity fire effects will occur on a given portion of the landscape. However, future probabilities for specific fire behaviors (e.g., crown-fire initiation) are difficult to estimate, and it is therefore difficult to quantify trade-offs associated with SPLATs in absolute terms (Finney 2005). We further note that the SPLATs which were implemented at Sugar Pine appeared to have relatively modest impacts on forest structure and simulated fire behavior, and that it may be necessary to evaluate additional SPLATs of different intensities over a larger scale to fully assess the effects of SPLATs on fisher habitat. Nonetheless, we have no reason to believe that Forest Service managers should alter their current policy of avoiding the placement of SPLATs near known fisher denning sites (U.S. Forest Service 2004) because these sites have significant biological importance for this species.

Management Implications of Findings from SNAMP Fisher

Fishers have been the focus of systematic monitoring in the southern Sierra Nevada by track plates, hair snares, and cameras since the mid-1990s (Truex et al. 1998, Zielinski et al. 2005, Jordan 2007). Analyses of baited track plate detection histories from 2002 to 2009 for the entire southern Sierra Nevada fisher population found no evidence that the population trajectory for fishers in the area has been significantly positive or negative, based on constant and positive persistent values (Zielinski et al. 2013). In contrast, Tucker et al. (2014) suggested that the fisher population in the SNAMP Fisher study area was produced by a significant post-1900s northward population expansion involving dispersal of animals from south of the Kings River. Tucker et al. (2014) reported evidence of ‘strong genetic clustering’ to the north of Little Shuteye Peak (part of a high elevation ridge that forms the east boundary of Subregion 2 in our study area; Fig. D5), which, along with evidence for other small genetic clusters, was suggestive of multiple founder
events associated with contemporary population expansion. Data from track-plate surveys in the Sierra National Forest in the early 1990s rarely detected fishers (Zielinski et al. 1995, 2005), which suggested a very sparse population in the SNAMP Fisher study area, compared to the more recent surveys in 2002-2009 (Tucker et al. 2014). Tucker et al (2014) postulated that very few fishers were present in the SNAMP Fisher study area prior to the 1990s, and that an expansion that occurred only during the last 20-25 years produced the population in this region.

Genetic data are not typically used to make inferences about population processes operating over extremely short periods in evolutionary time. The genetic analyses of Tucker et al. (2014), and the large increase in fisher detections in the region encompassing our entire study area between the early 1990s and 2002-2009 (Zielinski et al. 2013), suggest that a significantly positive population growth rate would be a requirement for understanding the current distribution and abundance of fishers in the SNAMP Fisher study area. During the period from 2007 to 2014, our results suggest that the fisher population in this region has not been experiencing consistently positive or significant population growth (Table D18).

The suggestion of an overall negative population growth rate, the low density, and the relatively small estimated number of fishers in Fisher Core Habitat area 5 \( (n = 93, \text{range} \ 80-107) \), warrants concern for the long term viability of fishers in the region. Any small population will be at high risk to stochastic events such as disease and large perturbations to critical habitats (e.g., forest fires or drought; Noss et al. 2006), and genetic limitation resulting from genetic drift after founder events (Tucker et al. 2014) will hinder population recovery and expansion (Reed et al. 2003). Minimum viable population size has been under debate (Shoemaker et al. 2013, Reed and McCoy 2014), but at <500 total individuals (Spencer et al. 2004), the current southern Sierra Nevada fisher population will likely require active management and conservation measures to maintain a positive growth rate across its entire range. The observed variation in fisher abundance and rates of population growth in the SNAMP Fisher study area reaffirms the vulnerability of the small, isolated population to external threats (Spencer et al. 2015), especially wildfires that are likely to increase in frequency and intensity with climate change (Bonan 2008, Safford et al. 2012). Moreover, our study spanned a limited period of six years when multiple
threats to fisher survival within the study area were identified and during which three large wildfires further isolated the population by significantly reducing the availability of suitable habitat immediately to the south and north of the study site. We recommend continuous monitoring of the status of fisher populations in the southern Sierra Nevada region. It will be necessary to mitigate for major threats to fisher survival while maintaining contiguous expanses of suitable fisher habitats, and detailed analyses using realistic and empirically developed data on population parameters are necessary for evaluating the long-term viability of fishers in the southern Sierra Nevada. Data developed from the SNAMP Fisher study have provided important new insights on the status of a fisher population at the northern margin of their current distribution in the southern Sierra Nevada Range, which will be useful towards developing a comprehensive conservation strategy for fishers in California.
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Appendix D1: Important and Key Protocols for Pacific fisher

Live Trap Site Selection, Trap Setup, and Trap Checking Protocol

This protocol was developed to guide and help standardize site selection, placement and camouflaging of live traps for the SNAMP Fisher Project.

General Instructions on Trap Site Selection
1. Trapping methods
   a) Target Fisher Trapping – Collared fishers will occasionally “drop” their collars, requiring focused trapping for recollaring the animals. Also, radiocollars have a limited battery life (16-24 months) and we will periodically need to recapture animals for replacing their radiocollars. Traplines will sometimes be designed to capture/recapture fishers for these purposes. In these cases we will use information on the most recent locations to target fishers for capture. A trapline consisting of 5-10 traps will typically be placed in areas where multiple locations have been logged during aerial telemetry, focused at the microhabitat scale in areas with large trees and snags, near steam courses, and in areas with relatively high canopy cover.
   b) Trapline trapping – Most of our trapping occurs along traplines of 6-12 traps, where the number of traps depends on the area being covered and the logistics of access for checking traps. Traplines are typically designed along roads or trails in likely fisher habitat that is designated from a map designed in ArcGIS. GIS produced maps will always be used to guide the placement of traps along traplines. Maps of traplines will usually include the “Estimated” UTM coordinates for where the traps should be placed; do not second guess the estimated UTM coordinates by ignoring them and placing traps in other, more convenient locations far from the actual waypoints (far = more than 500 m from the coordinate). When navigating to the estimated waypoint, look for likely fisher habitat (areas with large trees, moderate to steep slopes, near stream courses, areas with high canopy cover) when you get close to the suggested position.

2. For the safety/security of trapped animals, traps should be at least 50 meters from a road or trail. Depending on forest vegetation, a greater distance from roads/trails may be necessary.
3. Just as cameras are placed in areas believed to be good resting habitat for fishers (according to Zelinski et al. 2004), trap sites should also be placed in areas with the following forest characteristics: high canopy closure, large DBH black oaks, large decayed logs, large DBH conifers, large DBH snags, moderate to steep slopes, and near or in drainages with running and/or pooled water.

Guidelines and Procedures on Trap Placement and Setup

IMPORTANT: Do not place the trap in the middle of what you believe is poison oak!

1. Whenever possible place the trap on level ground/snow, preferably at the base of a tree or next to a downed log to provide stability and to prevent the trap from tipping over with animals inside. Use a shovel to clear the duff from the forest floor and place the trap on the hard ground. In cases where some slope is unavoidable, orient the door of the trap facing slightly downhill; animals are generally more comfortable walking up into something than feeling like they might slip down into it.
2. Use dirt and sticks or tree bark to level the trap side-to-side. Also, make sure the trap is stable when you press on each corner of the trap, and that the trap door is unimpeded by plants and debris. If necessary, insert a small stick or slab of bark under the front of the trap to elevate it above uneven ground that may block the door from closing.

3. Test the door mechanism to ensure the treadle does not require too much force to close the door. Adjust the metal bar along the inside of the trap attaching the treadle to the door release; bending it will create more treadle tension and make it easier to trigger, but could prevent difficulties for straightening. The best approach is to use a pair of needle-nose pliers to adjust the curvature of the metal catch piece.

4. Check that the trap opening is square so the trap door does not catch on the sides.

5. Fully inspect the wooden nest box; remove old scat, bait or debris, and clean the trap with a mild bleach/water solution before taking into the field – spray the trap with the scent masking solution once it has been cleaned/disinfected with bleach water.

6. Make sure the metal plate at the back of the nest box can be slid out and the fold is pointing toward the front of the trap. Also, check that the metal plate has a pin or ring to hold it in place.

7. Mark and average the trap location (min 25 positions) with your Garmin GPS and record the UTM coordinates on the Trap Setup Data form.

8. Place plastic flagging on a tree either directly above or near the trap, so that it can be seen when walking from the nearest and most likely parking spot.

**Baiting/Prebaiting and Setting Traps**

1. When the trap is to be pre-baited or “wired open,” place a clip on the trap door and attach it to the roof of the trap. If an extra clip is not available, use the one from the metal door in the back of the cubby. Make sure to note this on the trapping sheet.

2. Smear Fisher Lure onto a chicken/venison-filled sock. Push the bait sock to the back roof of the wire trap, directly in front of the wooden nest box *(Tip: drape the top of the sock over the tip of a stick and use it as an extended arm)*. From the outside of the trap, grab the top of the sock and pull it through a hole in the wires, knotting it on itself so it is suspended in front of the nest box opening and cannot be pulled back into the trap.

3. Place Gusto on trees near the trap; somewhere in front of the trap as well as on trees/shrubs on either side of the trap. Avoid dabbing Gusto on trees/shrubs where someone might accidentally lean into it.

4. Whenever possible and depending on equipment availability, all traps should have a trapse site transmitter affixed onto the top of the trap. Attach the trapse site transmitter to the metal crossbar halfway along the outside of the trap roof. Loop the plunger wire through the wire loop at the bottom of the door and put a little oil on the plunger to ensure it will not freeze or stick inside the transmitter. Record the transmitter frequency on the trap setup datasheet. Test the trap door to ensure the plunger pulls smoothly out of the transmitter and does not catch on trap wire; use a receiver to make sure the signal tempo increases (the rate should double) when the closing door pulls the plunger out of the sleeve. Double check that the plunger has been inserted back into the transmitter sleeve after testing the transmitter and the trap door mechanism.
5. Once the trapsite transmitter is in place and after testing, place a canvas tarp over the body of the trap, tucking it close to the sides and placing small logs on the edges to hold it down. Place the tarp edge flush with the edge of the open trap door.

6. Fully cover/camouflage the top and sides of the trap with pieces of bark, sticks and green branches. Be careful not to place too much weight on the trap or it could be bent and not close properly; be sure not to place any heavy camouflage objects on the front of the trap that will prevent the door latch mechanism from working properly. Avoid placing any materials on the trap that could impede the path of the transmitter plunger as it is pulled out by the closing of the trap door. *The metal handled shovels have a saw inside of them and are very helpful to cut branches for trap camouflage.*

7. Before leaving the trap site, test that the trap door works. Then take several minutes to completely fill out the trap setup form.

**Checking Traps**

1. Begin in the morning by planning how to split up the traps if you’re working with another person and who to call if you get a fisher. Make sure that you have all the trap site locations before heading out into the field. Print the Trap Location file located in Z:\SNAMP_Capture_Trapping\SNAMP_Trapping Files\Fisher_Trap_Locations.

2. Gather a GPS, receiver, camera, flashlight, extra bait and lure, a capture kit (if available), and a Forest Service radio for checking traps.

3. Head out to your nearest trap location. Upon the capture of an animal, use a flashlight to determine the species. If it is a fisher (“target”), look for a radio collar (“old target”). If it does not have a radio collar it is called a “new target”. Call the predetermined personnel on the forest service radio and let them know what type of “target” you have and the “station” (trap) number. Be sure that you do not say you have a new or uncollared fisher, but rather a new target. Also, do not give any indication of your location or the trap location, especially the UTM’s. If you have caught an old target, use the receiver to identify the individual. If this animal is in need of processing use the metal slat to block the fisher into the wooden cubby. If you do not have one, navigate to the nearest trap. If it is empty, borrow the metal plate and make sure you bring it back after the capture. Then finish checking your traps and head straight back to the trap to set up the capture gear for processing.

4. If you have caught a non-target, let the animal go and clean out the trap. Animals will usually defecate and/or urinate inside the trap and can keep fishers from entering the trap in the future. If the trap is to be set again, reset the trap with a new bait sock with fisher lure.

5. When checking traps, make sure to re-scent the traps with fisher and Gusto scent lures every three days. Bait should also be replaced after 4-5 days in the summer and every 7 days or so during winter. If you replace the bait, remove the bait sock and the bait from the sock. Burry the bait in the soft forest floor and throw the sock away back at the field station.

6. Check that the trap door closes after pressing on the treddle from outside. Cover the trap with the canvas tarp (if available) and camouflage. Then head to your next trap.

7. Upon checking all your traps on your line, call the predetermined personnel and tell them that your “stations are all clear”. If you haven’t already done so, make sure that all of the trap forms all filled out.
8. Once you have returned to the office, pull up the Excel TRAP CHECK DATAFILE from the server (folder = “SNAMP_Trapping Files”) and enter the data from the trap forms into the computer database, save, and close the file.
FISHER CAPTURE AND PROCESSING PROTOCOL

This protocol was developed to guide and help organize the process and procedures to be followed during a fisher capture and processing event. A minimum two staff should be present at a capture event, but the preferred number of participants is three.

Outline of Protocol
1. When a fisher is captured
2. Capture Site Set-up
3. Using the capture cone
4. Sedation
5. Processing the fisher
   a. data recording
   b. head, tail processing
   c. PIT tag
   d. radio collar
   e. Collected samples
   f. Capture clean-up/wrap-up
6. Releasing the fisher
7. Label Samples
8. Data entry
9. Restocking the capture bags

1. When a fisher is captured
   a) When a trap contains a fisher the first step is to identify whether the animal is a new capture, or recapture of a previously processed animal. Remove the tarp and camoufage from the trap. Use a strong flashlight to look into the cubby at the fisher. Move the trap around gently until you get a quality look at the fisher’s neck to confirm the presence/absence of a radio collar. It is easier to confirm a collar presence, than an absence. Next, turn the receiver on to high volume. Slowly scroll through all the known telemetry frequencies for collared fishers.
   b) If the receiver does not pick up a signal; skip this paragraph. If the receiver does pick up a signal; note the animal’s id and use the telemetry fisher data sheet to ascertain the fisher’s status. The fisher should be immediately released if we do not need to re-collar, or collect more information from this animal. To release the fisher: point the cubby end away from you & remove the metal plate. Some fishers will need encouragement to leave the cubby. First try remaining quiet to allow the fisher time to leave. If that does not work, a combination of the following will work: wait longer, walk farther away from trap, gently tap on the cubby to encourage the fisher to turn towards the opening, blow into the cubby from the trap side, use a plunger (stick w/ blunt object attached to end) to persuade fisher out back of cubby while plunger enters from front of trap. In a case where time is limited you can leave the trap w/ the plate removed, and return later in the same day (leaving & returning later is not an acceptable release method for fishers that were just processed). After the fisher has left the cubby, clean the inside of cubby with a scent neutralizing agent.
   c) If the fisher is a new capture or re-capture that needs to be processed, notify the other crew members of the capture and station number. Then cooperatively establish a time
and place for all the capture participants to meet. Usually there will be other traps to check along the line, so check the remaining traps and then return to the capture. Before leaving the trapped fisher, place the second metal door (from capture kit) in the front slot of the cubby. If there is not a second door available in the capture kit, temporarily borrow one from another trap (be sure to return the door at the end of the capture). **DO NOT** move a trap w/ a fisher in it unless the second door is in place. Before leaving, make sure the trap is located in a spot that will be shaded until your return. However, if the ambient temp is extremely cold, the trap should be placed in the sun if there is no chance of overheating. Observe the immediate area to confirm there are no insect colonies nearby. Cover the trap with the tarp in case of inclement weather.

2. Setting up for Processing Target Captures
   a) **Background:** During set-up and throughout the entire capture it is important to maintain a quiet environment to minimize the level of stress on the captured fisher. Preparing for a capture involves assigning capture roles, setting up all the equipment, and testing both the radio collar and PIT tag before sedating the fisher. There must be at least two people present to process a fisher, but three is ideal to allow for a recorder role.

   b) **Assign roles for the capture team:**
      1. Person processing the fisher from the head end
      2. Person processing the fisher from the tail end
      3. Recorder (if available): if a recorder is not available, the two staff present will tradeoff data entry as needed in the course of processing

   b) **Equipment Set-up:**
      - Find or create a nearly level tarp sized surface in the shade/sun (depending on season).
      - Lay out the tarp with the insulated pad on top, in the center of the tarp.
      - Lay out a clean towel on top of the insulated pad. If there is a slope; roll up the second towel and place it underneath the first towel. This creates a bump that cradles the fisher preventing the animal from sliding downhill.
      - Determine which side of the tarp people will kneel on. Then decide which way the fisher’s head will be oriented on the towel. Place the sock and collar kit above the towel near the head end, the drugs kit near the tail end, and the samples kit in between.
      - Find/remove a pre-prepared fisher capture samples kit from the Capture Kit – the pre-prepared kit will be in 1 gallon Ziploc bag. Extract the Capture Datasheet from the capture sample kit, place on a data form holder, and begin filling out the top portion of Page 1 on the form. **NOTE:** the Capture Event Start time is the approximate time that all members of the capture team arrived at trap position and began setting up for the capture/processing. The Capture Event End time is when the fully recovered leaves the cubby.
      - Place the extra/empty Ziploc bag between the two processors for the completed samples. Label the hair sample envelopes “tail” and “nape”. Label the three swabs “ocular”, “nasal”, and “fecal”. Place the hair sample envelopes and the fecal swab
near the tail end, the PIT tag near the middle, and all other sampling supplies near the head end.

- Remove the Kestrel weather station device from the kit and turn it on for determining the ambient temperature for the capture/processing – record once temperature stabilizes.
- Find the stopwatch, reset it, and give to the person assigned to record data.
- Open the Vaseline and thermometer sleeves and set them near the tail end. Insert thermometer in a sleeve and stick thermometer in Vaseline so it is lubricated and ready to use.
- Each capture generates two types of trash; biohazards and sharps (needles). Place the red biohazard bag where it will be accessible by both processors, place the sharps container near the head end of the tarp.
- Place the ocular ointment next to the ocular swab near the head end.
- Put the laminated foot pad sheet near the tail end.
- Remove the measuring tape and ruler from the samples kit and place near the head end.
- Remove the calipers from the case and place near the head end.
- Check the scale to confirm it is zeroed. Set the scale and sling on the tarp off to the side.
- Prepare the sedatives (see section 4)
- Choose the appropriate size rubber gloves and have a pair readily available for each processor.
- Don a pair of leather gloves and prepare to use the capture cone.

c) **Testing the PIT tag and radio collar:**

- Remove the PIT tag reader from the waterproof case and turn it on.
- The reader will take a moment to warm up and then display “ready”.
- Hold the button down continuously on the reader and scan over the PIT tag until it displays the PIT tag identification number.
- If the reader does not register the PIT, the display will say “No ID found”. Try scanning the PIT tag again and be sure the button is fully depressed continuously while scanning. If the PIT tag is still not registering, then do not use this tag. Mark the PIT tag package defective and test a different PIT tag from the capture kits.
- After successfully scanning the PIT tag, place the tag and reader near the middle of the set-up.

d) **Radio Collar test:**

- Update all receivers present with the capture team with the new animal’s radio frequency. This can be done before the capture event starts during preparations.

**Receiver Background:** The receiver has three swivel knobs on top. Turn on your receiver by turning the ‘Power/Volume’ knob located in the lower right corner to the right. Make sure the ‘Gain’ knob on the left is turned as far up (right) as possible. The third knob, ‘Dial’, allows you to scroll through the channels used for designating certain animal’s frequencies. Scrolling through channels can also be accomplished by using the up and down arrows on the buttons in the lower left corner of the receiver.
Channels: The receiver has 1000 different channels (0-999). Male fisher are assigned channels 1-200 while female fisher are assigned channels 201-400 and each fisher’s ID is identical to the channel. (Ex. M01 = Channel 001, M02 = Channel 002, F01 = Channel 201, F25 = Channel 225, etc.) If you do not know the sex of the fisher yet, select a channel in the 500 range to test the collar’s frequency.

1. Turn on the receiver.
2. Press the ‘Program’ button. The screen will show three lines: the top is the frequency, the middle the channel, and the bottom is text (Fisher ID).
3. Use the down arrow button to move the cursor to the channel row. (Note: You must always first select a channel you wish to use).
4. To test a new frequency, type in a channel in the 500 range if you do not yet know the sex of the animal.
5. Use the upward arrow button to move the cursor to the frequency row.
6. Type in the collar’s frequency you wish to test. Press the ‘Enter’ button when finished. (Note: pressing enter a second time will save this information.) Since you are only testing, just press enter once.
7. Make sure the magnet on the collar is removed and listen for the beeps/tones emitting from the receiver.
8. Adjust the frequency by using the dial knob in the top right corner. New collars will usually drift down a few thousand decibels over a period of hours and days. If the radiocollar is functioning properly, remove the label tape with the frequency and place it in the box on Page 2 of the Capture Datasheet. Find the serial number for the collar (usually written in permanent ink on the inside of the leather strap of the collar.
9. Once the capture event has commenced and sex of the animal has been determined, enter the animal’s ID.
10. Follow Steps 1-3 above.
11. Type in the appropriate channel. (Ex. New female capture # 28 = Channel 228.)
12. Use the upward arrow button to move the cursor to the frequency row.
13. Type in the collar’s frequency. Adjust the frequency as in Step 8 above if the frequency has drifted.
14. Use the downward arrow button to move the cursor to the text or bottom row.
15. Turn the dial knob in the top right corner to scroll through the alphabet and numbers for the proper ID.
16. Press ‘Enter’ twice to save this information.

Most likely you will only be able to update the one or two receivers the capture group has with them, and you will have to update the others at the office when all work colleagues and gear return from the field. Don’t forget to also update the receiver used for aerial telemetry flights.

3. Using/setting up the capture cone
- Place trap in an area as flat as possible, and accessible by all capture participants.
- Orient the back of the trap upslope if there is an incline.
• Determine the size/gender of the fisher by opening the front plate and observing size. Choose the larger capture cone for males and the smaller cone for females.

• Attach the large end of the cloth funnel bag around the back of the cubby. After the bag is attached to the cubby, use a stick to twist the drawstring of the funnel bag tightly around the cubby.

• Attach the small end of the cloth funnel bag around the large end of the capture cone. Then use either a clamp or strap to cinch the funnel bag securely to the capture cone.

• Place the dowel rods and laundry bag close to the cone, but out of view of the exiting fisher.

• Have the Persuader hold the persuader dowel rod in hand.

• The people responsible for operating the dowel rods need to crouch/sit very close to the capture cone (but out of view of the exiting fisher) with two rods each ready for insertion.

• When all participants are ready and quiet, the Persuader removes the back panel & observes the fisher’s position. The Persuader may need to coax the fisher into the funnel bag by simply waiting longer, blowing in the cubby by removing the front panel, or using a plunger.

• After the fisher has completely entered the cloth bag, the Persuader uses the Persuader rod (in a horizontal position pressing the cloth funnel against the ground) to prevent the fisher from entering the cubby. The Persuader quickly moves the persuader rod forward to move the animal forward into the capture cone.

• Once the base of the fisher’s tail is visible in the cone, insert the dowel rods completely through the cone behind the fisher to create a web of rods that prevents the fisher from backing out. Then push the rods towards the nose of the cone until the fisher is unable to move forward. Hold the rods in place throughout sedation until the fisher is unresponsive.

• As soon as the fisher is pushed forward in the cone, lay the laundry bag over the fisher’s face to help calm the animal down.

• Sedate the fisher (refer to section 4-Sedation)

• After the fisher is completely sedated: remove the dowel rods, laundry bag & fabric funnel from the capture cone. Then, one person holds the capture cone at an angle to slide the fisher out the open end. Meanwhile the second person inserts their hands into the cone to guide the animal out the opening and hold the sedated fisher once it has been removed.

• Take the animal over to the tarp and begin processing the fisher.

4. Sedation

• Background: The drugs used to immobilize/sedate fishers for processing are Ketamine Hydrochloride and Diazepam (liquid Valium). The drugs are premixed in a 10ml vial kept in the capture kit. Both drugs are controlled substances and we are required to closely track the volume used at each capture. Both capture kits have a Controlled Substances Log: it is imperative that the Controlled Substances Logs are filled out and by recording the drug amounts used during captures

• Draw up appropriate volume of drugs using a 1 cc syringe

  o NOTE: Each capture kit will have a laminated sheet providing precise volume information based on the estimated body mass of the fisher in the cubby
• Working together with the capture team, encourage the fisher to exit the back of the cubby (after removing the aluminum plate) into the cone
• As quickly but as carefully as possible feel through the bars of the cone and locate soft but thick muscle tissue on the rump or one of the two hind limbs
• Slowly but firmly insert the needle into the muscle tissue, and gradually inject the drugs
• IMPORTANT: Do not jab the needle into the tissue, or quickly inject the entire volume of drugs by rapidly pushing the plunger of the syringe (this is painful to the animal).
• Remove the needle and start the stopwatch immediately; ideally, one member of the capture team will be observing the injection process, and will have started the stopwatch upon seeing the injection occur.

5. Processing the fisher
This process is broken down into two separate roles with one person working from the head end while the other works from the tail end. However, many of the tasks can be done by either person. Below the tasks are marked with either an “H” or “T” to represent the tasks typically done by the head or tail person respectively. Tasks that are often done by either person are unmarked. Before touching the fisher all capture participants will put on rubber gloves.

There is not a concrete order that has to be followed, but the crucial aspects of a capture are to weigh and sex the animal, collect samples, attach the radio collar, and insert a PIT tag.

1. Weigh the fisher
   • Place fisher centrally in the weighing sling and attach all the D rings to the scale hook.
   • Hold the handle loop on top of scale and lift until the fisher is elevated just above the ground.
   • The scale reading will vary if the weighing sling is swinging or bouncing, so wait until the sling stops moving to take a measurement.
   • Read the measurement to the nearest hundredth kilogram (ex: 3.35kg, 2.75kg)
   • Place the sling gently on the ground and move the fisher to the towel on the tarp.

2. Collect 3 swab samples- ocular, nasal & fecal
   • Check each swab label to confirm that the correct swab is used for each sample.
   • (H) Ocular- Carefully swab along the inner corner of one eye. Do not rub the eye. Re-cap swab and place in the collected samples bag. Squeeze a thin bead of ocular ointment into each eye so that it drops onto the eye starting at the inner corner. Softly massage the skin around each closed eye with two fingers to assure the ointment contacts the surface of the eye.
   • (H) Nasal- Insert just the tip of the swab inside each nostril and twirl swab around against the inside of each nostril. Re-cap swab and place in the collected samples bag.
   • (T) Fecal- Insert just the tip of the swab into the anus and twirl the swab. Re-cap swab and place in the collected samples bag.

3. Measure temperature and observe respirations
   • (T) Temperature- After collecting the fecal swab, insert the thermometer (w/ a plastic thermometer sleeve lubricated w/ Vaseline) into the anus and press the button on the thermometer to begin taking a reading. Record the temperature and the current time from the stopwatch. Remove the thermometer and place the used sleeve in the biohazard trash bag. Continue to take temperature readings throughout the capture every five to ten
minutes. Use a new lubricated thermometer sleeve for each reading. If the fisher’s temperature is >105°F cool the animal with ice packs along the abdomen. If the fisher’s temperature is <98°F warm the animal w/ heat packs along the abdomen and cover the body with towels.

- (T) Respirations- Each time a temperature is taken, it should be followed by observing the fisher’s respirations for 15 seconds. Multiply the number of respirations by 4 and record this number and the time found on the stopwatch.

4. Head and body measurements-
- Use the flexible measuring tape to measure:
  1- *(H)* Tip of the nose to base of the skull
  2- *(H)* Tip of the nose to base of the tail. After measurements 1 & 2 are done, put the sock over the fisher’s face covering the eyes.
  3- Dorsal body length from base of the tail to bony end of the tail
  4- *(H)* Head circumference around largest part of the skull (near the ears)
  5- Neck circumference
  6- Chest circumference- find the bottom ribs and measure slightly above bottom ribs where chest girth is the largest
- Use the hard plastic or hard metal ruler to measure:
  1- Width from ear tip to ear tip at the widest point
  2- Front of ears ear tip to ear tip

5. Collect remaining biological samples & put in the collected samples bag
- (T) Nape & tail hair- With fingers grasp several hairs near their base and pull them out with a quick jerking motion. Collect a hair sample from both the nape and at the base of the tail. Place hair in the appropriate envelopes.
- (T) Ectoparasites- Use the thin toothed comb to comb key areas of the fisher’s pelage (groin, anus area, abdomen, armpits, and nape) while carefully checking the comb for ectoparasites. Place ectoparasites in the small vial full of alcohol, and record the abundance and types.
- *(H)* Ear tissue- Prepare the sample area of the ear by cleaning it with an alcohol wipe. Place the wooden tongue depressor behind the ear to provide a backdrop to push against. Line-up the punch slightly inward from the edge of the ear. Avoid puncturing the ear along the edge, because this may contribute to tearing after the animal is released. Press the punch firmly through the ear against the depressor, then rotate the punch in place to assure the punch went completely through the ear tissue. Find the straight wire in the collar kit, and disinfect it with an alcohol wipe. Use this wire to push the tissue sample out of the punch into the vial of blue desiccant. If the desiccant is any color other than blue, use a different vial because the quality of the desiccant has been compromised.
- *(H)* Blood-

6. Insert the PIT tag
- Remove the PIT tag syringe from the packaging and lubricate the needle with antibiotic ointment.
- Locate the fisher’s scapulas with one hand. Then move to a slightly posterior position where the skin is loose enough to grab a handful of skin pulled away from the spine area. Align the knuckles of the hand parallel with the fisher’s spine and grasp the fisher’s skin. Pull the skin away from the spine about two inches so that it makes a triangle where the pinched skin is the apex of the triangle. With the other hand (that will hold the syringe)
push the skin in the middle of the triangle towards the fisher’s head to create an inward depression.

- Insert the PIT tag syringe into this depression, careful not to hit the body of the fisher or push the syringe out of the skin on the other side of the triangle.
- Once the syringe is completely inserted, use the three fingers that were previously holding the apex of the triangle to carefully locate the tip of the syringe through the skin.
- Hold three fingers around the tip of the syringe and depress the plunger of the syringe to insert the PIT tag.
- Hold the PIT tag in place with three fingers. Remove with syringe with a spinning motion out the same path as insertion.
- Massage/rotate the PIT tag in place to discourage it from accidentally slipping out the path of the syringe.
- Check the pelage in the immediate area of the tag insertion to verify the tag did not slip out.
- Re-check the PIT tag with the reader to confirm the PIT tag is still functioning properly.
- Affix one of the labels from the back of the PIT tag package onto the datasheet, and affix remaining labels onto other collected samples.

7. Age & Reproductive Condition

- (H) Check the sagittal crest development and record whether it is:
  1- absent
  2- partially developed
  3- well developed- large & obvious
- (T) Determine the gender & measure the genitals with the plastic ruler
  1- Females: observe the color and status of the teats whether they are nulliparous or enlarged.
      Then measure width and height of each teat. Look for swelling, hair matting around the nipples and lactation.
  2- Males: Determine if the testes have descended, and measure length and width of each teste.
- Take up close photos of the genitals using the macro setting on the cameras.
- Estimate the fisher’s age based on weight, appearance, teeth wear (done later), sexual maturity, and sagittal crest development.

8. Measurements and overall condition of teeth

- Count the number and condition of upper and lower incisors. Note whether they are discolored or worn.
- Classify the upper/lower canines & cusps of molar teeth as: sharp/pointed, points rounded, flattened, broken, and/or discolored
- Use the calipers to measure the teeth. First turn on the calipers. Move the tips completely together and press the zero button to calibrate the calipers. The display will continue to blink as you move the tips for measurements. Collect the following measurements for all four canines and note the general condition of each.
  1- Length of canine from tip of tooth to base (lateral gum line at lowest point)
  2- Width of canine at tip of tooth
  3- Width at base of tooth- measure base anterior to posterior at gum line
• Measure distance between tips of upper canines, and distance between tips of lower canines.
• Take pictures of teeth.

9. Attach the radio collar (H)
• Determine if a breakaway collar is needed (directions for breakaway at end of section).
  If female neck circumference is ≤12.6 or ≥18.6; then use a breakaway collar.
  If male neck circumference is ≤18.5 or ≥24.5; then use a breakaway collar.
• Remove the nut from the bolt on the radio collar and set nut in a safe place.
• Trim the leather strap close to the bolt plate to remove excess strap length.
• Put the radio collar around the fisher’s neck with the antennae pointing down the spine.
• Overlap the long leather strap against the bolt and adjust the size of the collar until two fingers fit loosely between the collar and the neck. Mark the leather strap with a permanent marker in the spot where the bolt will pierce the strap.
• Use the leather punch to put a hole in the leather strap. Make sure the smallest punch size is selected.
• Put the bolt through the new hole, screw on the nut, and test the collar size with two fingers.
• If the size is too tight/loose, then adjust the size and mark a new spot for the hole.
• If the size is appropriate, then completely tighten the nut down. Trim the excess leather strap so that it does not protrude beyond the radio/battery.
• Take pictures of the collar from the nut/bolt perspective and a shot of the antennae displaying the reflective tape pattern.
• Using a breakaway collar- Breakaway collars are the same as regular collars with the additional piece of glued leather strap that will break upon extended wear.
  1- Trim the leather strap (of the radio collar) close to the bolt plate.
  2- The breakaway collar is glued together with a rough side of glue and a smoother side. Identify the smoother side of the collar and face the smoother side towards the fisher.
  3- Lay the breakaway section in line with the collar strap and slide the breakaway towards the bolt plate until the seam of the glued sections butts up against the bolt plate.
  4- Mark the breakaway with a permanent marker for the bolt hole.
  5- Attach the breakaway to the radio collar in the same orientation as used to mark the breakaway. Make sure the rough glued side is facing out and that the nut is fastened tight.
  7- Now place the collar around the fisher’s neck with the antennae facing down the spine.
    The two loose ends of the collar will be attached with an additional bolt plate/nut oriented with the nut on the outside of the collar. Adjust the collar until two fingers fit loosely between the collar and the neck. Mark both leather straps with a permanent marker observing approximately a 1cm margin from the end of both straps.
  8- Punch a hole in both straps. Push the bolt through the radio collar strap with the bolt facing out from the fisher’s neck. Push the bolt through the breakaway strap and tighten the nut. Check for appropriate collar size. If the size is too large/small adjust accordingly and punch a new hole.
9- Take photos of both bolt/nut hardware sections of the collar, and a shot of the antennae.

10. Foot Measurements-
- Use the plastic ruler to measure the following lengths on both right and left feet:
  3- Length of front foot- distance from the proximal edge of the I4 pad to the distal edge of the fourth toe pad.
  4- Length of front foot- distance from the proximal edge of the I4 pad to the tip of the 4th claw.
  5- Width of the front foot- measure the widest part of the foot dorsally while excluding the first toe.
  6- Length of front foot- measure dorsal distance from the wrist to the distal edge of the foot/digits.
  7- Length of hind foot- distance from proximal edge of heel to distal edge of toe pad.
  8- Length of hind foot- distance from proximal edge of heel to tip of claw.

11. Pelage Color/Markings, Injuries, and Body Condition
- Overall Coat Condition- Choose prime, shedding, summer, mangy, or other
- Color of Head & Back- choose blonde, pale brown, dark brown, black
- Check for Markings on the Chin & Throat, Chest, Abdomen- if present choose blonde, pale brown, dark brown, or black
- Describe any other markings found.
- Take photos of the markings.
- Note whether there are any visible injuries, capture related injuries, or old scars.
- Rank the overall body condition as poor, average//good, or excellent/very healthy.

6. Releasing the fisher
- Before returning the fisher to the cubby, check and collect feces that may be present in the cubby. Deposit feces in a whirl pak and label with date, fisher ID, and contents. Place sample in the collected samples bag. Wear leather gloves to gently put fisher in cubby and replace metal door.
- Document stopwatch time fisher placed in cubby. If the fisher was very alert when placed in cubby, you can check their level of coordination with the tip test as soon as 15 minutes later. Watch the fisher’s reaction and balance while you quickly lift the front of trap and tip the trap side to side. If the fisher successfully counters, keeping their footing and upright stance during the tip test, then they are ready to be safely released. If they tip or stumble; they should NOT be released. Wait another 15 minutes and re-assess fisher’s status with another tip test. DO NOT release a fisher until it passed the test with controlled reactions of balance and coordination.
- Record the stopwatch time and real time when the fisher actually leaves the cubby.

7. Label Samples and Clean-up Processing Area:
- Place the labels from the PIT tag on each on the three swab samples so that the sample type can still be read. Place the remaining labels on the 2 hair sample envelopes, blood sample, and the ear tissue sample.
- Label the hair sample envelopes and fecal sample with date, fisher ID, and capture or recapture.
• Use alcohol wipes to sanitize the ear punch wire, thermometer, ectoparasite comb, and calipers.
• Clean up all of the equipment and replace in the appropriate bags. Place all dirty linens in the laundry bag to clean upon return to the office.
• IMPORTANT: Review the data form for completeness before departing the capture site.

8. Data Entry: When you have returned to the office
1. Update the whiteboard with new fisher information
2. Create a new fisher folder
   ▪ Navigate to SNAMP_Capture_Files found in SNAMP_Capture_Trapping
   ▪ Create a new folder denoted by fisher ID last 5 digits of PIT tag identification #.
     For example: F34_12345 or M56_54643
   ▪ Within this folder create two more folders for the scanned data sheet and capture photos
   ▪ Label 1st folder “fisher id_Capture Photos_yyyymmdd” (ex: F34_Capture Photos_20081231)
   ▪ Label 2nd folder “fisher id_Capture Sheets_yyyymmdd” (ex: F34_Capture Sheets_20081231)
3. Download capture photos to capture photo folder you just created
4. Scan data sheet and save to capture sheets folder you just created
   ▪ Place side 1 of capture form (w/ top of page to the right) facedown in the HP PhotosmartC6180, Press “Scan Menu” button, then select “Scan to computer”
   ▪ Choose to scan to the computer that you are working on and select “Save to file”
   ▪ When the computer prompts you; flip the data sheet and scan side 2
   ▪ The comp will ask if you want to scan again, chose “done”
   ▪ Both pages will be saved as the latest JPEG file in “My Scans” in “My Documents”
   ▪ Copy this file and drag it into the Capture Sheets folder you just created
   ▪ Rename the file as “fisher id_Capture Sheets_yyyymmdd” (ex: F34_Capture Sheets_20081231)
5. Update all four receivers with the new animal’s radio frequency.
   **Receiver Background:** The receiver has three swivel knobs on top. Turn on your receiver by turning the ‘Power/Volume’ knob located in the lower right corner to the right. Make sure the ‘Gain’ knob on the left is turned as far up (right) as possible. The third knob, ‘Dial’, allows you to scroll through the channels used for designating certain animal’s frequencies. Scrolling through channels can also be accomplished by using the up and down arrows on the buttons in the lower left corner of the receiver.

   **Channels:** The receiver has 1000 different channels (0-999). Male fisher are assigned channels 1-200 while female fisher are assigned channels 201-400 and each fisher’s ID is identical to the channel. (Ex. M01 = Channel 001, M02 = Channel 002, F01 = Channel 201, F25 = Channel 225, etc.) If you do not know the sex of the fisher yet, select a channel in the 500 range to test the collar’s frequency.
   1. Turn on the receiver.
   2. Press the ‘Program’ button. The screen will show three lines: the top is the frequency, the middle the channel, and the bottom is text (Fisher ID).
3. Use the down arrow button to move the cursor to the channel row. (Note: You must always first select a channel you wish to use so the receiver).
4. To test a new frequency, type in a channel in the 500 range if you do not yet know the sex of the animal.
5. Use the upward arrow button to move the cursor to the frequency row.
6. Type in the collar’s frequency you wish to test. Press the ‘Enter’ button when finished. (Note: pressing enter a second time will save this information.) Since you are only testing, just press enter once.
7. Make sure the magnet on the collar is removed and listen for the beeps emitting from the receiver.
8. Adjust the frequency by using the dial knob in the top right corner. New collars will usually drift down a few thousand decibels. Adjust the frequency for the ultimate audio signal and be sure to record both the original and ‘actual’ drifted frequency on the capture sheet.
9. Once the capture event has commenced and sex of the animal has been determined, enter the animal’s ID.
10. Follow Steps 1-3 above.
11. Type in the appropriate channel. (Ex. New female capture # 28 = Channel 228.)
12. Use the upward arrow button to move the cursor to the frequency row.
13. Type in the collar’s frequency. Adjust the frequency as in Step 8 above if the frequency has drifted.
14. Use the downward arrow button to move the cursor to the text or bottom row.
15. Turn the dial knob in the top right corner to scroll through the alphabet and numbers for the proper ID.
16. Press ‘Enter’ twice to save this information.

Most likely you will only be able to update the one or two receivers the capture group has with them, and you will have to update the others at the office when all work colleagues and gear return from the field. Don’t forget to also update the receiver used for aerial telemetry flights.

9. **Restocking the capture bags**
   - Upon returning to the office, launder the capture gear (denim cone, towels, washcloths, sock, and laundry bag). Put these items back in the capture bags once they have completely dried. Re-stock the capture bags with a new capture kit and radio collar. Check all capture supplies such as thermometer covers, heat packs, iodine, etc. and restock if low. If a metal plate from the capture bags was used, make sure this is returned at the end of the day to the capture bags.
CAMERA TRAP SETUP AND CHECK PROTOCOL
This protocol was developed to guide field procedures used to prepare and deploy camera traps in the field for surveying for fisher presence/absence in 1-km² grid cells.

Camera Trap Preparation
1) Load 8 “C” batteries in camera.
2) Connect battery cable to camera cable.
3) Format CF photo card for camera:
   • Open Silent Image- Mapview Professional program.
   • Go to “change camera settings” to format camera time, and name CF card.
   • Use default for most settings, except naming CF card, and setting time and date.
   • Go to “images” to name the CF card. Call it “UCB X.” X corresponds to the number of the camera being used. A “b” card will also be used, as described later, but it does not need to be formatted.
   • Go to “set date and time” tab to set the camera time for the first use. Set the date. Set the time (24 hr) of the card ahead a few minutes, to give enough time to remove it from the computer and place it in the CF card location in the camera.
   • Set the temperature to Celsius.
   • Place CF card in Camera and switch camera to “on” at the exact time formatted on the computer. Now the camera is formatted with the correct time and is ready for use. New blank cards placed in the Reconyx camera after initial formatting do not need to be formatted at this point.
4) Heat 4 desiccant packages in oven for 3 hours, at 170°. Four desiccants can also be microwaved 30 seconds (not recommended to go longer than 2 minutes), but oven use is preferable. Use rubber band to secure packets to one of the cables. Camera is now ready to be placed in the field.

Site Selection and Field Setup of Camera Traps
1) Use center point grid coordinates (UTM: EEE500_NNN500) as waypoint in Garmin GPS to drive as close as possible to the center point of the target 1 km² grid cell for positioning the camera trap
   a. our goal is to site the camera in the best possible location in the grid area based on resting habitat selection by fishers (review Zelinski et al. 2004)
   b. ideal forest areas for camera traps will include high canopy closure, large dbh black oaks, large decayed logs, large dbh conifers, large dbh snags, moderate to steep slopes, and be near or in drainages with running and/or pooled water (do not place cameras in flat areas with an open canopy, or at elevations above 7000 ft. until further notice)
   c. identifying the best location for a camera trap will require an adequate survey of most of the grid area prior to settling on the final camera trap location. This is an
optimization problem trading off between ease of access, security of site away from areas likely used by people, and high fisher habitat suitability

2) Survey the area identified for the camera trap for trees that will provide good mounting positions for both the camera trap and the bait sock: *camera traps must face in a northward direction (approx 320° N to 40° N) to avoid triggering by sunlight*

3) Plan to hang both the camera and bait on sturdy trees: *the distance between the tree with the camera and the bait tree must be between 3m and 5.5m, measured by a meter tape*. It is critical to find two trees where the bait tree is larger than the camera tree, ideally the camera tree is 25 to 40 cm dbh and the bait tree is 50cm+ dbh.

4) Use/Complete a Camera Trap Set Up form for recording detailed information on the GPS location, positioning/orientation of camera to bait, and other details on the camera station

5) Clear a visual path between camera and bait tree
   a. *The goal is to minimize situations where animals triggering events/photographs are obscured from view by litter, woody debris or other vegetation*
   b. *Use the small hand saws, lopping shears, or hatchet (backside of a hammer) during this process*

6) Center bait in upper third camera view, and leave approximately 1-2 m of ground view in front of bait tree
   a. *Perform a walk test to aid in correctly positioning the angle of the camera to the bait*
   b. *Use digital image viewer to verify camera is functioning properly, and that the angle and view are acceptable – reposition camera as needed*

7) Place bait (chicken, road kill venison, etc.) in sock, and secure the bait sock firmly to the tree at about eye level
   a. *Place one piece of bait in sock*
   b. *Nail one nail at the top of a folded sock into the tree in summer. During winter months, nail both the top and bottom of the sock to the tree, which will require a duration of effort by the fisher or other animals for removal = more photographs*

8) Hang a string of walnuts and/or pecans above bait sock. This consists of a wire threaded through ten nuts. Smear a tablespoon-size glob of peanut butter over nut bracelet. This particular bait is intended to provide an index to abundance of squirrels and other small mammals within the 1 km² grid area

9) Nail a pre-prepared “reflective tape wood slat” or yardstick on bait tree, high end at bait sock level. The reflective tape wood slat is used to determine body size and sex of any fisher that climb the tree to get at the bait.

10) Place Gusto on one end of a small stick and insert the “clean” end under the reflective slat/yardstick. Smear a small amount of Gusto near the base of the bait tree and on 1-2 trees or shrubs in the immediate vicinity. Dab a small amount of Fisher Scent lure on the bait sock itself. IMPORTANT: The field worker that sets out the Gusto, fisher lure, and bait sock, etc. should not set up the camera. Any scent from Gusto, etc. may cause bears
APPENDIX KEY PROTOCOLS: CAMERA TRAP SETUP AND CHECK

or other large predators to “investigate” the camera, damaging it, or knocking it off center from the bait tree. Before leaving the camera trap site, spray scent neutralizer on a paper towel, and rub over camera.

11) Use a Pentax camera to take “reference photos” of the site and camera. IMPORTANT: The first picture should use the “macro” function to photograph the ID scratched in the side of the camera. Other photos should include a shot of the bait tree (from the camera location), a shot of the camera and it’s tree (from the bait tree location), and then a couple photos off to the side to show the general area, habitat type, notable features, etc.

12) After Reconyx camera is mounted on tree, turn it on. Once the cam has finished loading up and the yellow and green lights are on, enter the pass code. When the green light appears (approximately 5 seconds) press “A/1” button until green light flashes. Pass hand over camera image detector until red light appears inside camera body (a click will also be heard). The camera is now working. Pass hand over image detector again, and verify that pictures have been taken. The red light inside camera will flash three times if there is a high level of light. If it is sufficiently dark outside, the infrared light will flash on the outside body of the camera.

13) Take a few pictures of the bait tree, to verify if everything is centered. Use laptop or digital image viewer to verify this, and make necessary corrections.

14) If everything is centered well, turn camera back on, and repeat procedure listed in step 11. Leave site.

IMPORTANT: COMPLETE THE CAMERA STATION DURING CAMERA TRAP SETUP INCLUDING COMPLETE INFORMATION ON ALL ASPECTS OF THE CAMERA TRAP POSITION AND SITE

DATA ENTRY FOR NEW CAMERA TRAPS AND CAMERA TRAP CHECKS ONTO SERVER AT OFFICE

Folder Set-up for Camera Trap Data

2. “Save as” immediately (so as to avoid messing up the Template) by clicking the ribbon button on the top left corner within Excel and click Save As or press F12. In the pop-up browser window, navigate to Camera_Monitoring_SetUp_Results_Archive and rename file as “SNAMP_Camera_Monitoring_SetUp_Results_YEARMODD_XXX.xls” for the day you are entering your data, with your three initials at the end. Save as type: Excel Workbook. Click Save to save the file in the Archive. You can also save it in the Data Entry Templates folder on the desktop of the computer you are working on and then cut and paste it into the Archive when you finish data entry.

Downloading Reference Photos

1. When setting up a grid survey camera in the field for the first time, you will take several “reference photos” with our Pentax cameras. When you are entering your data for
camera setup, be sure to use the camera cord to download these pictures to a new folder within the grid survey folder, clearly labeled with the grid, UCB camera ID, and “ref photos”, for example: 267_4141_UCB050_Ref Photos. If a new camera has replaced the original, make sure to use the full name history in the data sheet, and to update the name for previous checks in the Master data file. For example, a station with original camera 014 replaced with new camera 028 should be entered as UCB_014_028 in the current data sheet and in all previous entries in the Master.

**Data Entry**
1. Enter all collected data into the template using the drop-down cells provided. Name the grid cell ID and camera ID cells in reference to the top row. If there is not an option within the drop-down box which describes your results, manually type it in. Be sure to name the grid cell and the camera used with the provided format.
2. Note: the monitoring period column will automatically calculate once the initial set-up date and check dates have been entered. If a camera was not taking pictures because it was disturbed or is malfunctioning make sure to count up the days it was inactive and enter into cell “AL”. Make sure to describe the issue in the detailed notes column.
3. Continue to enter all your camera monitoring and/or setup data and save when finished.

**Append your Data**
1. Open Z:\SNAMP_Cameras\Master Data Files\SNAMP_Camera_Monitoring_Setup_Results_Master.xls.
2. Expand your data sheet. Select and copy the rows of the new data you have entered.
3. Expand the Monitoring SetUp Results Master worksheet and click on the next empty row. Right-click and hit Paste Special… Select Paste Values and click OK. Check to see that all your new data has appended to the Master worksheet.
4. Save and close the Monitoring Results Master worksheet within Excel.
5. Save and close your data entry worksheet within Excel and close Excel.

**Update Active Camera Trap worksheet**
1. Open Z:\SNAMP_Cameras\Master Data Files\SNAMP_Active_Camera_Traps.xls.
2. Select all cells by clicking the top left corner of the worksheet, or press CTRL+A. In the home tab, click Sort & Filter: Custom sort. Make sure the box “My data has headers” is marked. Click Sort by: Camera.
3. Update the worksheet to reflect the cameras you checked today. Enter the day you checked the camera on the next available CheckDate cell. Change the Next Check Date cell to seven days from the day you checked the camera. If the rows of the cameras you checked were highlighted, un-highlight them.
4. Select all cells by clicking the top left corner of the worksheet, or press CTRL+A. In the home tab, click Sort & Filter: Custom sort. Make sure the box “My data has headers” is marked. Click Sort by: Next Check Date. Highlight all rows of cameras the need to be checked in the immediate future.
5. Save, print and close this file. Close Excel. Attach printed worksheet to whiteboard for others to view.

IMPORTANT: When finished with all data entry, be sure to return the field version of the camera data sheet to its correct folder in the black file box labeled “Camera Monitoring”. This should be done quickly so that others can access the data.

CAMERA TRAP CHECKS AND SERVICE 8-10 DAYS LATER

1. 8 days later, return/navigate to Reconyx camera site using previously recorded coordinates as waypoints in handheld GPS.
2. Turn camera off and replace CF card “a” with “b” card. This new card does not need to be formatted, but does need to be labeled properly with a marker. If you do not have a spare card, see PhotoViewer downloading below.
3. Turn on the camera according to the above instructions (Field Setup #12). Trigger the camera and then view several images from the CF card with a digital image viewer to verify the camera remains angled properly.
4. Use the digital image viewer to screen through photos for the check period. Record the species present/detected in area for “notable species” sightings on the data form.
5. Remove old bait sock even if it is undisturbed. Nail a new bait sock to the tree and refresh Gusto and fisher lure. Add new nut bait if missing or mostly eaten. Refresh nut bait with more peanut butter.
6. Turn camera back on, verify that the battery level is sufficient (>30%, on new style Reconyx) and depart the site. Do not touch the camera after you have touched the gusto and fisher lure. It will attract bears to the camera. If you are the only technician, use sticks to turn on the camera and close it. Or, you can spray scent neutralizer on a paper towel, and rub over camera.
7. Check every 8-10 days, for a total of 4 checks and 32-40 days. If the camera is checked late and the 32 survey days are reached by check 3, the camera still needs to stay out for a fourth check.
8. If the camera was operating properly for all checks and was not disturbed by bears, remove the camera, bait, wood slat and all nails on check 4.

PhotoViewer Downloading

1. Insert CF card into Epson PhotoViewer and turn on.
2. Go to: Memory Card ➔ Browse CF Memory Card. Press OK.
3. Press the Menu button. Go to: Copy/Mo➔ Copy to Folder. Press OK.
4. Press the Menu button. Highlight Select All and press OK. Press OK.
5. Go to: My Photos➔ Create New folder and press OK.
6. Name the file by the Grid cell, camera being used, the date you’re downloading and what monitoring check this is. For example 273_4144_UCB_005_20080108_Check1 corresponds to grid cell 273_4144, Camera UCB 005, downloaded on 8 January 2008 and it’s the first check for this camera location. Navigate to Done and press OK.

7. Check to make sure your photos were copied to the PhotoViewer. Navigate to My Photos and find the folder you created and press OK.

8. Once you have confirmed the photos were copied press back until you reach the main menu screen. Eject the CF card and turn off the PhotoViewer.

9. Return CF card to the camera, refresh bait and lure, turn camera back on and depart site.
CAMERA TRAP IMAGE TAGGING PROTOCOL

This protocol was developed to help guide the summary process for camera trap images. Each camera records important metadata with each photo taken. Metadata includes date, time, moon phase and other conditions recorded at the time of image capture. After photos have been downloaded to a computer via CF card or a PhotoViewer and the camera has been removed from the field, we will use RECONYX MapView to view the photos and add keywords (“Tag Images”) to the Metadata file. Our added keyword tags describe what the camera actually recorded in the image. The metadata for each monitoring session will then be appended to a Master data file, which can be used to investigate the efficacy of our camera trapping effort.

Loading Images into MapView
1. Open Mapview, located on the desktop, in the quick-launch taskbar, or in Start → All Programs → RECONYX → MapView.
2. Click on the drop down arrow under the heading ‘Site’ and ‘Choose a site’ to make sure the Grid Cell you are entering data for exists. If your location site is not there, click ‘Create a new site.’ Name the site by the Grid Cell you will be tagging.
3. Click on the down arrow to the right of the ‘View/Load New Images’ button located in the upper left hand corner of the program window. Click on ‘From Another Folder.’
4. Navigate to the camera folder you want to summarize. The pathway should resemble the following: Z:\SNAMP_Cameras\Camera_Images_Survey_Year_04\Removed Camera Images\Removed_Images_Backed_Up\[Grid ID]\[CheckX]. Click ‘OK.’
5. Click ‘Check-All.’ Click ‘Next.’
6. Click ‘Add a new location.’ Name the location as the grid cell and CheckX (for example: 268_4144_Check1) and click ‘OK.’ Select your new location and click ‘Next.’ Click ‘Next’ again. Once the images have been copied, click ‘Finish.’
7. MapView will revert back to the original screen. In ‘Camera locations’ select the images you want to summarize. Click on ‘View Images.’ This will load the images into the viewer. Select the view style that suits you. The default view style is Detail, which is preferable to Thumbnail because it is quicker to load.
8. If your images have not yet been flipped to Portrait, refer to After uploading images to computer, flip photos right-side up and transfer photos to server, located in: Z:\SNAMP_Protocols\SNAMP_Camera_Image_Download_Protocol.doc

Tagging Images with Keywords
1. First, tag all the images with the monitoring session (check) number. Click on the first image in the viewer. Press CTRL+A to select all. Right-click on the selected images and click on ‘Image Data.’, click ‘Add a Keyword.’, click ‘Click to choose a keyword.’, click on ‘CHECK #’ and select the appropriate check number. Click ‘OK.’
2. Next, tag all of the images for camera type using the same process as above for check number. Refer to SNAMP_Active_Camera_Traps for the camera type for the grid you selected. Press CTRL+A to select all. Right-click on the selected images and click on ‘Image Data.’, click ‘Add a Keyword’, click ‘Click to choose a keyword.’, click on
‘CAM_TYPE’ and select either ‘KW_Survey’, ‘1k_Survey’, ‘HE_Marten’, ‘4k_Survey’, ‘Smammal’, or ‘YNP_1kSurvey’. Click ‘OK.’

3. Once all the images have been tagged with the check number and camera type, it’s time to tag images that contain pictures of animals. An important fact about these Reonxy cameras is that when the camera is triggered, it takes photos in bursts of three images at a time. Thus, an animal that triggers the camera will have a minimum of three photos, even if it runs out of the camera’s view before the 2nd or 3rd photos are taken.

4. Flip through the images loaded into the viewer. Often there will be setup photos of our colleagues in the beginning and end of each check. Select these and tag as ‘Setup’ under the SPECIES_EVENT keyword option.

5. Begin tagging blocks of images by species. An image should be tagged with a particular species if it contains an animal or a blur of an animal, or if it is part of an animal-triggered series of images even if the animal is no longer visible. Hold CTRL and SHIFT and click on the series of photos associated with the first species you wish to tag. Right-click on this selection and click on ‘Image Data.’

6. Click ‘Add a Keyword.’ Click ‘Click to choose a keyword.’ Click on ‘SPECIES_EVENT.’ From the dropdown menu, choose the species depicted in the selected images.

7. If you cannot identify an animal to species, try to identify to family. In the ‘SPECIES_EVENT’ dropdown menu, there are options for Canid_Unknown and Squirrel_Unknown. Birds must only be identified to class, i.e., should be tagged as Bird, but you are encouraged to identify to species in the Narrative tab. If you cannot place an animal in any taxonomic group, select Animal_Unknown.

8. If the images were triggered by a domestic dog, tag ‘SPECIES_EVENT’ as Dog, and make notes in the Narrative tab as appropriate. In particular, note the presence of multiple dogs.

9. If the images appear to have been triggered not by an animal but by environmental conditions, choose the ‘SPECIES_EVENT’ option that best fits your assessment of the situation. As follows is a list of options and when they should be used:

<table>
<thead>
<tr>
<th>Shadows_Wind</th>
<th>Triggered by tree parts moving in the wind during daytime.</th>
</tr>
</thead>
<tbody>
<tr>
<td>Weather_Wind</td>
<td>Triggered by tree parts moving in the wind during nighttime.</td>
</tr>
<tr>
<td>Weather_Precip</td>
<td>Triggered by rain or snow, including snow falling from tree branches.</td>
</tr>
<tr>
<td>Sunlight_Exposure</td>
<td>Use when bright sunlight obscures a good portion of the image and no movement is detectable.</td>
</tr>
</tbody>
</table>
| Tree_Branch           | Triggered by a river or stream in the background uncommon.
| Stream_Water          | Triggered by tree or large branch falling or pine cone rolling independent of wind uncommon. |

10. If the images were triggered by a human or vehicle not affiliated with the SNAMP study, tag ‘SPECIES_EVENT’ as Human/Vehicle. Include any notes in the Narrative tab.
11. If the images do not appear to have been triggered by an animal, human, or environmental conditions, tag ‘SPECIES_EVENT’ as Unknown.

12. We may now affix the keyword, IMAGE QUALITY, to those images that require it. As follows is a list of IMAGE QUALITY options and when they should be used. If none of the following scenarios apply to an image, it should not be tagged for IMAGE QUALITY.

<table>
<thead>
<tr>
<th>IMAGE QUALITY</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>Poor</td>
<td>The image contains an animal, but the animal cannot be identified from that image alone.</td>
</tr>
<tr>
<td>Animal_Not Visible</td>
<td>The image is part of an animal-triggered series, but the animal is not visible.</td>
</tr>
<tr>
<td>Disturbed_Off View</td>
<td>The camera was previously disturbed, and is no longer focused on the bait tree.</td>
</tr>
<tr>
<td>Disturbing_Bear</td>
<td>The camera is in the process of being disturbed by a bear.</td>
</tr>
<tr>
<td>Disturbing_Fisher</td>
<td>The camera is in the process of being disturbed by a fisher.</td>
</tr>
<tr>
<td>Disturbing_Squirrel</td>
<td>The camera is in the process of being disturbed by a squirrel.</td>
</tr>
<tr>
<td>Disturbing_Unknown</td>
<td>The camera is in the process of being disturbed by an unknown animal.</td>
</tr>
<tr>
<td>Cam_Malfunction</td>
<td>The image is dark or blurry due to a problem with the camera.</td>
</tr>
</tbody>
</table>

13. When tagging bear images, there are two additional steps you may undertake. After tagging ‘IMAGE_QUALITY’, add the keywords ‘BEAR’ and ‘BEAR COLOR.’ ‘BEAR’ asks you to assign a size/sex category to the bear as follows:

<table>
<thead>
<tr>
<th>Size/sex Category</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>Sub_Adult</td>
<td>Smaller than adult bear, but unaccompanied by mother.</td>
</tr>
<tr>
<td>Female_1</td>
<td>Female with one cub.</td>
</tr>
<tr>
<td>Female_2</td>
<td>Female with two cubs.</td>
</tr>
<tr>
<td>Female_3</td>
<td>Female with three cubs.</td>
</tr>
<tr>
<td>Female_4</td>
<td>Female with four cubs.</td>
</tr>
<tr>
<td>Large_Male</td>
<td>Large adult bear travelling alone.</td>
</tr>
<tr>
<td>Unknown</td>
<td>Unable to place into size/sex category.</td>
</tr>
</tbody>
</table>

14. ‘BEAR COLOR’ asks you to describe the bear’s coat as Black, Dark_Brown, Light_Brown, Blonde, or Unknown.

15. At this point, the Add/Edit Image Data window associated with a particular image may have up to 6 keywords: CHECK #, CAM_TYPE, SPECIES_EVENT, IMAGE_QUALITY, BEAR, and BEAR COLOR.

***THE ORDER IN WHICH IMAGES ARE TAGGED SHOULD BE CHECK#, CAM_TYPE, SPECIES_EVENT, IMAGE QUALITY, BEAR (WHEN APPLICABLE), BEAR COLOR (WHEN APPLICABLE). IMAGES THAT ARE NOT TAGGED IN THIS ORDER WILL AFFECT THE CORRESPONDING CSV FILE.***
Exporting Image Data
1. Once you have gone through all the images, it’s time to export the Metadata from all the images. Press Ctrl + A to select all images. Click on ‘Image’ → ‘Export Image Data.’
2. Navigate to:
   Z:\SNAMP_Cameras\Camera_Master_Data_Files_Year_04\SNAMP_SilentImage_CSV
   a. Summary_Files_SurveyYr_04\Created_from_Silent_Image_Unprocessed, and either find the folder for the camera you are summarizing, or make a new folder and save file name as: XXX_XXXX_CheckX. This name refers to the camera location grid cell and check number. Click Save.

Additional Notes:
- You can only view the information you tagged on an individual photo. If you are tagging several images simultaneously, the previously tagged information will not be visible although it is there (not lost or deleted.) Same for correcting data—although you can tag as a group, you can only correct an entry one at a time.
- Narrative: Use this tab in the Add/Edit Image Data box to explain any tagging that is unclear, or to provide information not generated through the tagging process. For example, you may get a rare species that is not listed in the SPECIES_EVENT dropdown menu. Select ‘Unknown’ under SPECIES_EVENT in this case, and use the ‘Narrative’ tab to illustrate the animal’s identity (Mountain Beaver!)

Update Active Camera Sheet
1) When you’ve finished tagging your images update the active camera sheet located at Z:\SNAMP_Cameras\Camera_Master Data Files\SNAMP_Active_Camera_Traps.xls
2) Enter the date you tagged the images and your initials in Column N.
Camera Image CSV File Processing Protocol

The following protocol is a description on how to use the CSV Template when creating CSV files. The protocol must be followed as described below to ensure run time errors are not encountered.

1. Create a temporary folder on the desktop labeled with your initials_CSV (JAM_CSV). You will use this folder for the duration of the time you will be creating and proofing CSV files. However, be sure to delete this folder once you have finished with CSV files for the day.

2. Unprocessed CSV files may be found in Z:\SNAMP_Cameras\Camera_Master_Data_Files_Year_##\SNAMP_SilentImage_CSV_Summary_Files_SurveyYr_##\Created_From_Silent_Image_Unprocessed.
   - Browse to the camera grid you will be creating the CSV files for and copy all checks.
   - Paste the selected checks into the temporary folder you have created on the desktop.
   - Once the files have been pasted into your temporary folder on the desktop rename all files so that the file names consist simply of Check1, Check2, etc. (IF THE FILES ARE NOT LABELED AS INSTRUCTED ABOVE YOU WILL ENCOUNTER A RUN TIME ERROR.)

3. Once the files have been renamed open each check file.

4. Open the CSV_Template excel file, found in Z:\SNAMP_Cameras\CSV_Template.
   - Please note the template has been set as a read only file, this is to ensure the template is not accidentally overwritten. When prompted to work in the file as read only select yes.

5. The CSV_Template file opens to display the CSV format check options. These options consist of how many checks you will be formatting and range from a single check to seven checks.

6. Before proceeding confirm all checks you will be working with have been renamed correctly and all check files are open.

7. Choose the CSV format # Check button that corresponds with how many check files you have.

8. Once the format completes confirm all checks are present, open each tab, confirm that the file is complete with correct check # and all data are present, and add the visit #’s.
   - PLEASE NOTE: IMAGES OF GOOD QUALITY ARE NOT LABELLED AS SUCH DURING THE TAGGING PROCESS AND ARE LEFT BLANK. IF AN ENTIRE CHECK HAS GOOD QUALITY PHOTOS YOU MUST MANUALLY ADD THE IMAGE_QUALITY COLUMN. (See below for proper column arrangement).
9. In the visit# column will be NEW VISIT to indicate a new visit for that species. While previously we would manually change the NEW VISIT to visit number 1,2, etc. **we no longer need to do this.** Leave the visit column as is and the NEW VISIT tags in place. Visit numbers will be generated when the processed CSV is added to the CSV master.

10. **WHAT THIS PROCESS DOES NOT DO IS ORGANIZE THE SPECIES, VISIT, QUALITY, NARRATIVE COLUMNS, BEAR, AND BEAR COLOR FOR YOU. ALSO, IT DOES NOT COPY THE CHECKS INTO THE PROCESSED FOR SUMMARY TAB.** BE SURE THE ABOVE MENTIONED COLUMNS ARE SORTED CORRECTLY AND THEN COPY EACH CHECK INTO THE PROCESSED FOR SUMMARY TAB. The correct arrangement of the columns should be:

1. Site
2. Cam Type
3. Image name
4. Image path
5. Moon phase
6. Temp
7. Check#
8. Date
9. Time
10. Species Event
11. Visit#
12. Image quality
13. Narrative
14. Bear (When applicable)
15. Bear Color (When applicable)

- Note that the bear and bear color columns will only be present if a bear was present at the check.
- If you encounter a situation where the columns are not sorted correctly be sure to follow the above column arrangement as well as **RE-SORTING THE ENTIRE CHECK.** This may be done by choosing on the excel tool bar, Home-Sort&Filter-Custom sort. When the sort dialog box opens choose my data has headers and then add a level. You will add several levels and first choose Species, then Date, and lastly Time to populate the column field. **IF YOU FIND THE NEED TO RESORT BE SURE TO CONFIRM THE VISIT# COLUMN IS CORRECT.**

11. Save the file as “###_####_CSV_Processed_For_Summary.xlsx” within the Z:\SNAMP_Cameras\Camera_Master_Data_Files_Year_03\SNAMP_SilentImage_CSV_Summary_Files_SurveyYr_03\Processed_For_Summary folder.
12. Please note that the check files you have opened for processing will close automatically once the template macro is finished formatting.
MEASURABLE FISHER IMAGE J MEASUREMENT PROTOCOL

This protocol is used to measure/extract various physical (morphological) measurements from fishers photographed at camera trap stations surveyed with Reconyx automatic digital cameras. Based on body measurements taken from captured study animals, it is possible to determine the sex of individual fisher based on several of the measurements we obtain from the camera photos. It may also be possible to determine individual identification (individually discriminate) fisher from the measurements.

Before we begin please familiarize with the list of standard measurements, measurable data entry files, and the ImageJ program, which may all be found below. Additional information for the ImageJ program may be found in Z:\SNAMP_Office\ImageJ.

Standard Morphological Measurements

There are up 9 standard measurements that will be taken for each “measureable fisher photo”, although the number of measurements possible for each photo will often be fewer.

IMPORTANT NOTE: you do not need to obtain all of the listed measurements for each photo; only take the measures that are possible based on the orientation/view of the fisher in the photograph.

List of Standard Measurements:

- Head length (Hd_Lght): straight line tip of nose to base of skull
- Ear tip to ear tip (Ear_Ear): measure the widest distance between the ears
- Neck width (Neck_W): distance across neck immediately behind base of ears, measuring the width of fur (from hair edge to hair edge).
- Head+body length (Body): tip of nose to end of body at base of tail
- Tail length (Tail): top of tail to base of tail not including tuft of hair at very end of tail
- Length of right and left forefeet (RFt_L or LFt_L): base of “wrist” to tip of foot
- Width of right and left forefeet (RFt_W or LFt_W): distance across back of forefoot not including the pollex or “thumb” (will often be extended apart from the other digits for gripping the tree)
- Forward edge of ear to forward edge of ear (Inner_ear): measure distance between where the ears meet the front of the head.

Measureable Data Entry Files

- To record the measurable results to the SNAMP database open the Excel SNAMP_MeasurableFisher_Database file located in the Camera_Master_Data_Files_Survey_Year_03 folder on the server.
- Begin by filling in ALL of the basic information for the photo(s) you will be measuring: Grid_ID, Check_No, Check_Date, F_Visit No, F_Type, Photo_No, Photo_Date, Photo_Time. Information on the fisher visit number (F_Visit_No) is available on the “Fisher_Visits_Detailed” worksheet in the SNAMP_Active_Camera_Traps_SurveyYr_0X file. Information on the date and time for the photo should be taken directly off the image – upper right corner, which may be obtained from the image you are working with.
• Move to the Excel program window with the already open file
  “SNAMP_MeasureableFisher_Database” and name and record your measurement.
• From this point you easily can proceed through all of the measurements that are possible
  for the photo you are measuring.

IMAGEJ Measureable Image Instructions
• We will have pre-selected the “Measureable Fisher Photos” from the fisher active grids
during a given Camera SurveyYear. Please use the worksheet titled
MeasurableFisher_Database_SurveyYr_0X” in the
“SNAMP_Active_Camera_Traps_SurveyYr_0X” file in the
“Camera_Images_Data_Survey_Year_0X” folder on the server to track your
contributions to measuring fisher body sizes.
• The camera images for all grids from a Camera Survey Year where fishers were detected
should include a folder titled “XXX_XXXX_Measureable_Fisher_Photos.” These
photos are the only ones we will use for taking measurements from the fisher active
survey grids.
• For your information and for future reference, fisher images that are considered
measureable are those for which (a) the photo includes a wooden slat with at least two of
the reflective tape markers easily visible, and (b) fishers are on the bait tree with
back/body/head/tail oriented mostly towards the camera. Also, the best quality
measurable fisher photos are those where the animal is stretched to its full length along
the tree, not bunched up or twisted to the side. In instances where multiple photos are of
quality to measure, select the best 5 photos, ensuring that as many of measurements listed
below (page 1) are obtained. NOTE: Some images may be selected in order to obtain a
good measure of one of just a few body features.
• IMAGEJ
• Open “ImageJ” from its location on the desktop. The ImageJ program will open and
consist simply of the ImageJ toolbar (File, Edit, Image, Process, etc.), illustrated below.

![ImageJ](image.png)

• **Before beginning any measurements be sure to update the Image J program to
  accommodate any upgrades that may be available.**
  a) On the toolbar navigate to Help - Update ImageJ.
To open the image you will be measuring choose File/Open, and browse to the desired image using the path: Z:\SNAMP_Cameras\Camera_Images_Survey_Year_XX\Measurable_Fisher_Database.

To open the desired image simply double click on it and a new, separate window will open. This new window containing the image will be the platform from which measurements will take place.

Before any measurements begin you must first set the scale for the measurable fisher image. (NOTE:*Scale must be set for each measurable image*)

  a) To perform this operation we will be using the Zoom (magnifying glass icon), Line (straight icon), and Set scale feature, located on the ImageJ toolbar.
Before adjusting scale, first suggest to zoom into the slat, then enlarge the measurable image, which may be done by either stretching the image vertically or horizontally.

To set the scale follow the steps below:

**ZOOM**

1. Zoom into the woodslat by choosing the magnifying glass icon found on the ImageJ toolbar. Once the zoom feature has been chosen the cursor will appear as an “iron cross” once placed over the image. To zoom in, left click on the section of the image (wood slat) you want enlarged, right clicking on the image will zoom out.
   a. Note that hot keys are also available once zoom mode is activated: ctrl + + (plus sign) zooms in, ctrl + - (minus sign) zooms out.

**STRAIGHT LINE**

During this step we will be creating a single line that stretches from the center of one piece of reflective tape to the center of the next consecutive piece of reflective tape.

a) On the ImageJ toolbar choose the straight line icon. When your cursor has been placed over the image it will appear as a light gray + sign.

By using the reflective strips that are most visible this will give you the most accurate measuring standard. Each reflective strip has a small amount of height, but the accurate measurement marking is in the middle of the strip’s height. Also, remember that the space between each reflective strip is 20 cm, or 0.2 m.

b) Single left click in the desired area and drag the cursor to your destination point. This will create a single, straight, yellow line on the measurable image. Illustrated below.

*Please note that the line may appear staggered, if so, straighten the line by slightly dragging the endpoint either to the left or right.*
• **SET SCALE**
  • Once you have established your straight line on the slat set the scale for the image.
    • On the ImageJ toolbar choose Analyze/Set Scale.
    • The Set Scale dialog box will open in a separate window. Enter the known distance of 0.2 (this is only if you have placed your line between two pieces of consecutive reflective tape, see above for further explanation).
    • Enter unit of length as **m**, indicating the unit of measurement as meters.
    • Once the known distance and unit of length fields have been entered click OK.

At this point the image is now ready to be measured and processed.

• **MEASURE**
  • Measuring is accomplished by using the Straight Line and Segmented Line feature. The straight line will only measure from the beginning point of your measurement to a single end point, such as when measuring **ear tip to ear tip**. The
segmented line has the ability to measure from the beginning point of the measurement to multiple end points. This becomes helpful when taking the head and body measurement. *Please note body length may be taken using the straight line feature by first measuring the head, record the measurement given, then by clicking on the lines point of origin, (tip of nose) then drag that point of origin to the base of the tail. The tail may be measured similarly by taking the point of the lines origin (base of tail) and dragging it to the tip of the tail.*

- The segmented line feature may be accessed by right clicking on the straight line icon, a drop down box will appear and from there you will be given several line options, including the segmented line.

- **NOTE:** When measuring the head length, it may be helpful to position a line across the back of the head, from behind each ear. The middle of this line will usually be very close to the back of the skull, depending upon how the head is tilted.

- Re-zoom and pan around the image as needed for obtaining all other measurements.

- The measurable results may be obtained from the ImageJ toolbar once you have placed your endpoint at the desired location. However, the cursor must not move once it is placed at the end location or the displayed results will no longer be available. You may also obtain the results by choosing ctrl + m, which will open the results dialog box. Once the data has been collected close the results box and do not save it.

- The final step is to rename the original image file in the Server folder from which it was originally obtained. If the original file name was
M0001102 it should be renamed to include your initials (M0001102_RAS) indicating that the photo has been measured.

- Once you are done obtaining all possible/reasonable standard measurements, close the image, File/Close.
DEN SITE LOCATION AND DEN MONITORING PROTOCOL

The purpose of this protocol is to outline the methods we will use to search for and identify the den structures used by reproductive-aged female fishers. The methods primarily involve ground-based telemetry and “walk-ins” on female fisher in the early morning and, less frequently, early evening hours.

Equipment needed for Den Site Walk-ins:

a. Telemetry receiver + collapsible Yagi antenna + coaxial cable
b. Blue telemetry receiver headset
c. Garmin GPS unit
d. Pentax camera
e. Den site data forms: Den/Rest Site Form, Den Check and Walk-in Monitoring Form + Den Camera Check Form
f. Flagging
g. Truck omni (if travelling by truck)

Finding a new den or rest site

1. Individuals involved in performing aerial radio-telemetry will provide information on the locations of adult female fishers during the early morning period between 7:00 and 8:00 am. Adult females will be top priority for locations during late March and early April for all telemetry flights.

2. Once aerial locations are acquired for the targeted adult females for the day, the information will be communicated by radio to the ground crew waiting to perform walk-ins. It is critical that this location information is given as “Location reference” with no reference to a possible den structure. In addition, the numbers broadcast over the radio will be limited to the “LAT  mm.mm” and “LONG mm.mm” without the degrees [always 37° (LAT) and 119° (LONG)]. To further protect the security of fisher den locations while broadcasting over the USFS radio, the last four digits of each Latitude and Longitude (minutes and fraction of minutes) should be transposed or reversed. (For example, the aerial readout is 37.42.18 and 119.54.68. The aerial technician should report these digits to the ground crew: 18.42 and 68.54.)

3. Ground crews will need to temporarily change the “Units” under the Setup menu for their Garmin GPS units for inputting the Lat/Long data as UTM waypoints. In the Units directory, highlight and open the drop-down menu for the “Position Format.” Once this menu is visible, toggle down and select the “hddd˚ mm.mmm’ format (This is degrees, minutes, and fraction of minutes.) Exit out of the menu and enter the LAT, LONG numbers provided from the airplane (remember that they are reversed!) (In the above example, the ground crew would enter 37.42.180 and 119.54.680. A zero is added on to the fraction of minutes.) Once the numbers have been entered, return to the Setup then Units menu and change the Position Format back to “UTM UPS”.

4. The ground crew should now be in position to walk-in on the predetermined fisher, or can reach the location obtained via aerial-telemetry quickly. Once a strong signal is obtained on the ground (i.e., several bars flashing on the receiver), “walk-in” toward the animal.
**Helpful Telemetry Hints:**

- Make sure the gain is turned all the way up when first trying to listen for the animal. If the animal is far away and the gain is too low, you may miss it. Once you get a signal, adjust the gain lower (and volume louder) as you get closer. This will help in pinpointing the animal. **When you think you are getting close (within a few hundred meters), put on your blue headset to minimize noise disturbance to the animal.**

- Position of the yagi can greatly affect signal strength and direction. A vertical yagi is good for hearing a signal from a great distance (use this position if the animal seems far away (i.e., only one or very few bars flashing in the receiver). Again, make sure the gain is turned all the way up. A horizontal yagi is good for ascertaining direction, especially when getting close to the animal. Using both positions during a walk-in can greatly assist the speed and precision in which you locate the animal. Try to keep the yagi in the same plane when switching from vertical to horizontal positions (i.e., the yagi is not a magic wand.)

- Signal bounce can make ascertaining direction of a signal confusing. Try using both vertical and horizontal yagi positions. If this doesn’t help, try changing position yourself. If you or the animal is near a large granite outcrop or steep canyon, you may have to do some walking around to get into a better position.

5. Once you think you have located the animal in a tree, confirm by circumnavigating the proposed tree using your telemetry gear. Flag a nearby tree (NOT the actual den tree) for future reference. Use your pentax camera to take several reference photos from differing angles. Take a waypoint with your GPS and fill out a new den/rest site data sheet. Although these data are important, completion of this form should be done quickly as to minimize further disturbance to the animal. If this is a new den or rest site and you are finding the animal for the first time at this location, the den/rest site form and the den check monitoring form are all the data forms that are needed.

**Den Camera Set-Up and Checks**

Confirmation of a new fisher den site usually happens the day after the initial location and is performed during an early morning walk-in. **Special circumstances:** Denning females that are logistically difficult to visit frequently are an exception to this rule. In addition to the telemetry equipment needed for Den Site Walk-ins (see page 1), a photoviewer, 2-3 Reconyx cameras, and the Den Camera Check Data Sheet are needed. Den camera checks and walk-ins are to be conducted every 3 days.

1. Before heading out, take a look at the reference photos your fellow technician took of the suspected den tree you will be confirming.

2. Set up 2-3 Reconyx cameras focusing on the base of the den tree. **NOTE** All denning female fisher are assigned 2-3 permanent Reconyx cameras that will remain with them throughout the denning season. Be sure to get adequate ground coverage at the base of the tree. Cameras should be placed 5-8 meters away. If using the new style Reconyx cameras, make sure the camera is programmed to “rapid fire.” Double check to make sure date and time are correct.
3. When checking Den cameras, if the fisher is in the den tree or close by, you need to act swiftly and silently to minimize disturbance. Use your photoviewer to quickly download the photos. Save copies of the photos from each of the den tree cameras onto the photoviewer. Create new folders in the photoviewer and label with Fisher ID, UCB, and date. (Example: F01_UCB_056_20090406). Once you are a good distance away from the den tree or at your vehicle, scroll through the photos for each den camera and note date and times of fisher activity, if any.

4. (Quickly) fill out the Den Camera Check Data Sheet. Alternatively, complete the data form when you’ve gotten back to your vehicle, or somewhere away from the den tree to minimize disturbance.

5. Mating typically occurs about 1 week after females give birth (late March-early April), so male fishers will frequently be around natal den trees in early-mid April. When at natal den trees you should also scan through the frequencies for adult males and look around. If you hear or see any other fishers in/near natal den trees note this on the data form.

6. If the fisher is not in the suspected den tree, continue to locate her using telemetry to pinpoint her new location. Mark her new location with GPS and flagging. If the animal is located in a new tree, (quickly) fill out a new den/rest site data form. If it is suspected you have “bumped” her off her den tree or that her signal indicates she is active and on the move, take a general waypoint and leave the area. DO NOT continue searching for a fisher on the move. Fill out the Den Check Monitoring Log Data Sheet.

Naming Den Trees
A female fisher will change dens several times throughout the denning season. The female is thought to spend a longer period of time (2-3 weeks) at her first or ‘natal’ den. The Natal den is where her kits are born. Over the next two months, she’ll very likely move her kits to a different den every 7-10 days. These secondary dens are ‘maternal’ dens. The female will spend more time foraging as the kits get older and have better thermoregulation while inhabiting the maternal dens. Label the dens Natal, Maternal 1, Maternal 2, Maternal 3 and so on.

Moving Den Cameras
Den cameras are to be moved ONLY after a den confirmation walk-in has been performed. A confirmation walk-in is performed the day after finding a suspected new den in the early morning hours. It is unlikely that new dens will be more than a kilometer from the old den as the female will have to transfer her kits over this distance. After confirming the new den site, return to the previous den site, remove the cameras, and set-up at the new site using the Den Camera Set-Up protocol above.

Office Data Entry:
- Update the Fisher_Denning_Database_Master_20XX (Z:\SNAMP_Fisher_Denning_Kit_Data\Denning Season 2011\Den_Season_2011_Master_Data_Files). There are three different worksheets in this spreadsheet that correspond to our three datasheets. If you found a new den site, enter your information into the “Fisher Den Site Data 2011” worksheet. For a walk-in, the “Den Check
Monitor Log 2011” will need to be updated. If you installed or checked cameras, open the “Den Cam Check Log 2011” worksheet and update your new information.

- If you installed or moved den cameras, open the “Den_Camera_Images_2011” folder located on the main server (Z:\SNAMP_Fisher_Denning_Kit_Data\Denning Season 2011). Open the folder for your denning female and create a folder for the den tree type (i.e., Natal, Maternal 1, Maternal 2, etc.). Within that folder create another folder named “Reference Photos” and upload your reference images there.

- If you checked den cameras, open “Den_Camera_Images_2011” folder and find the female’s folder you have camera images of. Open the folder for your den tree type (i.e., Natal, Maternal 1, Maternal 2, etc.). Create a new folder for your check (simply Check 1_Date) and create a new folder for each UCB camera inside of this. Label it with UCB_XXX. Transfer the images to the new folders you have created.

- Even if your cameras did not have any photos during the check, make sure you still create an empty folder for each camera you visited and add “No photos” to the end of the label (Example: UCB_024_NoPhotos).

- Open the Active Camera Traps Year 4 spreadsheet. There are two different worksheets that are present during the denning season only. Enter your walk-in information into the “Fisher Den Walk-in Schedule” worksheet. Add three days to the “Next Check Date”. Open the “Den Cams” worksheet and enter the information from Reconyx den cams you setup, moved or checked. Den cams should be checked every 3 days as well.
PROTOCOL FOR ASSESSING HABITAT/VEGETATION CHARACTERISTICS AT FISHER DEN TREES

The purpose of this protocol is to begin to understand the combination of biotic and abiotic characteristics female fishers are likely selecting/using for denning habitats. Our goal is to collect similar types of data as those being recorded by the Forest Health Team on the Core Plots in the Sugar Pine area, while also capturing the same types of data being recorded by the Kings River Fisher Project at their den trees.

For each natal and maternal den tree we will define a circular plot centered on the den tree with a fixed radius of 18 meters (area ≈ 1 hectare), represented by the diagram below. Each circular plot will include four 18 meter transects, one each oriented N (0/360°), E(90°), S (180°), and W (270°), originating from small nails in the base of each den tree (removed at completion of plot). We will next define four 4m X 18m belt transects as the sampling space extending 2m from either side of each 18 meter line transect. Finally, within the circular plot centered on the den tree, we will also define 4 plot sections ordered A, B, C, and D clockwise from the N transect.

**Figure D1.1:** Diagram illustrating the layout of circular plots and associated measurement transects, including 4 quadrats A, B, C, and D.
Assessing/Measuring Canopy Cover: Canopy cover will be measured at the 2m, 6m, 10m, 14m, and 18m positions along each of the four 18 meter transects using a sighting tube device called a “moosehorn coverscope.” At each sample point hold the coverscope to your eye, sighting as you would a periscope, center the bubble in the center of the bullseye level, then maintain the coverscope plumb and level while counting the number of line intersections on the grid obscured by forest canopy. NOTE: the grid is viewed as being crosshairs, and the number of squares that are open or obscured is unimportant. Also, there are a possible 25 possible hits or intersections, including the outside intersections at any one sampling point. See Figure D3.

Figure D1.2: Diagram illustrating multiple types of measurements to be taken along the four 18 meter line transects.
Assessment of Litter, Duff and Fuel Height: We will measure the thickness of the duff and litter layers and fuel height at several positions along each 18m transect. Measurements of the duff layer, litter layer, herbaceous vegetation, and fuel height will be done at the 2m, 10m, and 18m positions.

Height of herbaceous vegetation (herbs or grasses), if present within a 10 cm radius of the point, should be measured first, prior to excavating holes for measuring the duff and litter layers. Use a hand trowel to dig three small vertical holes down through the litter+duff to mineral soil at 2m and 10m and 18m along each 18m transect. If a tree of stump occurs at the position, offset your digging 30cm to the right.

Measure the thickness of the duff (± 0.5cm) from the mineral soil to top of duff, and the litter layer (± 0.5cm) from the duff to top of litter, not including woody debris (branches/sticks/logs). Fuel measurements will start at the bottom of the litter layer and end at the top of the tallest fuel up to 1.83m (6 feet). Fuels are any woody twig (> 0.64 cm diameter), branch, or log that is severed from the original source of growth with its central axis lying above the duff layer.

**Figure D1.3:** Overview of using the moosehorn coverscope; the coverscope should be held with the longest length perpendicular to the ground. The small side tube should be held up to the observer’s eye with the bubble level. The user will then count the number of intersections which are covered by canopy. The lower left photo points out what the intersections look like (corners, side intersections, middle intersections). The diagram on the lower right provides an example of what canopy cover might look like on the grid. In this example, the count would be 10 out of a total of 25.
(excluding needles, grass, bark or pine cones). Basically, if the fuel object isn't more than 1/2 buried in the duff, its height up to 1.8 m/6 feet will be measured. If no fuel exists, record the height of the litter at the point.

**Figure D1.4**: Diagram illustrating how to properly discriminate between the litter and duff layers for taking measurements.

- Duff thickness (mineral soil to top of duff; (see diagram)
- Litter layer thickness (duff to top of litter; ± 0.5cm) (see diagram)
- Fuel height (bottom or litter layer to top of tallest fuel up to 1.8m/6 feet); if no fuel at point record the depth of the litter layer
- Herbaceous/grass vegetation height (nearest 1 cm)

**Shrub level vertical cover** (i.e., concealment): Shrub level vertical cover will be measured using a cover-board/drop-cloth design. As we are concerned with concealment in general, not just shrub cover or foliage density, anything that provides potential cover to the fisher as it is coming or going from the tree should be counted (e.g., tree trunks, sapling foliage, shrub cover, boulders). The drop-cloth is 3 m x 0.5 m and is composed of 0.1 x 0.1 m squares separated into 4 categories: 0-0.3 m (15 squares, low ground), 0.3-1m (35 squares, high ground), 1-2m (50 squares, low shrub), and 2-3 m (50 squares, high shrub). One observer stands with their back to the den tree while a second person holds the cloth at the 10 m position along each transect. The observer counts the number of squares within each height interval at least 50% obscured by ‘cover’ and records this number. The observer should squat while reading the two lowest sections (low ground and high ground) and stand while reading the upper sections (low shrub and high shrub). The technician holding the cover-board should try to hold it as straight and steady as possible, taking care to make sure the bottom dowel is hitting the ground and that the cloth is taut in all directions.
Measuring Topographical/Landscape Features: The prevailing aspect for the slope upon which the den tree is located will be measured using a clinometer. Walk to a position on the slope adjacent to the den tree with your back to the hillside behind you. Use the clinometer to estimate the prevailing aspect (Hint: if you spilled your water, which way would it flow?). Slope will also be measured for the hillside upon which the tree is located. Use a clinometer to take two measures of slope, which will be averaged for the recorded measurement on the dataform. To obtain an appropriate measure of slope look directly uphill and locate a tree or object about 15-25 meters away. Estimate where your eye level would be on that tree/object (around 1.5 m), look through the viewfinder on the clinometer and read the % slope on the right hand side.

Figure D1.5: Design and use of coverboard for estimating concealment cover around the base of den trees. The coverboard is divided into four sections representing low ground cover, high ground cover, low shrub cover, and high shrub/small tree cover.
Assessment of Sizes, Numbers of Trees and Snags Within Circular Plot: We will enumerate and measure all medium and large size trees and snags (see below) within each area of the circular plot. All medium and large size trees will be characterized by Vigor Class and Crown Class.

Trees and snags will be grouped into three size classes for measurements:
- Size class 1: greater than or equal to 19.5cm Dbh
- Size class 2: 5.0cm to 19.4cm dbh
- Size class 3: ≤ 5.0cm dbh

Live trees and snags are further be broken down into 6 vigor classes as follows:

Live trees:
- Class 1: healthy tree with no visible defects
- Class 2: healthy trees with minimal damage or defects (broken/dead tops, abnormal lean, etc.)
- Class 3: live trees that appear near death or likely to die within 5 years

Dead trees
- Class 4: a recently dead tree with little decay (retains bark, branches, top, even some needles)
- Class 5: Tree showing some decay including loss of some bark, broken off branches/top
- Class 6: tree shows extensive decay including loss of most bark, branches, broken top

Figure D1.6: Illustration of multiple different possible vigor classes for trees and snags. For our habitat assessments, vigor class 3 will be trees that are near death, vigor class 4 will be recently dead snags, vigor class 5 snags are those with loss of limbs and loose bark (Figure stages 4-5), and vigor class 6 snags are those with broken tops, few if any limbs and visibly decomposed (Figure stages 6-7).

Live trees only will be characterized by Crown Class as follows:
- D (Dominate): trees with crowns extending above the general level of the crown cover and receiving full light from above and some from the sides
- CD (Co-dominate): trees with crowns forming the general level of the crown cover and receiving full light from above, but little light from the sides
- **I (Intermediate):** trees that are shorter than the class D and CD trees but with crowns either below or extending into the above crown cover and receiving little direct light from above or the sides
- **S (Suppressed):** trees with crowns entirely below the general level of the crown cover, receiving no direct light from above or the sides

**Figure D1.7:**
Diagram illustrating trees of different crown classes, and associated crown height measurements.

**Trees/Snags within Size Class 1 and 2:** We will measure the straight line distance from the den tree to every tree or snag within Size Class 1 and 2 in each of the four quadrats (A, B, C, D) of the circular plot around the den tree. In addition, we will take a bearing from the den tree to each of the Size Class 1 and 2 trees in order to create a stem map once back in the office. After measuring the distance (± 5 cm) and bearing to each of these trees/snags, record/assess the vigor class, crown class (live trees only), height to live crown base (live trees only) and total tree/snag height for each of these trees/snags.

**Small Trees/Snags (Tree Size Class 3):** All live and standing dead trees less than 5.0cm dbh and a minimum height of 1.37 meters will be counted in each of the four quadrats around the den tree. Track on the dataform the live/dead status of these trees.

**Assessment of Woody Shrubs:** Woody shrubs will be sampled along each of the four 18m transects using the line-intersect technique. For all woody shrubs that occur along and intersect the transect line record/measure:
- Shrub species (see species list below)
- Length of shrub intersect (nearest 10cm)
- Average height (± 5cm)
- We will also record an ocular estimate of overall shrub cover by species within each of the 4 quadrats

**Assessment of Cover by Herbaceous Plants:** In addition to measuring the height of herbaceous cover (± 1cm) at the 2m, 10m, and 18m positions along the four line transects, we will also assess and record an ocular estimate of the percent cover of herbs and grasses combined in each of the four quadrats around the den tree.

**Assessment of Course Woody Debris (CWD):** Course woody debris is defined as dead tree boles, large limbs, and other large wood pieces either lying on the ground or elevated off the ground up to 45°, but no longer supported by roots (i.e., dead trees hung up or leaning on other vegetation). CWD does not include live material, standing dead trees, stumps, dead foliage, separated bark, non-woody
pieces, roots, or the part of the bole below the root collar. We will assess/measure CWD along two of the four 4 X 18m belt transects, either transects 1 and 3, or transects 2 and 4. Use a coin flip to chose between Transects 1 and 2; if Transect 1 was selected then CWD will be assessed/measured along Transects 1 and 3, whereas if Transect 2 was selected then CWD will be assessed along Transects 2 and 4. Each piece of CWD sampled within the belt transects must have a large end diameter (LED) of 15cm and be at least 1m in length. Core variables to be measured include:

- Species (if determinable)
- Small end diameter (SED) in cm
- Large end diameter (LED) in cm
- Total length in meters to nearest cm
- Length of CWD within transect in meters to nearest cm
- Whether or not the midpoint of the CWD object fell within the belt transect
- Decay class of the CWD object (see Table below)

**Other sampling rules for CWD pieces and logs:**

- If a log is partially suspended by other logs or tipped against other trees, measure only the portion of the log that is within 2m of the ground.
- Pieces that are tipped must have an angle < 45˚with the ground to qualify as CWD.
- For logs with their root wad still attached, the large-end diameter (LED) is measured just above the butt swell, but the length is taken to extend into the mass of wood within root wad.
- When a tree is forked or has a very large branch attached to the main bole and both segments intersect the transect, they are tallied as two separate pieces (see figure below), if each meets the required minimum dimensions. Forked trees are examined to identify one fork as the main bole by measuring both diameters at the fork location. The forked segment with the largest diameter is considered the main bole and the length is measured from the tip of the fork to the end of the log. The smaller segment is recorded as a second piece with a length measured from the fork tip down to the point where the fork joins the main bole.

**Table D1.1: Overview and definitions of five Course Woody Debris (CWD) decay classes based on structure integrity, texture and overall condition.**

<table>
<thead>
<tr>
<th>Decay Class</th>
<th>Structural Integrity</th>
<th>Wood Texture</th>
<th>Condition of branches, twigs, and bark.</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>Sound</td>
<td>Intact, no rot.</td>
<td>Branches, twigs, and needles still attached with tight bark. Log solid.</td>
</tr>
<tr>
<td>2</td>
<td>Heartwood sound; sapwood somewhat decayed.</td>
<td>Mostly intact; sapwood soft-starting to decay. Wood cannot be pulled apart by hand.</td>
<td>Branches present, fine twigs or needles gone. Loose or peeling bark, 75-100% remaining.</td>
</tr>
<tr>
<td>3</td>
<td>Heartwood sound; log supports its weight. Sapwood decaying.</td>
<td>Large hard pieces of sapwood can be pulled apart by hand.</td>
<td>Branches not present; stubs will not pull out. Bark loose but 50-75% remaining.</td>
</tr>
<tr>
<td>4</td>
<td>Heartwood rotten; does not support its weight but shape maintained.</td>
<td>Soft, small blocky pieces can be pulled apart.</td>
<td>Branch stubs pull out easily. 0-50% of bark remaining.</td>
</tr>
<tr>
<td>5</td>
<td>No structural integrity. Log circumference flattened.</td>
<td>Soft and powdery when dry.</td>
<td>Bark gone.</td>
</tr>
</tbody>
</table>
Figure D1.8:
Illustration for how to count and measure branched logs during CWD assessments.
### Table D1.2: List of the species of conifers, hardwoods, and woody shrubs present in our study area and around den tree structures.

<table>
<thead>
<tr>
<th>Type and common name</th>
<th>Genus and species</th>
<th>Data code</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Conifer trees</strong></td>
<td></td>
<td></td>
</tr>
<tr>
<td>White fir</td>
<td><em>Abies concolor</em></td>
<td>ABCO</td>
</tr>
<tr>
<td>Incense cedar</td>
<td><em>Calocedrus decurrens</em></td>
<td>CADE</td>
</tr>
<tr>
<td>Sugar pine</td>
<td><em>Pinus lambertiana</em></td>
<td>PILA</td>
</tr>
<tr>
<td>Douglas fir</td>
<td><em>Pseudotsuga menziesii</em></td>
<td>PSME</td>
</tr>
<tr>
<td>Jeffrey pine</td>
<td><em>Pinus jeffreyi</em></td>
<td>PIJE</td>
</tr>
<tr>
<td>Ponderosa pine</td>
<td><em>Pinus ponderosa</em></td>
<td>PIPO</td>
</tr>
<tr>
<td>Western white pine</td>
<td><em>Pinus monticola</em></td>
<td>PIMO</td>
</tr>
<tr>
<td>Giant sequoia</td>
<td><em>Sequoiadendron giganteum</em></td>
<td>SEGI</td>
</tr>
<tr>
<td>Red fir</td>
<td><em>Abies magnifica</em></td>
<td>ABMA</td>
</tr>
<tr>
<td>Lodgepole pine</td>
<td><em>Pinus contorta</em></td>
<td>PICO</td>
</tr>
<tr>
<td>Gray pine</td>
<td><em>Pinus sabiniana</em></td>
<td>PISA</td>
</tr>
<tr>
<td><strong>Hardwood trees</strong></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Black oak</td>
<td><em>Quercus kelloggii</em></td>
<td>QUKE</td>
</tr>
<tr>
<td>Canyon oak</td>
<td><em>Quercus chrysolepis</em></td>
<td>QUCH</td>
</tr>
<tr>
<td>Live oak (uncommon)</td>
<td><em>Quercus spp.</em></td>
<td>QUSP</td>
</tr>
<tr>
<td>Tan oak (uncommon)</td>
<td><em>Lithocarpus densiflorus</em></td>
<td>LIDE</td>
</tr>
<tr>
<td>White alder</td>
<td><em>Alnus rhombifolia</em></td>
<td>ALRH</td>
</tr>
<tr>
<td>Mountain dogwood</td>
<td><em>Cornus nuttallii</em></td>
<td>CONU</td>
</tr>
<tr>
<td><strong>Woody shrubs</strong></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Pine mad Manzanita</td>
<td><em>Arctostaphylos nevadensis</em></td>
<td>ARNE</td>
</tr>
<tr>
<td>Greenleaf Manzanita</td>
<td><em>Arctostaphylos patula</em></td>
<td>ARPA</td>
</tr>
<tr>
<td>Birchleaf mountain mahogany</td>
<td><em>Cercocarpus betuloides</em></td>
<td>CEBE</td>
</tr>
<tr>
<td>Mountain whitethorn</td>
<td><em>Ceanothus cordulatus</em></td>
<td>CEBE</td>
</tr>
<tr>
<td>Buckbrush</td>
<td><em>Ceanothus cuneatus</em></td>
<td>CECU</td>
</tr>
<tr>
<td>Deerbrush</td>
<td><em>Ceanothus integerrimus</em></td>
<td>CEIN</td>
</tr>
<tr>
<td>Mahala mat</td>
<td><em>Ceanothus prostratus</em></td>
<td>CEPR</td>
</tr>
<tr>
<td>Mountain misery</td>
<td><em>Chamaebatia foliolosa</em></td>
<td>CHFO</td>
</tr>
<tr>
<td>Bush chinquapin</td>
<td><em>Chrysolepis sempervirens</em></td>
<td>CHSE</td>
</tr>
<tr>
<td>Bitter cherry</td>
<td><em>Prunus emarginata</em></td>
<td>PREM</td>
</tr>
<tr>
<td>Huckleberry oak</td>
<td><em>Quercus vaccinifolia</em></td>
<td>QUVA</td>
</tr>
<tr>
<td>Western azalea</td>
<td><em>Rhododendron occidentale</em></td>
<td>RHOC</td>
</tr>
<tr>
<td>Currant</td>
<td><em>Ribes spp.</em></td>
<td>Ribes</td>
</tr>
<tr>
<td>California rose</td>
<td><em>Rosa californica</em></td>
<td>ROCA</td>
</tr>
<tr>
<td>Thimbleberry</td>
<td><em>Rubus parviflorus</em></td>
<td>RUPA</td>
</tr>
<tr>
<td>Blackberry</td>
<td><em>Rubus ursinus</em></td>
<td>RUUR</td>
</tr>
<tr>
<td>Willow</td>
<td><em>Salix spp.</em></td>
<td>Salix</td>
</tr>
<tr>
<td>Snowberry</td>
<td><em>Symphoricarpus mollis</em></td>
<td>SYMO</td>
</tr>
</tbody>
</table>
Definition/Details on how to measure Dbh (diameter at breast height) in different situations

Dbh is outside bark diameter at 4.5 feet above the forest floor on the uphill side of the tree. To determine breast height, the forest floor includes the duff layer that may be present, but does not include unincorporated woody debris that may rise above the ground line. If a dead tree (snag) is missing bark, measure the Dbh without the bark and record that measurement.

Forked tree: In order to qualify as a fork, the stem in question must be at least 1/3 the diameter of the main stem and must branch out from the main stem at an angle of 45 degrees or less. Forks originate at the point on the bole where the piths intersect. Forked trees are handled differently depending on whether the fork originates above or below 4.5 feet.

Trees forked below 4.5 feet are treated as distinctly separate trees. Dbh is measured for each stem at 4.5 ft above the ground.

Trees forked at or above 4.5 feet count as one tree. If a fork occurs at or immediately above 4.5 ft, measure diameter below the fork just beneath any swelling that would inflate Dbh.

Stump sprouts originate between ground level and 4.5 ft on the boles of trees that have died or been cut. Stump sprouts are handled the same as forked trees, with the exception that stump sprouts are not required to be 1/3 the diameter of the dead bole. Stump sprouts originating below 1.0 ft are measured at 4.5 ft from ground line. For multi-stemmed woodland species, treat all new sprouts as part of the same new tree.

Tree with irregularities at Dbh: On trees with swellings, bumps, depressions, and branches at DBH, diameter will be measured immediately above the irregularity at the place it ceases to affect normal stem form. If this is not possible, because of the vertical extent of the irregularity, then adjust the Dbh measurement to better reflect the diameter of a regular bole.

Tree on slope: Measure diameter at 4.5 ft from the ground along the bole on the uphill side of the tree.

Leaning tree: Measure diameter at 4.5 ft from the ground along the bole.

Independent trees that grow together: If two or more independent stems have grown together at or above the point of DBH, continue to treat them as separate trees.

Missing wood or bark: If 50% or more of the circumference of the bole is intact, reconstruct the diameter at Dbh.
1.1.1 Figures Illustrating the Proper Use of a Diameter Tape

Correct Method

Press the tape firmly against the tree. Do not pull it out at a tangent to the tree at the point of measurement.

End of tape (with the '0' mark or hook) crossed under.

Correct

Incorrect

Tape must be at right angles to lean of tree. Do not place tape at abnormal location on bole of tree.

1.1.2 Point of Measurement for DBH

DBH

4.5'

Tree on slope

DBH

4.5'

Tree on level ground

Diameter Point

4.5'

Tree deformed at DBH by swelling or crook. Take DBH above deformation.
Diameter Point

Tree with branch at 4.5 feet

Windthrown tree

Diameter Point

Leaning tree

DBH

1.5'

3' or more

Bottleneck tree

Adjust diameter tape to normally rounded position to allow for the missing catface portion

- If you can see light between the two stems, at DBH, measure as two separate trees.

- If you cannot see light between the two stems, at DBH, measure as one tree.
Tree forked at 4.5 feet or higher. Record as one tree and consider only the main fork. Take DBH below the swell of the fork.