United States
Department of
Agriculture

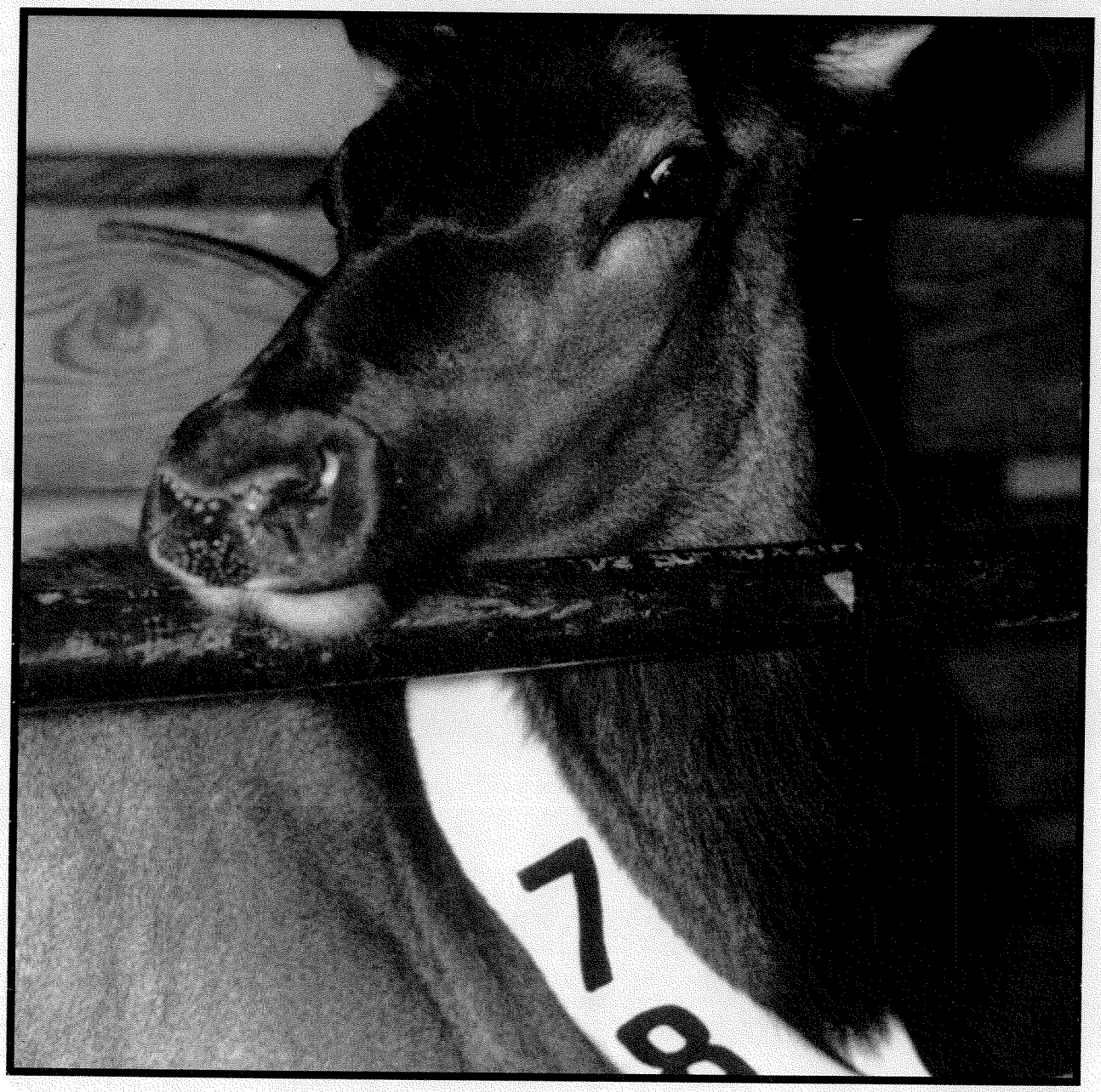
Forest Service

Pacific Northwest
Research Station

General Technical
Report
PNW-GTR-311
June 1993

Protocols for Care and Handling of Deer and Elk at the Starkey Experimental Forest and Range

Michael J. Wisdom, John G. Cook, Mary M. Rowland,
and James H. Noyes



Authors

MICHAEL J. WISDOM is a wildlife biologist, U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850; JOHN G. COOK is a wildlife biologist, National Council of the Paper Industry for Air and Stream Improvement, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850; MARY M. ROWLAND is a consulting wildlife biologist, 401 Aquarius Way, La Grande OR 97850; and JAMES H. NOYES is a wildlife biologist, Oregon Department of Fish and Wildlife, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

Abstract

Wisdom, Michael J.; Cook, John G.; Rowland, Mary M.; Noyes, James H. 1993. Protocols for care and handling of deer and elk at the Starkey Experimental Forest and Range. Gen. Tech. Rep. PNW-GTR-311. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 49 p.

Several hundred Rocky Mountain elk (*Cervus elaphus nelsoni* V. Bailey) and Rocky Mountain mule deer (*Odocoileus hemionus hemionus* Rafinesque) inhabit a fenced, 25,000-acre enclosure at the Starkey Experimental Forest and Range in the Blue Mountains of northeast Oregon. Research there requires handling most of these animals each winter. In addition, 33 elk calves have been captured and raised for research. Protocols for care and handling of deer and elk are described. Legal requirements for the operation of facilities and research within the enclosure also are discussed.

Keywords: Elk, mule deer, animal welfare, Starkey Experimental Forest and Range, Blue Mountains (Oregon), tame elk.

Contents

1 **Overview**

1 **Introduction**

1 The Setting

1 The Starkey Research Project

6 Animal Welfare Act of 1985

6 Research Activities Covered or Exempted by Law

8 Legal Requirements and Process

10 Registration of Research Facilities

10 Exempt Institutions

11 **Care and Handling of Animals**

11 Assumptions

14 Protocols for Public and Resource Management Activities

15 Protocols for Handling Deer

20 Protocols for Handling Elk

34 Other Protocols

36 **Functions of the Institutional Animal Care and Use Committee**

36 Membership

36 Responsibilities and Legal Authority

36 Biannual Meeting and Inspection

36 Annual Report to APHIS

37 Courtesy Inspections by APHIS

37 **Acknowledgments**

37 **Literature Cited**

39 **Appendix 1: Justification and Protocols for Studies on Forest Cover-Elk Energetics Relations in the Blue Mountains of Northeast Oregon**

46 **Appendix 2: Definition of Terms**

Overview

Protocols outlined in this document describe the humane and safe treatment of elk and mule deer used for research at the Starkey Experimental Forest and Range near La Grande, Oregon. Research there includes field studies of deer and elk that require seasonal care and handling of animals. Partners in the research include the Oregon Department of Fish and Wildlife (ODFW), the Pacific Northwest Research Station of the Forest Service (FS), the National Council of the Paper Industry for Air and Stream Improvement (NCASI), and Oregon State University (OSU). Research began in 1989 and will continue for 10 years or more.

These protocols prescribe treatments that fully comply with the Animal Welfare Act of 1966, as amended in 1985 (U.S. Laws, Statutes, etc. 1985) and administered under 1989 regulations promulgated by the U.S. Department of Agriculture (USDA) Animal and Plant Health Inspection Service (APHIS). The protocols are designed to ensure that animals are treated humanely, safely, and with minimal stress.

Most of the safeguards and procedures described here are self-imposed by the researchers and not required by law. Rather, they were adopted as the most prudent process for meeting both "the letter and the intent" of the Animal Welfare Act. These protocols demonstrate the care, concern, and commitment of scientists to the maintenance of healthy research animals in all phases of study at the Starkey Experimental Forest and Range.

This document primarily emphasizes protocols used for the four studies developed during early planning of the Starkey Project. Additional, cooperative studies between the FS, ODFW, and several private and public institutions have been and will continue to be conducted. This document does not provide guidelines for all future projects. Such projects will be assessed by the Institutional Animal Care and Use Committee (IACUC), and justification and procedures for them will be appended to this document. Justification and protocols for the first of these additional projects, the elk-thermal cover study cooperatively conducted by the FS and a private research organization (NCASI), have been attached in appendix 1.

Introduction

The Setting

The Starkey Experimental Forest and Range is 28 miles southwest of La Grande, Oregon, within the Wallowa-Whitman National Forest (fig. 1). The 25,000-acre area was dedicated for research by the FS in 1940. Since then, it has been the site of many range and wildlife studies (Skovlin 1991). Today, Starkey supports intensive research on both game and nongame wildlife, specifically in relation to management of cattle, timber, roads, recreation, and other human activities (Johnson and others 1991, Thomas 1989).

Geology, soils, physiography, climate, and vegetation of the area, described by Strickler (1965), are typical of the Blue Mountains of northeast Oregon (Franklin and Dyrness 1973). Bull and Wisdom (1992) list fauna of the area.

The Starkey Research Project

In 1987, the Starkey Experimental Forest and Range changed dramatically, when about 25,000 acres of the research area were enclosed by an 8-foot New Zealand fence (fig. 2).[1] Additional fencing subdivided the area into three parts: the main study area of 20,673 acres; the northeast study area (intensive timber management area)

[1] Bryant, Larry D.; Thomas, Jack W.; Rowland, Mary M. 1992. Techniques to construct New Zealand elk-proof fence. 34 p. Unpublished manuscript. On file with: Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

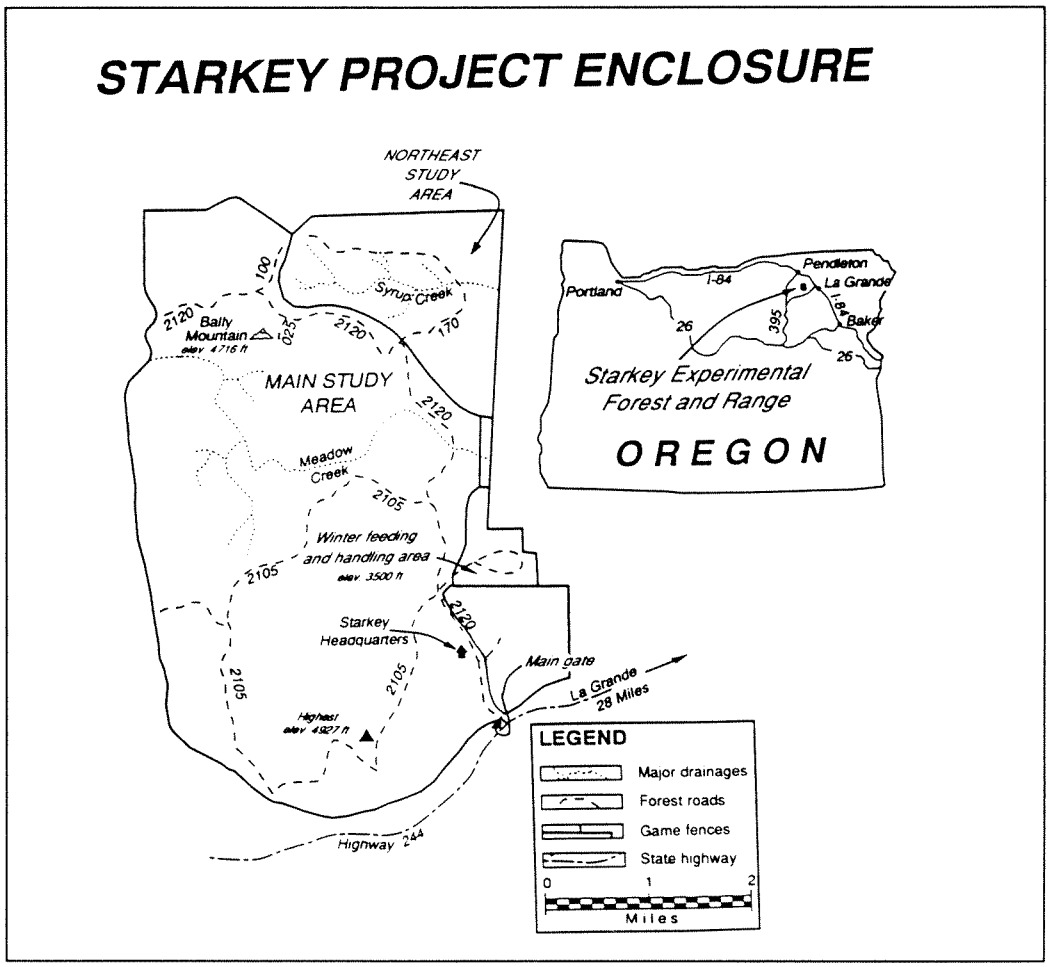


Figure 1—Location of Starkey Experimental Forest and Range and its 25,000-acre enclosure.

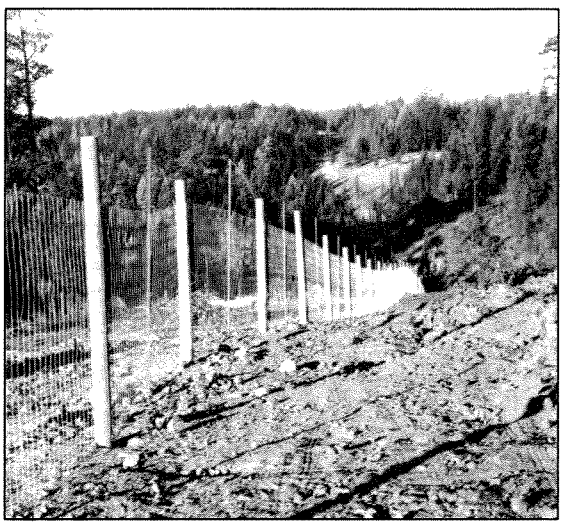


Figure 2—Over 27 miles of game-proof fence surround deer and elk summer range at the Starkey Experimental Forest and Range.

of 3,587 acres; and the winter feeding and handling area (hereafter referred to as the winter area) of 805 acres (fig. 1). Population densities of deer and elk in the enclosures were planned to resemble those outside the fence. Study plans call for spring (that is, preparturition) populations of 475 elk and 250 deer in the main study area, and 85 elk and 50 deer in the northeast study area.

Within these study areas, Rocky Mountain elk (*Cervus elaphus nelsoni* V. Bailey) and Rocky Mountain mule deer (*Odocoileus hemionus hemionus* Rafinesque) are the subjects of intensive research. One set of four studies—referred to as the Starkey Project—examines deer and elk response to cattle grazing, timber management, traffic on forest roads, and recreation (Johnson and others 1991).

The four studies of the Starkey Project take place on summer range within the main and northeast study areas, beginning each year in April and ending in mid-December. During winter, deer and elk are baited with alfalfa hay and pellets to the winter area (fig. 3). Animals not responding to bait trails are either live-trapped and hauled by truck to the winter area, fed in place within the main and northeast areas, or left to forage on natural vegetation from mid-December to April.

At the winter area, deer and elk are fed daily and handled periodically (fig. 4) from late December to April to meet objectives of the four summer range studies (Thomas 1989). Animals are fed high-quality alfalfa (fig. 5) at high rates to maintain healthy body condition and minimize the variable effects of winter weather on animal condition. Consequently, measures of herd productivity can be attributed to summer habitat conditions within the main and northeast study areas and are not confounded by effects of winter weather (Thomas 1989).

Additional studies already begun (appendix 1), or now planned,[2] rely on tame elk. These elk are raised at a calf-rearing facility within the winter area (figs. 3 and 4). There, elk calves are reared so that they can be handled and fed easily, efficiently, and with minimal stress. The tame calves are being used to evaluate relations of forest cover and energetics of elk, at a study site outside the Starkey fence on corporate timber lands. The decision to conduct the study elsewhere was made because unsuitable forest cover conditions are found within the Starkey fence (appendix 1). Additional tame elk are cared for year-round at the winter area and may be used for future research at Starkey (see footnote 2).

Regulations published by APHIS (USDA APHIS 1989) require documentation that research under jurisdiction of the Animal Welfare Act not be "needlessly duplicative" of past studies. Thomas (1989), Irwin and others,[3] Johnson and others (1991), and Wisdom (1992) thoroughly describe the unique nature of the Starkey research, and in doing so, document the need for it and the lack of duplication with previous studies.

---

[2] Vavra, M.; DelCurto, Tim; Bryant, L. 1992. Comparative dry matter intake, diet quality, digestion and digestion kinetics of elk, mule deer and cattle on a Blue Mountain summer range. 3 p. Draft research proposal. On file with: Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

[3] Irwin, L.L.; Thomas, J.W.; Cook, J.G. 1990. Influence of thermal cover on weight dynamics and condition of elk: plans for winter and summer studies. 43 p. Unpublished manuscript. On file with: Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

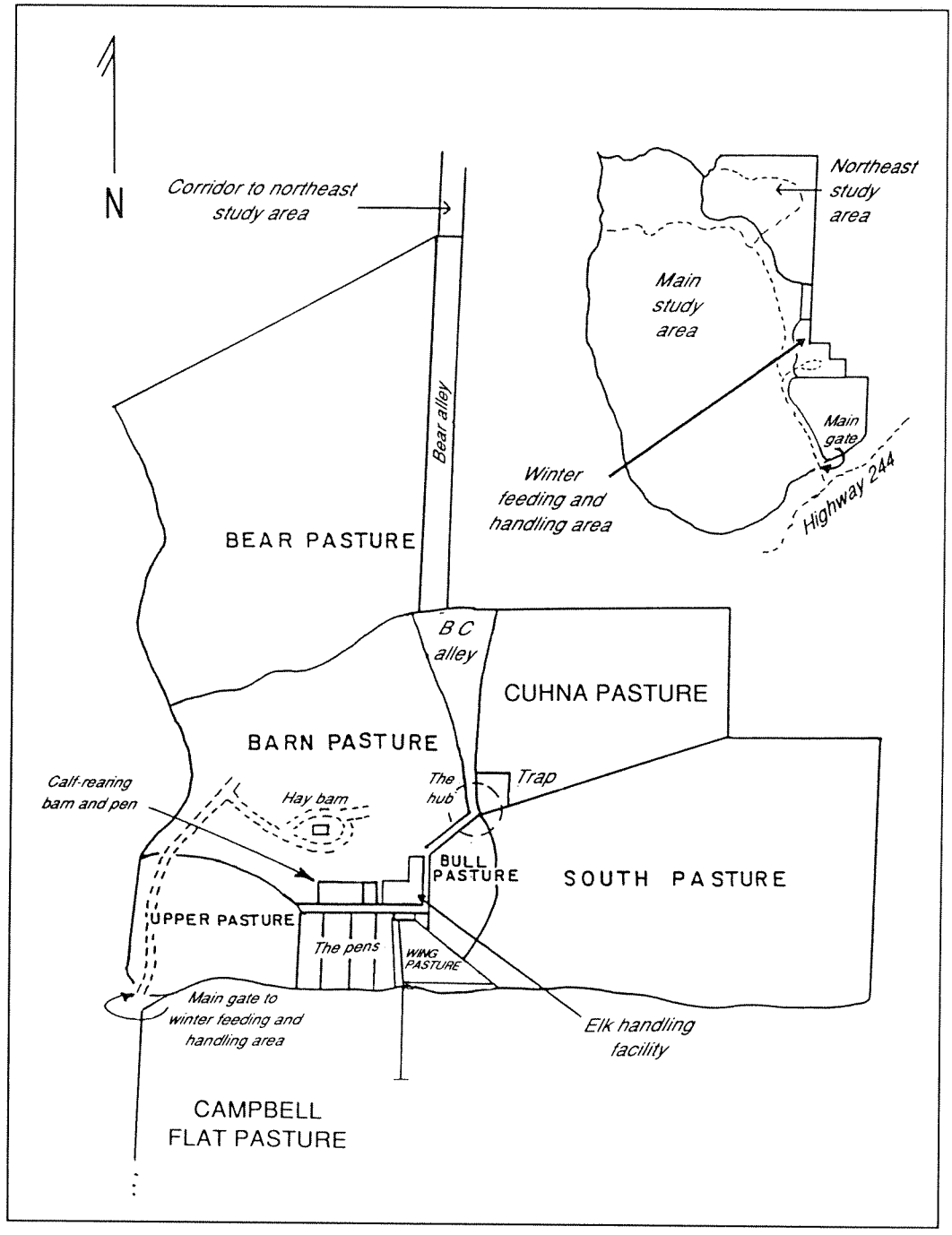


Figure 3—Pastures and facilities within the 805-acre winter feeding and handling area.

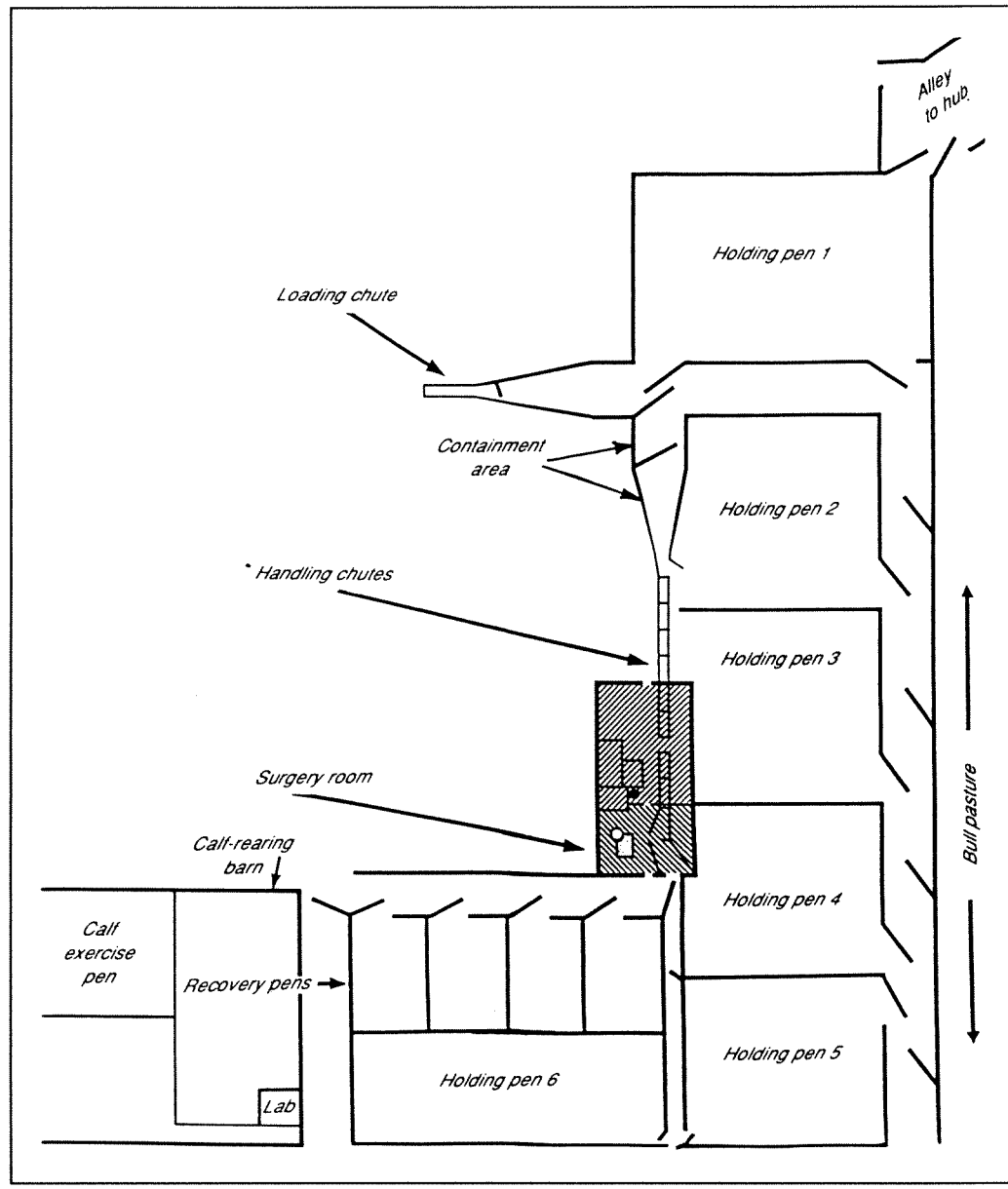


Figure 4—Holding pens and chutes at the elk handling facility allow efficient, safe movement and processing of animals.

Figure 5—Winter range is not present within the Starkey enclosure; elk and deer are fed each winter to maintain uniform health and provide experimental control over winter conditions.

Animal Welfare Act of 1985

Passed by Congress in 1966 and amended in 1970, 1976, and 1985, the Animal Welfare Act (U.S. Laws, Statutes, etc. 1985) protects the welfare of animals not reared for either food or fiber. The law specifically addresses the care and handling of mammals that are bought and sold, exhibited to the public, transported commercially, or used in research (USDA APHIS 1991). Invertebrates, cold-blooded vertebrates, birds, domestic rats and mice, and animals raised for food and fiber are exempt. Also exempt are animals used for breeding, management, production efficiency, or for improving animal nutrition (USDA APHIS 1991). Garbe and Wywialowski (1991) provide additional details about the Animal Welfare Act, its potential applications, and its administration by APHIS.

Research Activities Covered or Exempted by Law

The Animal Welfare Act and Code of Federal Regulations (CFR) standards for its implementation (USDA APHIS 1989; U.S. Laws, Statutes, etc. 1985) do not apply to normal management activities routinely performed by resource agencies (USDA APHIS 1989). Also exempt is any field study (research) "conducted on free-living wild animals in their natural habitat, which does not involve an invasive procedure, and which does not harm or materially alter the behavior of the animals under study" (USDA APHIS 1989).

Invasive procedures, however, have not been defined by APHIS, nor has APHIS defined what activities may harm or materially alter the behavior of regulated animals.[4] Dr. Malaby (see footnote 4) believes such definitions will be developed. Until then, he recommends that any contact with wild mammals, for research that involves live-trapping, physical handling, immobilization, containment, or transport, be considered as having potential to harm or materially alter animal behavior. For field studies of wildlife, this includes all research techniques listed in table 1.

[4] Personal communication. 1991. Homer Malaby, DVM, Animal Care Specialist, USDA Animal and Plant Health Inspection Service, Regulatory Enforcement and Animal Care, Western Sector-Animal Care, 9580 Micron Avenue, Suite J, Sacramento, CA 95827.

Table 1—Research techniques that are potentially invasive to wild mammals or that have potential to materially or harmfully alter the behavior of wild mammals under CFR standards of the Animal Welfare Act[a]

Techniques	
Transmitter, transponder implants for radio telemetry	Marrow-extracts
Catheterization	Laparotomy
Marking techniques:	Time-release drug implants
Toe clipping	Endoscopy
Branding	Rectal palpation
Ear-notching, tagging	Drug injections:
Biological markers	Sedatives
Fluorescent powders	Immobilizers
Use of dyes	Other drugs that alter awareness and reaction time of animals
Banding	Amputation of limbs
Organ biopsies	Toxicant testing:
Skin biopsies, scrapings	Predator/species control
Swabbing of body orifices	Chemical trials
Field sacrifice	Radiography
Ultrasound reading	Fluoroscopy
Externally applied telemetry harnesses	Routine capture and measurements:
Capture/recapture	Weights
Oral vaccination	Age/sex (hair clipping, tooth extraction)
Blood sampling	Size

[a] APHIS has not identified specific techniques that are invasive or that could alter animal behavior under their regulations. Consequently, this list is unofficial. Depending on the level of care and safeguards used, any of these common techniques have potential to materially or harmfully alter animal behavior, or to be invasive, or both.

Depending on the level of care and safeguards used, any of these common techniques have potential to materially or harmfully alter animal behavior, or to be invasive.

The U.S. Fish and Wildlife Service (USFWS) identified specific wildlife research techniques considered invasive or noninvasive under the Animal Welfare Act.[5] They also identified techniques that do or do not materially or harmfully alter animal behavior under CFR standards (see footnote 5). These guidelines were developed originally to help monitor compliance of Federally funded state research projects.[6] The guidelines, however, have been deferred from inclusion in USFWS policy and manuals for several reasons.

One reason is that the USFWS is not the regulatory agency for the Animal Welfare Act; therefore, it does not monitor compliance or ask for compliance reports from Federally aided research projects. Rather, it depends on APHIS for information on any Federally aided research that is not in compliance; Federal funds are withdrawn until the project complies.

Legal Requirements and Process

Any wildlife research subject to jurisdiction of the Animal Welfare Act (U.S. Laws, Statutes, etc. 1985) and CFR standards for its implementation (USDA APHIS 1989) must demonstrate compliance with legal requirements by adhering to the following process. This process, as described here, is quoted from a summary by USFWS (see footnote 5); it accurately describes major requirements for compliance that are outlined in 51 pages of standards issued by APHIS (USDA APHIS 1989) regarding research on wild mammals and other regulated animals.

Registration: Each non-Federal research facility or organization will register with APHIS, Regulatory Enforcement and Animal Care Sector supervisor in the State where research activities occur, by filing a form supplied by APHIS. This registration form will be signed by an individual with legal authority to bind the organization and will be updated every 3 years.

Review of Activities: The head of each registered and Federal facility will appoint a standing Institutional Animal Care and Use Committee (IACUC) charged with carrying out the intent of the rules for the institution. Each IACUC will consist of at least three individuals, one of whom is a Doctor of Veterinary Medicine; and one of whom is not affiliated in any way with the facility, but represents general community interest. IACUC is an agent of the facility and performs the following major functions:

1. Review the facilities and care given all captive species every 6 months. Report significant deficiencies (those which are a threat to health and safety of animals) along with a specific schedule and plan for correction to the head of the facility. Report uncorrected deficiencies to APHIS or the agency fifteen working days after the scheduled correction date.

---

[5] U.S. Department of the Interior, Fish and Wildlife Service. 1990. Draft requirements of wildlife research facilities and organizations, 1989 Animal Welfare Act rules. 5 p. On file with: U.S. Department of the Interior, Fish and Wildlife Service, Federal Aid Program, Arlington Square, 4401 N. Fairfax Drive, Arlington, VA 22203.

[6] Personal communication. 1991. Tom Williams, Federal Aid Coordinator, U.S. Department of the Interior, Fish and Wildlife Service, 911 N.E. 11th Avenue, Portland, OR 97232.

2. Review public or in-house concerns voiced about care and use of animals by facility personnel.
3. Make recommendations to the facility head regarding use of animals, facilities, and necessary staff training.
4. Review and approve, require modification to, or deny animal care and use procedures proposed in facility research projects. This approval must be gained before project initiation. IACUC may also review ongoing procedures and can suspend animal use activities if not in accordance with procedures submitted before project initiation. If an activity is suspended the head of the facility will review the reasons, take corrective actions, and report those actions to APHIS or the Federal agency involved.

IACUC review of research animal use will focus on the following areas: Alternatives to the use of animals, minimization of pain and distress in research animals, avoiding the use of animals in needlessly duplicative projects, use of appropriate pain-relieving drugs in consultation with a veterinarian, and appropriate methods of euthanasia. IACUC may use appropriate consultants in reviewing technical procedures, and may delegate review of animal use procedures to one of their members, if no member of the IACUC requests full IACUC review. Only IACUC members may vote on approval of any procedure. Researchers must be given opportunity to further explain and support procedures not initially approved by the IACUC. Procedures denied by IACUC cannot be approved by other facility officials.

Other institutional responsibilities include:

1. Provision of adequate training and credential review for personnel involved in animal use at the facility in order to ensure they can perform required tasks and know the proper procedures for questioning or reporting suspected care and use deficiencies.
2. Assurance that the veterinarian attending IACUC meetings provides an adequate veterinary care program for all species used at the facility.

Record-Keeping and Reporting: Each IACUC will maintain the following records for three years after the duration of activities and have them available to APHIS or agency personnel upon request:

1. Minutes of IACUC meetings.
2. Records of proposed animal use and IACUC's action on each.
3. Facility reviews with summary reports.

Annually, each registered institution or agency will report to APHIS:

1. Assurances that adequate veterinary care, consideration of alternatives to the use of animals in research, and adherence to the Animal Welfare Act rules are occurring at the facility.
2. The location of each site holding animals, and the species and number of animals used in research and teaching in the following categories:

(a) Involving no pain or distress;

(b) Involving pain or distress with alleviation;

(c) Involving pain or distress with no alleviation.

3. The number and species of animals being held or bred for future use in research, experimentation or teaching.

Each instance in 2(c) must be accompanied by an explanation justifying procedures Scientifically.

Registration of Research Facilities

Any institution using live mammals for research, testing, teaching, or experimentation must register with APHIS. Registration is mandatory if the institution includes regulated animals and any of the following activities, as stated by USDA APHIS (1991):

- Investigations on animal propagation and control, such as wildlife ecology.
- Laboratory tests, including pregnancy tests, allergy tests, and other diagnostic procedures.
- Quality control studies, such as studies on the safety, effectiveness, durability, or other quality tests of commercial products.
- College instruction, whether for research, education, or to improve medical treatment techniques and methods.
- Professional continuing education courses.

The APHIS guidelines further state the following:

> Registration is required to assure that laboratory animals are provided with care and comfort meeting USDA standards. The law and regulations require the use of appropriate pain-relieving drugs whenever possible. Registered research facilities and all agencies of the Federal government must submit an annual report stating how many regulated animals were used and if any painful experiments were conducted. The report must include a list of pain-relieving drugs or an explanation as to why it was necessary to omit pain relief.

Exempt Institutions

The U.S. Department of Agriculture, APHIS (1991), states that Federal institutions "are not required to register with USDA and are not inspected by APHIS, but each Federal agency is responsible for complying with all USDA standards of animal care and for submitting an annual report to USDA on the use of regulated laboratory animals." The Starkey studies are conducted through the Pacific Northwest Research Station of the Forest Service. As a Federal institution, this agency is exempt from registration and inspection.

Public institutions administered or funded by state or local government must register with APHIS as research facilities, however, as do private institutions (USDA APHIS 1991). If private institutions (privately owned facilities on privately owned land) receive partial funding from the Federal government or jointly conduct research with Federal researchers, private institutions must still register with APHIS (see footnote 4).

State wildlife agencies receiving research funds from Federal aid programs, such as Pittman-Robertson, also must register their research facilities with APHIS, unless such research is conducted jointly with Federal agencies at a Federal facility or unless such research involves only field studies (see footnote 4). If research is conducted at a Federal facility, state wildlife agencies must follow the same procedures that apply to Federal agencies. Annual reports and other compliance work are then done jointly with and through Federal research partners working with APHIS. The same applies to

private companies conducting research at Federal facilities in conjunction with Federal partners (see footnote 4). The ODFW research at Starkey is conducted jointly with the FS; therefore, it is exempt from registration and inspection by APHIS, but the State agency does submit joint annual reports with the FS.

Care and Handling of Animals

Specific protocols used for care and handling of deer and elk in research at the Starkey Experimental Forest and Range are described below. Additional protocols for care and handling of tame elk used in the forest cover-elk energetics study are described in appendix 1. A specific program of veterinary care for tame elk used in both studies has been established.[7]

The forest cover-elk energetics studies and those at Starkey will use the same IACUC because the studies are subject to the same requirements for compliance with APHIS regulations (USDA APHIS 1991), and they use similar methods of handling and care of tame elk. Both research sites will be inspected during courtesy visits by APHIS personnel, and Starkey Project and NCASI researchers will submit joint reports and other required information to APHIS.

Protocols described here will be updated whenever deemed appropriate by the IACUC and by researchers working in coordination with the committee. Proposed research will also be reviewed by the IACUC, and approved protocols will be appended to this document. As mentioned earlier, research for which protocols are described here is not duplicative of previous studies and therefore is allowed under APHIS regulations (USDA APHIS 1989).

Assumptions

1. Deer and elk residing in the northeast and main study areas of Starkey (fig. 1) during the research field season—April through December of each year—are considered wild, free-living animals in their natural habitat as defined under "Field Studies" of CFR standards of the Animal Welfare Act (USDA APHIS 1989).

The reasons are twofold: these study areas (fig. 1) encompass areas equal to or larger than the summer home ranges of most deer and elk living under free-ranging conditions in northeast Oregon (Leckenby 1984, Pedersen 1985), and habitat conditions within these areas are representative of those available to deer and elk residing on summer ranges throughout northeast Oregon (Leckenby 1984, Pedersen 1985). Accordingly, deer and elk have the same choices of space, food, water, and other habitat components in these areas as do other free-ranging herds in northeast Oregon. The only difference is the use of technologies to monitor animal selection of available habitats within the Starkey enclosure (Thomas 1989).

2. All human activities that occur as part of resource management of the northeast and main study areas from April through December each year are exempt from CFR standards of the Animal Welfare Act (USDA APHIS 1989). Such activities include, but are not limited to, cattle grazing and all range management practices; road and timber management activities; motorized vehicle use and its regulation; camping, mushroom-picking, hiking, hunting, photography, bike and horseback riding; and other recreational activities normally allowed on National Forests. Firewood gathering is not allowed on the Starkey Experimental Forest and Range.

---

[7] U.S. Department of Agriculture, Animal Care. 1991. Program of veterinary care for tame elk used for Starkey and forest cover-elk energetics studies. 6 p. Unpublished. On file with: Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

These activities are normal procedures used by state and Federal agencies to manage natural resources, including wildlife, on nearly all allocations of Federal land, including research areas. They are therefore exempt (USDA APHIS 1989, 1991) and also do not qualify as specific research techniques under the CFR definition of "Field Studies" (USDA APHIS 1989, appendix 2). Deer and elk in the northeast and main study areas have many habitats to select from in response to human disturbances, no different than the habitat choices available to other free-ranging herds in northeast Oregon (see assumption 1).

3. Human activities that occur directly as part of research of deer and elk in the northeast and main study areas from April through December are subject to jurisdiction of the Animal Welfare Act and its regulations if such research activities have the potential to be physically invasive to animals or to harm or materially alter the behavior of animals under study.

This assumption is in accordance with CFR standards that apply to "Field Studies" of wildlife in their natural habitat (USDA APHIS 1989). Included are all research techniques listed in table 1. Also included are physical handling, immobilization or containment in live traps or squeeze chutes, and transportation of animals. Appropriate, humane treatment of animals under these conditions is part of the protocols in this report.

4. Human activities that occur as part of either resource management or research—as identified under assumptions 3 and 4—in the northeast and main study areas from January through March are subject to jurisdiction of the Animal Welfare Act.

There are two reasons: (1) habitat in the northeast and main study areas, from January through March (outside the research field season), does not contain adequate space, habitat components, or sufficiently mild weather to constitute self-sustaining winter range for deer and elk like that found on typical winter ranges in northeast Oregon; and (2) animals cannot freely move to such winter ranges other than to an artificial one established at the winter area (fig. 3). Consequently, all aspects of care and handling of deer and elk residing in these study areas from January through March are detailed in our protocols. These include a list of safeguards that minimize human activities and disturbances to animals during that period.

5. All human activities, for either management or research, taking place year-round at the winter area are subject to the jurisdiction of the Animal Welfare Act and therefore are addressed in our protocols.

Deer and elk in the winter feeding and handling area are not living under free-ranging conditions that provide adequate food or space to maintain animal health without intensive, supplementary care by humans. Also, the winter area falls under the legal definition of "primary enclosure" as specified in CFR standards (appendix 2, USDA APHIS 1989). Any animals living in areas defined as primary enclosures are subject to CFR standards of the Animal Welfare Act.

6. Given these assumptions, all human activities subject to jurisdiction of the Animal Welfare Act and CFR standards are summarized for each area of the Starkey enclosure by season (table 2). This list includes all present or planned activities that may fall under jurisdiction of the Animal Welfare Act. Additional research techniques not listed but potentially subject to the jurisdiction of the law, like many of those in table 1, are not addressed because no plans exist for their use. Protocols will be developed for such techniques if and before they are used.

Table 2—Human activities at the Starkey Experimental Forest and Range that are potentially subject to CFR standards of the Animal Welfare Act

	Area and time activity is subject to CFR standards		
Activity	NE and main study areas, April-December	NE and main study areas, January-March	Winter feeding and handling area, year-round
Public and resource management activities[a]		x	x
Live-trapping deer with panel traps	x	x	x
Live-trapping deer with drive or drop nets	x	x	x
Live-trapping deer with net guns	x	x	x
Capturing deer with chemical anesthesia	x	x	x
Handling deer to collect data	x	x	x
Transporting deer	x	x	x
Live-trapping elk with portable traps	x	x	x
Capturing elk with chemical anesthesia	x	x	x
Collecting data from elk outside the handling facility	x	x	
Collecting data from elk at the handling facility			x
Transporting elk	x	x	x
Baiting animals to the winter area		x	
Live-trapping and transporting animals to winter area		x	
Winter feeding in study areas		x	
Pasture management, winter area			x
Releasing animals from winter area in spring		x	

Table 2—Human activities at the Starkey Experimental Forest and Range that are potentially subject to CFR standards of the Animal Welfare Act (continued)

	Area and time activity is subject to CFR standards		
Activity	NE and main study areas, April-December	NE and main study areas, January-March	Winter feeding and handling area, year-round
Care of animals living year-round at winter area			x
Operative procedures (surgery)	x	x	x
Care of tame elk	x	x	x
Pain alleviation and euthanasia	x	x	x
Monitoring the welfare of animals	x	x	x
Training personnel	x	x	x

[a] Includes, but is not limited to cattle grazing and all range management practices; road and timber management activities; motorized vehicle use and its regulation; camping, mushroom picking, hiking, hunting, photography, bike and horseback riding; and other recreational activities normally allowed on National Forests. Firewood cutting is prohibited.

Protocols for Public and Resource Management Activities

- Close the northeast and main study areas to all public access from about mid-December to mid-April each year to minimize human disturbance to those animals remaining in the study areas. Allow public entry into the study areas only with permission of or when accompanied by research personnel.
- Do not allow routine resource management activities within the northeast and main study areas from January through March to minimize human disturbance of wintering animals. Exceptions are care, feeding, and handling of animals, and maintaining facilities and equipment (for example, telemetry equipment and fences). Prohibited activities include all recreation and public access and all forms of timber and range management. Activities allowed are those for which protocols have been developed in this document.
- Each winter, attempt to lure, or live-trap and transport, as many deer and elk as possible to the winter area; this is where direct, active care of wintering animals is easiest. See protocols for baiting, live-trapping, and transport of animals.
- Prohibit public entry into the winter area year-round. Allow public entry only with permission from or when accompanied by research personnel.

- Limit resource management activities that occur in the winter area year-round. Exceptions are activities necessary to improve or maintain the humane, safe, and healthy care of animals. Included are timber and range practices to improve condition of pastures; all feeding and handling of animals and use of support machinery, technologies, and facilities; and repair and maintenance of facilities and technologies. Protocols for allowed activities are outlined in the sections that follow.

Protocols for Handling Deer

Live-trapping deer with panel traps—These traps are well suited to capture and hold single deer safely for extended periods (fig. 6). They are designed and function like a Clover[8] single-gate deer trap (Day and others 1980, p. 67-68), except that they are triangular instead of rectangular. The trap has two wooden sides, each 8 feet high and 5 feet wide, joined together at a 45° angle to form a "V" at the back. The entrance, or open end, is covered with mesh netting that completes the triangle. Deer are lured into the entrance with salt, alfalfa hay, pellets, or other baits located near a trip cord. Feeding on the bait triggers closure of the net gate. The triangular, narrow design of the trap restricts deer mobility sufficiently to prevent injury, yet is large enough for deer to stand or lie comfortably. The wooden sides are constructed of 1- by 6-inch slats, spaced 1 inch apart to maintain air flow through the trap.

Protocol—

- Do not set traps under severe weather or microclimatic conditions that could cause heat or cold stress to animals. In summer, refrain from setting traps when trapped animals may experience direct solar radiation combined with ambient air temperatures above 80 °F. During summer and winter, set traps away from openings and wind-swept ridges. Place traps instead in forest stands to modify extremes in microclimate around the traps and thus minimize any harmful effects of weather on trapped deer.
- Check traps at least daily. Make sure traps are not inadvertently set (that is, net gate open and trigger set) when traps are not being checked.
- Use two capable and trained personnel to remove deer from traps. Remove deer safely by pressing deer into the narrow portion of the trap. Grasp the front legs by reaching around the deer's body and then pull the front legs toward the rear of the animal. (Pulling the front legs this way restricts the deer from rising up on its front legs). At the same time, the second handler enters the trap and grasps the hindlegs. Once the legs are restrained, place the animal on its side within the trap. Then, keeping the back partially bowed to prevent spinal injury, gently slide the deer out of the trap. Blindfold the animal to keep it calm and begin collecting data (fig. 7). Have one person maintain gentle but firm pressure on top of the animal's midsection while data are collected. See "Handling deer to collect data" for the appropriate protocol for sampling and processing of captured animals.

---

[8] The use of trade or firm names in this publication is for reader information and does not imply endorsement by the U.S. Department of Agriculture of any product or service.

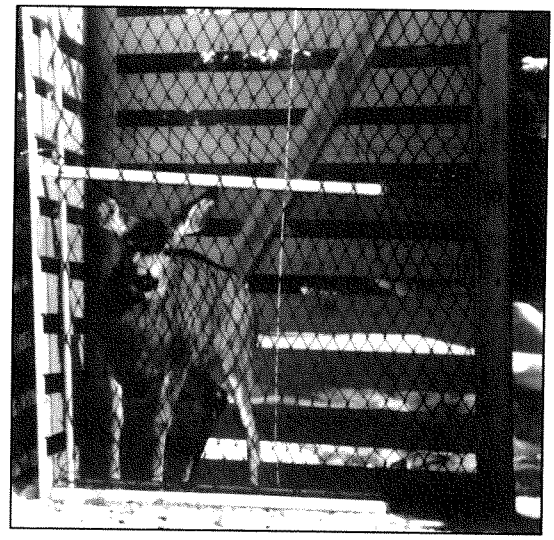


Figure 6—Panel trap used to catch mule deer.

Live-trapping deer with drive or drop nets—Deer are captured with drive nets by setting 50-foot sections of nylon rope netting, about 6 feet high and perpendicular to and touching the ground, across an area into which deer are driven. See Day and others (1980, p. 68-69) for details about setup and design. The same or similar nets can be dropped over deer by releasing nets suspended on poles or posts 6 feet or higher above the ground. Deer are attracted to the area with various baits and are captured when researchers electrically trigger the release of nets from the poles.

Protocol—

- Set up nets in an area free of potential hazards to deer. Make sure the area is free of large rocks and other protrusions that may injure entangled deer. For drive netting, select an area that requires herding deer a relatively short distance (usually less than 200 yards) and that affords deer few choices for escape (for example, avoid areas of dense cover and steep relief). These safeguards will minimize energy loss and potential stress to animals by minimizing the time that deer are driven toward nets and the time they are entangled.
- Use at least four research personnel for drive- or drop-netting; use only personnel who are capable and trained to drive and handle deer with maximum care and safety. For drive-netting, herd deer in the direction of nets either on foot, with all-terrain vehicles, or with a helicopter.
- Approach each entangled deer immediately; physically immobilize the animal by gently but firmly placing and holding the animal on the ground in a position that allows the animal to lie comfortably on its side. The head should be elevated to avoid aspiration of rumen contents. With at least one crew member keeping the animal immobilized, untangle the deer from the net as quickly as possible and place a blindfold over the animal's head. Use one person to keep the animal immobilized while other crew members begin sampling and processing techniques. See "Handling deer to collect data" for the appropriate protocol at this stage.

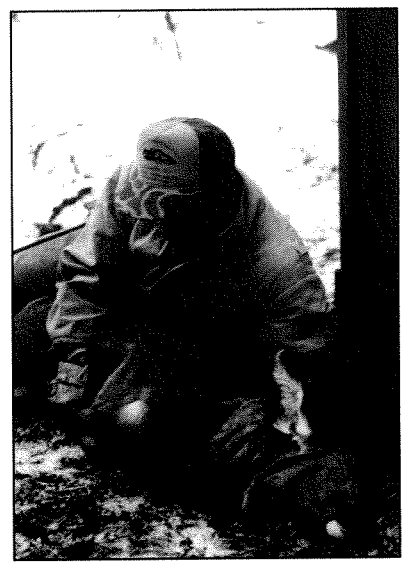


Figure 7—After a mule deer is captured, it is removed from the panel trap, blindfolded, and held securely and safely while being processed.

Live-trapping deer with net guns—This technique involves shooting a rifle-mounted net over the top of a deer within a target range of 50 feet (Day and others 1980, p. 69).

Protocol—Use at least two capable and trained research personnel to live-trap deer with a net gun. Have one person carry the equipment needed to sample and process the netted deer while the other person shoots.

To minimize chasing and harassment of animals, shoot the net only when the deer is within range (50 feet or less), in an area where the net will fully immobilize a deer quickly, and where a partially immobilized deer has little chance of escape (for example, running into a deep canyon or into dense cover).

Once the deer is netted, one person should keep the deer immobilized and lying on its side while the other person untangles the animal. Immediately place a blindfold on the animal's head to keep it calm. Begin sampling and processing techniques with one person making sure the animal remains immobilized and lying comfortably on its side. See "Handling deer to collect data" for protocols for sampling and processing of the captured animal.

Capturing deer by using chemical anesthesia—A syringe dart filled with anesthetic is shot from a rifle, blowgun, or pistol, or anesthetic is injected by hand with a syringe or jabstick (Day and others 1980, pp. 69-71; Fowler 1989, pp. 36-40). With successful penetration of the syringe in the animal's hide, the deer is fully anesthetized in less than 10 minutes. Because anesthetic is used, the animal is not aware of being handled during immobilization, thus reducing stress.

Protocol—

- Use analgesics or anesthetics only when such chemicals are judged to help reduce stress, prevent pain, and increase the safety of the animals being handled or of the project personnel. Use physical restraint (as described elsewhere) without analgesics, anesthetics, or tranquilizers when such chemicals are judged to increase the potential for stress or mortality or both. Often, the use of such chemicals on wild animals significantly increases the time of animal handling and confinement, and thus may increase physiological stress. Also, wild

animals that are highly excited react unpredictably to various anesthetics and dosages, thereby increasing the probability of physiological stress or a fatal reaction. The decision to use or not use anesthetics and other chemicals is the judgment of the researchers and their assessment of the particular conditions under which animals are being handled. The California Department of Fish and Game (1992:1-1) described the situation aptly:

> After developing chemical immobilization techniques for most of California's wildlife species, California Fish and Game (DFG) personnel realized that chemical immobilization was not a panacea for all wildlife capture and restraint. Many times, it was actually safer, efficient, and less stressful to the animal and the biologist to use some form of physical capture and restraint.

- Allow research personnel that have been trained and certified by ODFW or a qualified veterinarian to use chemical anesthesia. Have authorized ODFW personnel and the Starkey attending veterinarian (or other qualified veterinarian) train other research personnel in all aspects of chemical anesthesia. This includes training in rifle and gun ballistics (for example, trajectory and velocity) to ensure that darts hit the correct body location with the correct impact so that deer are immobilized quickly, humanely, and with little or no tissue damage. This also includes training in proper drug dosage, knowledge of the reaction of the animal, and proper first aid and care of the anesthetized animal.
- Use drugs that contain analgesics for pain relief, such as xylazine (for example, Rompun, Bay 1470) or combinations of xylazine and ketamine hydrochloride (for example, Vetalar, Ketalar, Ketaset). The preferred drug is Capture-All 5, a combination of Rompun and ketamine, used in the manner prescribed by Jessup and others (1983, 1985) and California Department of Fish and Game (1992). Use yohimbine hydrochloride to reverse the immobilizing effect of Capture-All 5 once the animal is ready to be released (Jessup and others 1983, 1985).
- Minimize use of immobilizing drugs not providing an anesthetic effect, such as succinylcholine chloride. Unlike anesthetics, such drugs allow the animal to be fully conscious during the handling process, with potentially stressful physiological effects (California Department of Fish and Game 1992). Follow dosage charts and methods prescribed by the California Department of Fish and Game (1992) for drug injections. See Fowler (1989, pp. 44-52) for detailed information about the physiological effects of chemical immobilizers commonly used to restrain animals.
- Follow procedures described by the California Department of Fish and Game (1992) to monitor the medical condition of anesthetized animals. This includes monitoring respiration, pulse, color of mucous membranes, response to auditory and visual stimuli, and body temperature. Make sure the animal is comfortable in body position, not subjected to extremes in ambient temperature, not under undue physical restraint, and that sensitive areas such as eyes are covered and protected while the animal is sedated. While handling, check the animal carefully for all signs of physical health (California Department of Fish and Game 1992), and administer appropriate antibiotics or other drugs to treat wounds, infections, or other unhealthy symptoms.
- See "Handling deer to collect data" for detailed protocols for sampling and processing a captured animal.

Handling deer to collect data—

Protocol—

- Once the deer is captured, immobilized, and lying on its side comfortably, blindfold the animal to keep it calm. If anesthetic has not been administered and only two people are processing the animal, use restraining boards to keep the animal safely immobilized. If anesthetic has been administered or three or more people are helping process the animal, have two people maintain gentle but firm pressure on top of the animal's mid-section. Use at least two trained personnel to collect data: one person to monitor the condition of the animal, as described by California Department of Fish and Game (1992), and a second person to sample and process the animal.
- Limit the time of restraint and handling to no more than 30 minutes. Minimize talking and keep voices quiet during handling to reduce the likelihood of a stressful reaction. If, at any time, the animal shows visible signs of stress (for example, hyperventilating or a rise in body temperature above 106 °F), either release the animal immediately or apply cool water over the animal's body, especially on the inside of the pelvis, around the neck, and under the stomach; administer vitamin B and selenium to reduce capture myopathy.
- Determine sex and age of the animal, and attach a numbered, aluminum ear tag to each ear with ear tag pliers. If the animal already has ear tags, record the number(s).
- Draw blood from the animal (if needed to meet research objectives) by inserting a 1-inch needle into a major artery or jugular vein in the animal's neck and withdrawing up to 20 milliliters of blood. Blood may be used to check for diseases and pregnancy and to monitor other parameters of physical health.
- If necessary for research, place a plastic identification or radio collar (Pedersen 1977) around the neck of the animal, ensuring a proper fit. Deer collars weigh no more than 3-1/2 pounds. This weight and the configuration of collars allow them to fit comfortably, safely, and humanely on deer without significantly altering their behavior. If the animal is already carrying a collar, check for signs of improper wear on the animal's neck. If found, remove the collar and administer topical antibiotics or other healing agents to bare skin or surface wounds.
- Check the animal for existing wounds or injuries, or those that could have resulted from handling. Administer topical antibiotics or other appropriate agents to surface wounds, and inject antibiotics if needed. If injuries are more serious, transport the animal to a veterinarian. See protocols for transporting deer. If the animal has injuries that appear fatal, euthanize the animal, following protocols outlined under "Pain alleviation and euthanasia."
- Weigh the animal by placing it in a wooden, enclosed box built specifically for holding and transporting deer. These boxes are about 42 inches tall, 22 inches wide, and 56 inches long, with ventilation holes on all sides and removable doors. Place the box on the weight scale, record the total weight, and subtract the weight of the box to obtain the animal's weight. Release the animal immediately in a safe location, or place the boxed animal on a truck if it is to be transported elsewhere (fig. 8).

Figure 8—Transporting deer in wooden boxes to the winter feeding and handling area. Note ventilation holes in sides of boxes.

Transporting deer—

Protocol—

- Use only wooden, enclosed deer boxes (described above) to transport deer. These boxes are large enough for deer to lie comfortably, but small enough to limit movement that could cause injury or stress. The wooden sides also provide darkness, in which deer remain calm. A series of 2-1/2-inch-diameter holes, 6 inches apart across the long sides of each box, provide sufficient airflow to prevent heat stress and provide adequate ventilation for animals in transit. The boxes meet all specifications for safe and humane transport of a wild ungulate, as described by Fowler (1989, p. 242).
- When lifting, moving, or transporting boxes, keep them level with the ground to prevent deer from being pushed to one end of the box and becoming excited and potentially stressed.
- When releasing a deer from the box, monitor its initial movements to confirm that it has not been injured in transit or during handling. If the animal limps, moves listlessly, or shows other signs of injury, try to recapture it for examination by a veterinarian.

Protocols for Handling Elk

Live-trapping elk with portable traps—A portable corral and a squeeze chute (Day and others 1980, pp. 66-67) may be used to capture and immobilize elk (fig. 9). The corral, made of steel bars or wood, encloses about 20 by 30 feet. A squeeze chute is connected to the corral. Animals are baited with alfalfa hay, pellets, or other attractants into the corral and trigger the closure of the entrance gate while feeding on the bait. Animals are moved singly into the squeeze chute to be handled and measured.

Protocol—

- Do not set traps under severe weather or microclimate conditions that could cause heat or cold stress to captured animals. Set traps away from openings (that is, in forest stands) to help modify extremes in microclimate around the traps, thus minimizing harmful effects of weather on trapped elk.
- Check traps at least daily. Make sure traps are not inadvertently set (that is, gate open and trigger set) when trapping is completed.

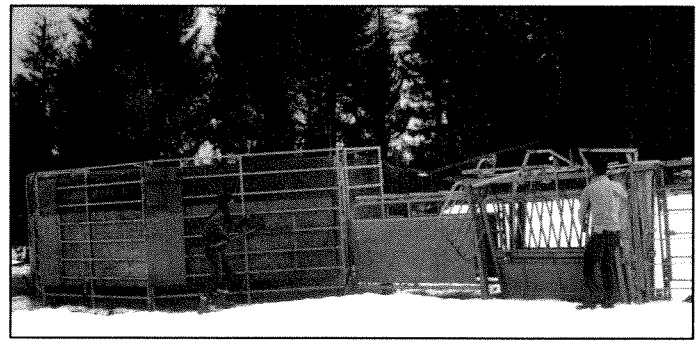


Figure 9—A portable corral trap, sometimes used to capture elk in the main and northeast study areas.

- During winter, de-antler branch-antlered bull elk before they are handled in the squeeze chute. This will lessen injuries to both animals and researchers and allow researchers to treat bulls as humanely and safely as possible during processing in the squeeze chute. During winter, antlers are nonliving material; they are shed annually in late winter or early spring. Consequently, removal of antlers during winter poses minimal risk to the health or safety of bull elk.
- To de-antler, drop a lasso over the animal's antlers while it is inside the corral or pen. Then quickly tie the free end of the lasso to a corral post so that the antlers are pulled snug against the fencing; the bull should not be able to move its antlers. Avoid cinching the antlers too snugly against the post; this could stress the bull and increase the likelihood of injury. Once the antlers are snug, quickly saw them off above the pedicles. If any jagged edges protrude above the pedicles after sawing, file the edges down so that the area above the pedicles is smooth; this is done later when the elk is in a chute in the handling facility. Use at least two trained and physically capable research personnel to lasso and secure the antlers for removal.

Capturing elk with chemical anesthesia—As with deer, this technique involves shooting a syringe dart filled with anesthetic from a rifle, blow-gun, or pistol, or injecting by hand with a syringe or jab-stick (Day and others 1980, pp. 69-71; Fowler 1989, pp. 36-40). With successful penetration, an elk is fully anesthetized in less than 10 minutes. Anesthetic and analgesic are used as part of the immobilizing agent. Consequently, the animal feels little if any pain and is not acutely aware of being handled while immobilized. Because new and safer drugs are continually being developed, researchers will use new drugs or new combinations of established drugs whenever such drugs are judged safer, more effective, or more humane than drugs previously available.

Protocol—

- Use analgesics or anesthetics only when such chemicals are judged to help reduce stress, prevent pain, and increase the safety of the animals being handled or of project personnel. See "Capturing deer by using chemical anesthesia" for further details.

- Allow research personnel that have been trained and certified by ODFW or a qualified veterinarian to use chemical darts to immobilize elk. Have authorized ODFW personnel and the Starkey attending veterinarian (or other qualified veterinarian) train other personnel in all aspects of chemical anesthesia. This includes training in gun and rifle ballistics (for example, trajectory and velocity); this will ensure that darts hit the correct body location with the correct impact so that animals are immobilized quickly, humanely, and with little tissue damage. This also includes training in proper drug dosage, knowledge of the reactions of the animal, and proper first aid and care of the anesthetized animal.
- Follow all rules and guidelines of the Federal Drug Administration (FDA) that govern the use and administration of drugs to capture and immobilize deer and elk. Make sure the rules and guidelines pertaining to each drug are included in employee training sessions.
- Use drugs that contain analgesics, such as xylazine (Rompun, Bay 1470) or combinations of xylazine and ketamine hydrochloride (Vetalar, Ketalar, Ketaset). Preferably, use a combination of Rompun and ketamine, such as Capture-All 5, as prescribed by Jessup and others (1983, 1985) and California Department of Fish and Game (1992). Use yohimbine hydrochloride to reverse the immobilizing effect when the animal has been processed and is ready for release.
- Consider using more efficient and effective drugs as they become available, such as combinations of medetomidine and ketamine.[9] Allow such drugs to be tested at Starkey with FDA permission and monitoring. Allow the use of carfentanil citrate when xylazine and medetomidine would not be effective, such as when darting a running or highly excited animal.
- Minimize use of immobilizing drugs not providing anesthetic effect, such as succinylcholine chloride. Unlike anesthetics, such drugs allow the animal to be fully conscious during the handling process, with potentially stressful physiological effects (California Department of Fish and Game 1992). Follow dosage charts and methods prescribed by California Department of Fish and Game (1992) for administering drugs. See Fowler (1989, pp. 44-52) for detailed information about the physiological effects of chemical immobilizers commonly used to restrain animals.
- Follow procedures of the California Department of Fish and Game (1992) to monitor the medical condition of anesthetized animals. This includes monitoring respiration, pulse, color of mucous membranes, response to auditory and visual stimuli, and body temperature. Make sure the animal is comfortable in body position, not subjected to extremes in ambient temperature, not under undue physical restraint, and that sensitive areas such as eyes are covered and protected. While handling, check the animal carefully for all signs of physical health (California Department of Fish and Game 1992), and administer appropriate antibiotics or other drugs to treat wounds, infections, or other unhealthy symptoms.
- See protocols for collecting data from elk for details about handling and processing a captured animal.

---

[9] Lance, William R. 1992. Letter dated April 7 to Jim Noyes. On file with: M. Wisdom, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

Collecting data from elk outside the handling facility—Most elk will be handled at the elk handling facility inside the winter area (figs. 3 and 4). At times, however, data will be collected from elk captured in portable corral traps set up throughout the study areas (figs. 1 and 9). These elk will be handled on-site by using a squeeze chute connected to the corral (Day and others 1980, pp. 66-67) and released, or they will be trucked to the handling facility. Elk captured in portable traps also may be anesthetized and data collected inside the corral, without using the squeeze chute. Sometimes, free-ranging animals (that is, not trapped) may be captured with chemical anesthetics. See "Live-trapping elk with portable traps" for detailed protocols about trapping.

Protocol—

- To begin collecting data, move elk singly from the corral into the squeeze chute by using trained personnel. Have one person work the squeeze chute, keeping it open while the animal moves into it in response to herding from other people.
- Once the elk is in the squeeze chute and immobilized, blindfold it to keep it calm (fig. 10). Minimize the time an elk is restrained and being handled. If at any time the animal shows visible signs of stress (for example, hyperventilating or a rise in body temperature), either release the animal or apply cool water over the animal's body, especially on the inside of the pelvis, around the neck, and under the stomach; administer vitamin B and selenium to mitigate capture myopathy.
- Verify or determine sex and age of the animal, and attach a numbered, aluminum ear tag to each ear with ear tag pliers. If the animal already has ear tags, record their numbers.
- Draw blood from the animal, if necessary, by inserting a needle into a major artery or jugular vein in the animal's neck and filling a 20-milliliter syringe. Blood samples are used to check for diseases, to monitor other parameters of physical health, and to determine pregnancy status of adult females.

Figure 10—Blindfolded elk in the squeeze chute of a portable corral trap.

- If needed, place a plastic identification or radio collar (Pedersen 1977) around the neck of the animal, making sure the collar fits properly. Elk collars weigh about 3.7 pounds. This relatively light weight and the design of the collar allow it to fit comfortably, safely, and humanely on elk without altering their behavior. If the animal is already carrying a collar, check for signs of improper wear on the animal's neck. If found, remove the collar, and administer topical antibiotics or other healing agents to bare skin or surface wounds.
- Check the animal for wounds or injuries that might have existed before handling, or that could have resulted from handling. Administer topical antibiotics or other appropriate agents to surface wounds, and inject the animal with antibiotics if needed. If injuries are more serious, transport the animal to a veterinarian. See protocols for transporting elk. If the animal has injuries that appear fatal, euthanize the animal by following protocols outlined under "Pain alleviation and euthanasia."

Collecting data from elk at the handling facility—The handling facility is in the winter area (fig. 3). Once elk are pastured there (see "Feeding and pasturing animals in the winter area"), they can be moved in small numbers to pens or containment areas adjacent to the handling facility (figs. 4 and 11). Animals can then be run through the facility and handled quickly, safely, and humanely for data collection.

Protocol—

- To lessen heat stress, handle elk only when temperatures are below 35 °F and preferably below 25 °F. Avoid handling in direct sunlight. The best times for handling elk are at dawn, dusk, or night.
- Select a pasture in the winter area from which animals will be handled. Open the gate that leads from the selected pasture to the "hub" (fig. 3). Over a period of 2 to 4 days, progressively move the feeding line from the pasture into the hub and down the alley leading to the pens adjacent to the handling facility.
- When animals are habituated to feeding in the alley and pens, and handling is scheduled to start, close the gate that connects the pasture to the hub, as well as the gate connecting the hub with the alley (fig. 3).
- Use personnel trained and experienced in moving elk. Herd elk in the alley toward the pens with four-wheelers, rather than with people on foot. Using these machines moves animals faster and more humanely in confined spaces, such as the alley. Once elk start moving down the alley, the four-wheelers prevent them from turning back. If elk are not caught in the pens after the fourth or fifth attempt, they are left alone until the following day. Try to contain animals in a small enough space so that they cannot pace back and forth, but without herding too large a group to minimize injuries (fig. 11). Once animals enter the pens or containment area adjacent to the handling facility, quickly close the gates that connect the pens or containment area with the alley. Check animals for signs of injury or heat stress. Release any animals from the pens or containment area that appear stressed; allow these animals to return to pastures. Move injured animals to a recovery pen for examination by a veterinarian; if injuries are minor, treat animals and monitor their condition for at least 24 hours before release.
- In the pens, de-antler any branch-antlered bull elk that will later be handled. Use the same protocol described earlier under "Trapping elk with portable traps."

Figure 11—Elk are herded into a containment area before being moved through the chutes inside the handling facility.

- Keep quiet while handling elk; conversations should be limited. Also, use only a few people to handle deer or elk, usually four. People who have worked together previously as a team will be more efficient.
- Begin handling immediately after de-antlering the bulls. From the pen or containment area, herd an animal into the outside chutes that lead to the entrance of the handling facility (fig. 4). Move the animal through the outside chutes by closing sliding doors behind the animal as it moves forward. Once the elk enters the building, move the animal into the desired chute (fig. 4) by closing and opening the appropriate sliding doors in front of and behind the animal. Handle elk in various chutes, listed below, to collect data as elk walk through in order:

1. V-squeeze chute—After the animal enters the chute, close the sliding doors ahead of and behind the animal. Then push the free end of the side panel inward toward the animal, which will narrow the area around the animal to a small triangle. This "V-squeeze" restricts the mobility of the animal, yet is safe and humane. Attach or replace radio or identification collars and ear tags.
2. Rectal palpation chute—When a cow elk is in the chute, the opening in the rear sliding door can be used to check for pregnancy by rectally palpating the uterus. This technique (Follis and Spillett 1974) should be performed only by a qualified veterinarian or by research personnel trained by the attending veterinarian of the IACUC.

3. Weight chute—At entry, each animal steps onto a padded scale on the floor (fig. 12). After closing the sliding doors behind the animal, record the animal's weight from the digital read-out modem. Once recorded, open the front door and allow the animal to move forward.
4. Surgery preparatory chute—This chute, adjacent to the surgery room (fig. 13), is used when injecting elk with anesthetic before surgery. The chute resembles the V-squeeze chute. Use the same protocol outlined for chute 1 to temporarily immobilize the animal in a "V-squeeze." Once immobilized, inject the elk with anesthetic by using the protocols outlined under "Capturing elk with chemical anesthesia." When the anesthetic takes effect, open the side gate facing the surgery room and slide the animal onto the lowered operating table (fig. 13). After the table is raised (by hydraulic power), the animal is ready for surgery. See "Operative procedures (surgery)" for protocols. We anticipate very few surgeries each year.
5. Powder River squeeze chute—Usually all elk are moved quickly through chute 4, and into the Powder River squeeze chute (fig. 14). This chute, originally designed for cattle (Fowler 1989, p. 129), has side panels of metal bars designed to gently "collapse" against both sides of an animal to provide restraint without injury. Pressure from the bars against the animal's body is controlled with metal levers (fig. 14). This squeeze chute provides more control and safety for the researcher during handling than does the V-squeeze chute. The Powder River squeeze chute is preferred for attaching or changing ear tags and collars, drawing blood, and taking other samples or measurements.

Before closing the squeeze chute, make sure the animal is fully inside, with its head facing forward and beyond the chute. Close the chute slowly, making sure the bars collapse around the animal's body while its head remains in front of the chute. Maintain adequate but not overly restrictive pressure on the bars against the animal's body; do not restrict its breathing.

Once the animal is immobilized, record its ear tag numbers, or attach ear tags. If necessary, attach or replace a radio or identification collar. Draw blood samples by using protocols identified under "Collecting data from elk outside the handling facility."

- Check for wounds or injuries that might have existed before handling or that could have resulted from handling. Administer topical antibiotics to surface wounds, and inject the animal with antibiotics if needed. If injuries are more serious, transport the elk to a veterinarian or have a veterinarian examine the animal on site. If the animal has suffered injuries that appear fatal, euthanize it by following the protocol outlined under "Pain alleviation and euthanasia."
- Monitor elk for obvious signs of stress (California Department of Fish and Game 1992) during the entire handling period. If at anytime an animal appears heat-stressed (for example, hyperventilating and body temperature rising), immediately move it to a recovery pen outside the handling facility. Once the animal has recovered, release it to the pasture from which it came. If recovery does not occur within a few hours, have a veterinarian examine the animal. Euthanize the animal if injury or stress appear fatal, as outlined under "Pain alleviation and euthanasia."

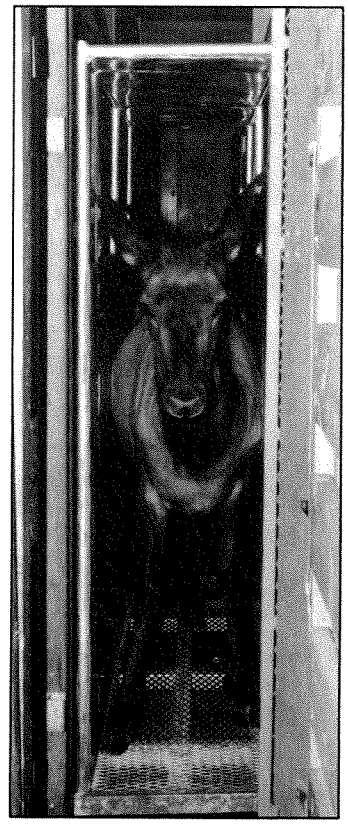


Figure 12—A cow elk in the weighing chute; weights are displayed on a digital modem outside the chute.

Figure 13—Surgery room within the elk handling facility.

Figure 14—The Powder River squeeze chute, used to hold elk while collecting data and attaching radio collars and ear tags.

Transporting elk—Elk can be transported efficiently and safely with a cattle truck. A 2-ton cattle truck, like that used at Starkey, is typically designed with panels of metal bars inserted vertically into the sides of a flatbed. The panels provide a visual barrier between the animals and the outside environment, and are spaced to allow adequate airflow around the animals during transport. The floor of the flatbed typically has a nonslip surface of foam or rubber to provide safe, sure footing for animals.

Protocol—

- Load elk directly from the squeeze chute of the portable traps or from the loading ramp at the elk handling facility (fig. 4). If a 2-ton flatbed is used, transport no more than 20 cow, calf, or de-antlered bull elk. If more than one bull is transported, or when transporting bulls with cows and calves, de-antler the bulls before loading. If elk appear stressed, do not load them, but hold temporarily in pens for observation.
- Minimize the time elk are in stock trucks, such as a 2-ton pick-up. Try to load and transport elk during cooler periods (for example, at night, dawn or dusk, or during days with cloud cover and temperatures below 40 °F).
- Upon release, monitor animal behavior for signs of stress or injury (as described in previous sections on handling elk). Release animals in a secure area, and return within a few hours to check the status of animals that appeared stressed. If stress or injuries appear fatal, reduce suffering by euthanizing.

Baiting animals to the winter area—

Protocol—Place bait trails of alfalfa hay and pellets throughout the study areas each December when the areas have been closed to the public. Progressively move the bait trails toward the winter area, as quickly as animals will follow them. Attempt to attract many deer and elk near the entrances to the winter area by feeding heavily in these locations. Once animals are concentrated near entrances, move bait lines inside pastures of the winter area until deer and elk follow in large numbers.

Winter feeding in the study areas—

Protocol—

- Feed animals in the study areas (that is, those animals that do not follow bait trails to the winter area) *ad libitum* throughout the winter. In general, feed elk at a maintenance rate of 8 to 12 pounds of hay/(day•elk), or at a higher rate if weather and snow dictate. Feed deer at a maintenance rate of 4 to 6 pounds/(day•deer), or at a higher rate under severe weather and deep snow. Keep main roads open throughout the study areas to expedite regular feeding of animals.
- Fly over the study areas periodically to monitor distribution of animals in relation to the bait trails and feeding lines. Adjust locations of the feeding lines so that most animals have adequate hay and pellets available to them throughout the study areas. Use snowmobiles to carry feed in areas inaccessible by truck.

Live-trapping and transporting animals to the winter area—In winter, try to live-trap deer and elk that do not follow the bait trails to the winter area. Transport the animals by truck to the winter area, or release them on site, as appropriate. See protocols for live-trapping and transporting deer and elk.

Feeding and pasturing animals in winter area—Pastures in the winter area (fig. 3) contain sufficient cover and water for thermoregulation by deer and elk during most winter conditions. Because the area receives heavy snowfall and is not historical winter range, forage is limited; consequently, deer and elk here are fed daily to maintain healthy physical condition and meet research objectives (fig. 5).

Protocol—

- Once deer and elk are transported to or arrive at the winter area, separate the two species by baiting deer into one pasture by themselves. This will reduce the likelihood of injuries from fighting between the species.
- Feed animals daily, throughout the winter, at rates as high as animals desire. In general, feed elk at an equivalent rate of 8 to 12 pounds of hay/(day•elk), or at a higher rate if weather and snow dictate. Feed deer at an equivalent rate of 4 to 6 pounds/(day•deer), or at a higher rate under severe weather and deep snow. Use metal feeders to supply pellets, unless conditions are cold and dry, in which case they can be spread thinly on the ground. If pellets absorb moisture, they deteriorate quickly. To reduce fighting between animals and possible injuries, distribute food in long, narrow lines throughout each pasture. Vary location of the feeding lines so that newer hay and pellets are not mixed with older, poor quality hay left over from past feeding lines. Animals are also provided sulphur blocks, iodized salt, and a multimineral supplement in granular form.

- Monitor water in the pastures throughout winter to ensure either snow or free-standing water is available. This is especially important during late winter and early spring when snow has melted but animals have not yet left the winter area. When needed, place and fill water troughs in pastures. Check troughs daily and re-fill as necessary.
- Reduce human disturbance to animals by allowing people in the pastures only to feed animals or provide veterinary care. Minimize public entry in the pastures and allow only with permission of, or when accompanied by, research personnel.
- Control coyote (*Canis latrans* Say), wild dog, "coydog," and other predator harassment and predation of deer and elk in the pastures when necessary. Due to the small pasture size and relatively long fence perimeter, the winter area is an unnatural setting that facilitates predation by canines and felids. Some predator control, either by shooting or trapping, will likely be necessary each year. Authorize only personnel trained and certified in using control methods to remove predators, and only after a problem has been documented.

Releasing animals from winter area in spring—Release deer and elk from the winter area when there is sufficient forage to support them in the main and northeast study areas, usually in late March and early April.

Care of animals living year-round at winter area—Less than 100 wild deer and elk live within pastures of the winter area on a year-round basis. These deer and elk function as a reserve that can supplement specific age and sex classes of the study herds when needed to meet research objectives. These herds are also a source of calves and fawns for research with tame animals. See "Raising and care of tame elk" for details about such research.

Protocol—

- During spring, summer, and fall, allow resident deer and elk the use of all pastures of the winter area. Provide supplemental alfalfa hay and pellets during periods of drought when high-quality forage may be lacking. Provide free-standing water in troughs during periods of drought, refilling troughs when necessary.
- To minimize human disturbance and harassment of animals, limit public entry into the winter area during spring, summer, and fall; allow entry only with permission of, or when accompanied by, research personnel. Do not allow any management or human activities within the winter area except those actions providing direct benefits to deer and elk (table 2).

Operative procedures (surgery)—

Protocol—

- Have a licensed veterinarian conduct, supervise, or approve all surgery in as clean and sterile an environment as possible. Conduct nonemergency procedures in the surgery room at the winter area, in the elk handling facility (figs. 4 and 13). Have the attending veterinarian for the IACUC or another qualified veterinarian conduct, supervise, or approve all surgical procedures. If the attending veterinarian does not conduct the surgery, he or she should monitor the results for compliance with the program of veterinary care (see footnote 7).

Raising and care of tame elk—Many papers outline procedures for raising and training juvenile ruminants (Deming 1954, Hobbs and Baker 1979, Krzywinski and others 1980, Neil and others 1979, Parker and Wong 1987, Pekins and Mautz 1985, Reichert 1972, Robbins and others 1987, Schwartz and others 1976, Wood and others 1961, Youngson 1970). The procedures outlined below are an amalgamation derived from the written record, personal communication with researchers who have much experience with neonatal ruminants, and 2 years of experience raising nearly 50 elk calves. Many of the documented attempts to handraise ruminants were plagued with variable success, often without specific identification of causes of illness and mortality. Thus, the following procedures are intended to be general; animal handlers must be flexible and creative to deal with the many diseases and animal personalities encountered in raising neonatal ruminants.

Justification—Tractable, hand-reared animals that tolerate nearby observers are desirable or mandatory for many research purposes. Examples include studies of energetics, disease, physiology, nutrition, reproduction, and food habits. Stress and mortality associated with close constraint and confinement of wild-caught ungulates often can be avoided with tamed, bottle-raised animals (Parker and Wong 1987).

Facilities—A barn has been built at the Starkey Experimental Forest and Range specifically for raising juvenile deer and elk (fig. 15). This barn, measuring 30 by 60 feet, is built on sealed concrete and is completely enclosed with insulated walls. It contains two large windows and five doors for ventilation, a small lab with running water, a feed storage area, and 32 stalls, each 4 by 4 feet, for housing juveniles separately. The roof contains translucent sheeting to provide natural light, and the barn is equipped inside and out with electrical lights. Because the barn is intended primarily for summer use and provides ample protection from wind and rain, heaters are unnecessary. When young calves are maintained in the rearing barn, excrement is removed from stalls daily, and bedding is replaced every other day. Stalls and walkways are scrubbed once per week with disinfectant, and the entire barn is washed and scrubbed with disinfectant twice per month. Nursing bottles are individually labeled to prevent sharing among calves, and washed with soap and hot water after every feeding. Watering buckets in stalls are emptied and cleaned daily, and feed buckets are cleaned as necessary.

A 1-acre pen, immediately adjacent to the barn, provides an area for juveniles to exercise (fig. 4). Four enclosures, each about 2 acres in size, also provide areas to keep elk and segregate them as desired by sex, age, or experimental group (see "the pens," fig. 3). There is also a 5-acre enclosure at the facility, permitting access to natural forage (fig. 3, "bull pasture").

Capture of juveniles—There are many techniques for capturing neonatal ruminants; procedures using helicopters are probably the most efficient for capturing neonatal elk from free-ranging herds. A helicopter crew, working in tandem with a ground crew, searches suitable parturition habitat. Noise from the helicopter generally evokes a hiding response from neonatal calves, facilitating their capture by a ground crew. Bruce Smith, National Elk Refuge, and Tom Hobbs and George Bear, Colorado Division of Wildlife, have extensive experience capturing calves by using helicopters.

Ground searches are used to capture neonatal calves at the Starkey Experimental Forest and Range. Wild adult cow elk are held on the winter area through parturition in fenced pastures of about 200 acres (natural diets are supplemented with alfalfa pellets and alfalfa hay). Searches are conducted every 2 or 3 days by six to eight people through these pastures during late May and June to locate newborn elk.

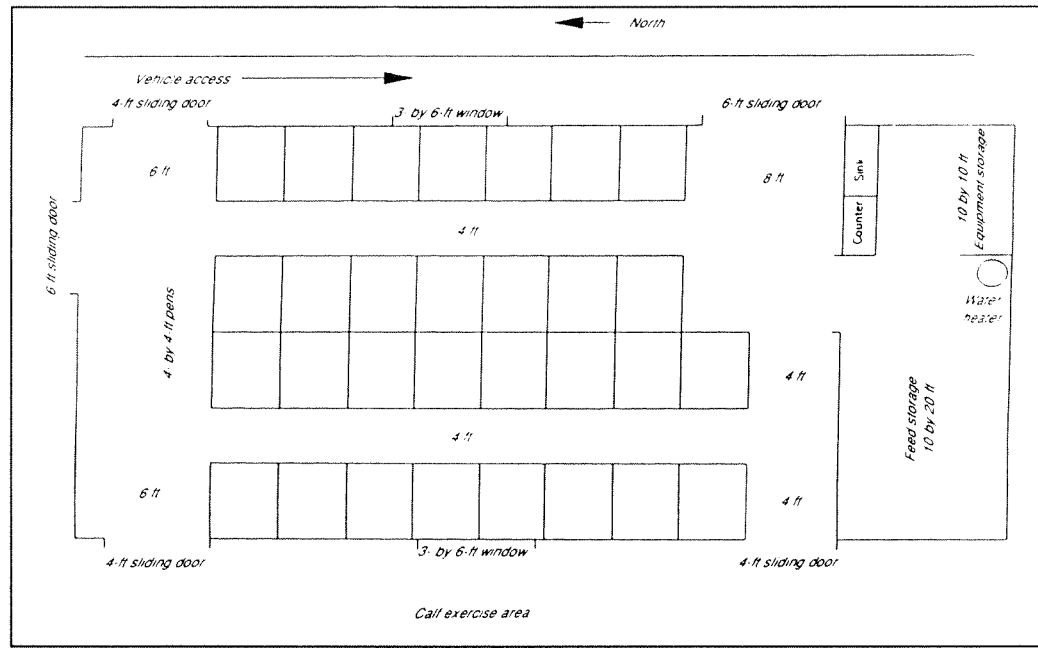


Figure 15—Floor plan of the calf-rearing barn within the winter feeding and handling area.

Neonates are captured by surrounding the animal with 4 to 5 people and restraining by hand or net. This generally requires little effort; their hiding instinct precludes attempts to escape. They are quickly blindfolded to reduce stress, and age is estimated. If less than 24 hours old, they are immediately released. Calves or fawns that have not had the opportunity to nurse will likely die if taken from their dam and will be susceptible to disease if they have received inadequate colostrum. Acceptable minimum age for separation from the mother is 24 hours, because neonates can absorb intact immunological components from colostrum only during this time (Robbins and others 1987). Johnson (1951) provides detailed aging criteria for elk calves, and frequent searches (for example, alternate-day) increase the likelihood that calf age is accurately identified.

Elk calves left with the mother for 3 to 4 days are likely more able to withstand the stress of handling and disease after separation from the mother. And they seem to be as tractable as calves taken when 1 to 2 days old. Reichert (1972) notes, however, that leaving deer fawns with their mothers more than 2 days decreases tractability.

Neonates are blindfolded when transported to the barn. They are given injections of vitamin ADE and antibiotics (Naxcell and penicillin), and their navels are swabbed with iodine. They are left alone in their stalls for about 12 hours to permit habituation to their new surroundings.

Handling and feeding—Nearly constant attention 12 to 16 hours a day during the first week or so after capture is required to facilitate nursing from a bottle and maximize tractability later in life. Elk calves in particular are reluctant to nurse from a bottle, and require many creative techniques to get them to nurse.[10] Sheets hanging

[10] Personal communication. 1991. Charles T. Robbins, professor, Department of Zoology, Science Hall, Washington State University, Pullman, WA 99163.

across their stall and lab coats worn by technicians are helpful. Also aprons and vests made of brown fake fur and fake fur covers for milk bottles are often useful. Animals that have not nursed within 3 days after capture are force-fed with a syringe equipped with soft plastic tubing (Hobbs and Baker 1979).

Many studies have presented milk formulas for hand-raising captive cervids. Few have closely simulated the fat, protein, and dry matter content of wild cervid milk, and most have been associated with enteritis (Parker and Wong 1987). Charles Robbins (see footnote 10) and Parker and Wong (1987) have had good success with whole cow or goat milk combined with a lamb milk replacer, with the milk replacer added at 10 percent of the milk weight. Our success with this formula also has been good.

The frequency of feeding ranges from five to six times per day at 3- to 4-hour intervals during the first 2 to 3 weeks postpartum. Frequency is reduced gradually to once or twice per day in August, and juveniles are weaned between late August and November (Hobbs and Baker 1979, Parker and Wong 1987, Reichert 1972).

Feeding volume requires special attention because growth and development are highly dependent on intake. We use data from Robbins and others (1981, fig. 3) to guide feeding levels for elk calves. Neil and others (1979), Parker and Wong (1987), Pekins and Mautz (1985), Reichert (1972), and Sadlier (1980) provide data on feeding levels for deer fawns. Solid food is offered within a few weeks after capture, although we have found that calves remain uninterested until 3 to 6 weeks old. Solid food includes dairy-quality alfalfa hay, alfalfa pellets, calf manna, and grain rations consisting mostly of rolled oats and corn. Calf manna and grain rations contain vitamin and mineral additives. Milk and solid food consumption are recorded each day.

Juveniles require extensive handling to tame and train during the first 3 months after capture. Many hours are spent sitting with neonates and providing tender love and care (Parker and Wong 1987). Several authors have presented specific techniques for training cervids for various purposes (Hobbs and Baker 1979, Reichert 1972, Schwartz and others 1976). Some general procedures are important regardless of the purpose of training: training should begin early in life, negative reinforcement should always be avoided, positive rewards must be incorporated into training protocols, and training procedures must be repeated frequently.

Care during hunting seasons—Tame elk will be kept in areas closed to hunting and in areas protected from the view of hunters during hunting seasons. Tame elk at the elk-handling facility will be moved to pastures furthest from hunters or kept in pens adjacent to the elk-handling facility to ensure their protection and minimize the likelihood of their being shot accidentally during the hunting seasons.

Treatment of disease—Disease and occasional mortality typically plague efforts to raise neonates in captivity (Hobbs and Baker 1979, Parker and Wong 1987). Unsuitable milk formula accounts for many difficulties identified in earlier literature, and recent improvements apparently can reduce disease and mortality substantially (Parker and Wong 1987). Wildlife disease research, however, is documenting an increasing number of livestock pathogens present in free-ranging ungulate populations, and these pathogens can cause losses in captive cervids (Smits 1991).

We work extensively with Dr. T. McCoy of La Grande, Oregon, and Drs. S. Parish and G. Barrington at Washington State University (WSU) to diagnose and treat illness in sick calves. Postmortem examination is conducted on all carcasses, and sick calves are transported to WSU for treatment when necessary. Facilities exist to house sick animals separately from healthy animals. Treatments are recorded for all sick animals.

Other Protocols

Pain alleviation and euthanasia—The prevention of pain during all aspects of care and handling has been central to the protocols outlined in this paper. All research at Starkey involving care and handling of animals (tables 1 and 2) uses the most humane and safe methods available. The protocols in this report prescribe methods that prevent rather than alleviate pain, and the protocols include quick responses to alleviate pain or to treat injuries. This is particularly true of protocols that deal with capture myopathy. See "Handling deer to collect data" and "Handling elk outside the elk handling facility" for examples.

Regardless of how well animals are handled and cared for, some mortality will occur in association with research activities. With the level of care outlined here, and the experience of the Starkey Project to date, we anticipate that annual mortality will amount to no more than a few animals. Additional mortality may occur each year, however, that is not associated with research operations; this includes mortality from disease, predation, hunting, and weather. Regardless of the cause of mortality, some animals will require euthanasia as the most humane form of death.

Protocol—

- Have the Starkey attending veterinarian train research personnel to recognize fatal conditions or injuries that may impose great suffering and that present little chance of recovery for the affected animal(s). Authorize personnel to euthanize all deer and elk that exhibit such fatal symptoms whenever these animals are found, preferably with high-powered rifles. Euthanize wild animals with a single shot fired into the animal's brain at close range. This is humane and maximizes the personal safety of researchers. Personnel authorized to euthanize animals should have training in rifle and gun ballistics and knowledge of the proper body locations for placing shots for a quick and humane kill. Euthanize tame animals by injecting them with sodium pentobarbital, with or without phenytoin sodium solution (see footnote 7).
- Allow researchers to euthanize a small number of deer and elk each year for research purposes. Use the methods of euthanasia described above. It is estimated that fewer than 10 animals will be sacrificed for research each year, although the actual number will vary with annual data requirements. Animals will be sacrificed only if associated data are deemed essential for meeting research objectives and only after all other means of data collection have been considered and subsequently deemed less humane, less safe, or less plausible.
- Remove euthanized animals from the winter area or other areas where animal numbers are high. Bury, burn, or place carcasses in a lime pit if the animals were diseased or were euthanized with sodium pentobarbital. This will prevent secondary transmission to wildlife scavengers such as bald eagles. Thoroughly sterilize any indoor facilities where animals have died. Maintain records of all animals that are euthanized, and list the reasons for euthanasia. Collect and maintain this information as outlined in "Monitoring the welfare of animals."

- Donate to charitable organizations those carcasses that were euthanized by rifle shot. Carcasses of animals euthanized with drugs cannot be donated for this purpose.

Monitoring the welfare of animals—

Protocol—

- Each time an animal is handled or cared for, record the results or effects of treatment on the behavior and physical health of the affected animal(s). Specifically record the following information: type and number of animals handled, painless handling or care procedures used, painful handling or care procedures used, pain alleviation used, and posttreatment status of each animal, including any mortalities. Record the date and location of treatment and the people involved. Record information on data sheets that can be entered directly into a computer database for convenient storage and retrieval.
- Summarize these results in an annual report to the Starkey IACUC. Include the following information: number of deer and elk that were handled and the types of handling or care administered, number of deer and elk mortalities that resulted from research activities compared to the total number of deer and elk handled, and the number of deer and elk requiring euthanasia in association with research activities. Also summarize the number of animals euthanized not in association with research activities; this includes animals found injured from hunting or predators, or those found suffering from a fatal disease or physical condition.
- Summarize results for the same reporting period required by APHIS, which is the Federal fiscal year that begins October 1 and ends September 30. Transfer data required by APHIS to their annual report form,[11] which must be submitted to APHIS no later than December 1 each year.
- Before submitting data to APHIS (see footnote 11), meet with the IACUC to review results of the annual report. Interpret results of the report for the IACUC and recommend improvements in animal care and handling. Incorporate changes approved by the committee in updates of the written protocols.

Training personnel—

Protocol—

- Issue new employees a copy of the written protocols for animal care and handling. Offer training for new employees to implement these protocols when deemed necessary. Provide additional training for all personnel when any new handling techniques, particularly those involving anesthetics, are tested or adopted for use. Conduct additional training sessions whenever deemed appropriate by the IACUC.

---

[11] U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1991. APHIS form 7023, and directions for completing the annual report. 4 p. On file with: Pacific Northwest Research Station, Forestry and Range Sciences Laboratory, 1401 Gekeler Lane, La Grande, OR 97850.

Functions of the Institutional Animal Care and Use Committee

Membership

The Institutional Animal Care and Use Committee (IACUC) for the Starkey and forest cover-elk energetics studies consists of three members: an attending veterinarian for these studies; a local citizen, representing community interests; and an authorized representative of institutional officials of the Starkey Project. For information about other key officials, contact the authors.

Responsibilities and Legal Authority

The IACUC is legally charged with carrying out the intent of the Animal Welfare Act, its regulations, and the protocols developed by the research institution. The IACUC is specifically charged to do the following:

1. Review the facilities and care given all captive species every 6 months. Report significant deficiencies (those that are a threat to health and safety of animals) along with a specific schedule and plan for correction to the head of the facility. Report uncorrected deficiencies to APHIS 15 working days after scheduled correction date.
2. Review public or in-house concerns voiced about care and use of animals by facility personnel.
3. Make recommendations to the facility head regarding use of animals, facilities, and necessary staff training.
4. Review and approve, require modification to, or deny animal care and use procedures proposed in facility research projects. The IACUC must review ongoing procedures; the committee can suspend all animal-use activities not in accordance with protocols developed and approved for the research. If any activity is suspended, the head of the facility will review the reasons, take corrective actions, and report those actions to APHIS.

Biannual Meeting and Inspection

- Conduct two meetings each year—one in January or February and another in October or November—of the IACUC and representatives of the research institution. Inspect the Starkey and NCASI research facilities and the care given all captive animals. During the winter meeting, include a field inspection of care and handling of both tame and wild elk residing at the Starkey winter area, as well as a field inspection of the NCASI site. During the fall meeting, visit one or both sites, depending on where tame animals are being cared for at the time of inspection.
- Summarize the minutes of each IACUC meeting and the results of each inspection in a written report to the institutional officials. Identify significant deficiencies, if any, and a specific schedule and plan for correction. Report uncorrected deficiencies to APHIS within 15 working days after scheduled correction date.

Annual Report to APHIS

- Submit required data to APHIS by December 1 of each year on their report form (see footnote 11). Provide additional information about animal care and handling to APHIS, other agencies, and the public, as requested. Public information is considered to be any written report, summaries of data, or data sets that can be reasonably and efficiently retrieved, copied, and distributed to the requesting party. Raw data, individual field forms, or other unanalyzed field information also may be available when it can be efficiently copied and distributed in a comprehensive form, and when such distribution does not interfere with its analysis by research personnel.

Courtesy Inspections by APHIS

- Invite the State Regulatory Enforcement and Animal Care (REAC) Veterinary Inspector for APHIS to attend the biannual meetings and inspections of the Starkey and NCASI sites. Treat any findings, deficiencies, or recommendations made by the State Inspector like those of the IACUC or the attending veterinarian.

Acknowledgments

Bill Brown, Larry Bryant, Jerry DePoyster, Bruce Johnson, Ryan Kennedy, Terry McCoy, and Jack Thomas reviewed the manuscript and provided important comments about the protocols.

Literature Cited

Bull, Evelyn L.; Wisdom, Michael J. 1992. Fauna of the Starkey Experimental Forest and Range. Gen. Tech. Rep. PNW-GTR-291. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 28 p.

California Department of Fish and Game, Wildlife Investigations Laboratory. 1992. Wildlife restraint handbook. Rancho Cordova, CA. 1-1 to 17-1.

Day, Gerald I.; Shemnitz, Sanford D.; Taber, Richard D. 1980. Capturing and marking wild animals. In: Shemnitz, Sanford D., ed. Wildlife management techniques. 4th ed. Washington, DC: The Wildlife Society: 61-88.

Deming, O.V. 1954. Rearing bighorn lambs in captivity. California Fish and Game. 40: 131-143.

Follis, Thomas B.; Spillett, J. Juan. 1974. Winter pregnancy rates and subsequent fall cow/calf ratios in elk. Journal of Wildlife Management. 38(4): 789-791.

Fowler, M.E. 1989. Restraint and handling of wild and domestic animals. Ames, IA: Iowa State University Press. 332 p.

Franklin, J.F.; Dyrness, C.T. 1973. Natural vegetation of Oregon and Washington. Gen. Tech. Rep. PNW-8. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Forest and Range Experiment Station. 417 p.

Garbe, Jo Anne L.; Wywialowski, Alice. 1991. The role of the Federal government in humane treatment of captive wildlife. Transactions, North American Wildlife and Natural Resources Conference. 56: 372-376.

Hobbs, N. Thompson; Baker, D.L. 1979. Rearing and training elk calves for use in food habits studies. Journal of Wildlife Management. 43(2): 568-570.

Jessup, D.A.; Clark, W.E.; Gullett, P.A.; Jones, K.R. 1983. Immobilization of mule deer with ketamine and xylazine, and reversal of immobilization with yohimbine. Journal of the American Veterinary Medical Association. 183(11): 1339-1340.

Jessup, D.A.; Jones, K.; Mohr, R.; Kucera, T. 1985. Yohimbine antagonism to xylazine in free-ranging mule deer and desert bighorn sheep. Journal of the American Veterinary Medical Association. 187(11): 1251-1253.

Johnson, B.K.; Noyes, J.H.; Thomas, J.W.; Bryant, L.D. 1991. Overview of the Starkey Project: current measures of elk vulnerability. In: Christensen, A.G.; Lyon, L.J.; Lonner, T.N., comps.: Proceedings of the elk vulnerability symposium; 1991 April 10-12; Bozeman, MT. Bozeman, MT: Montana State University: 225-228.

Johnson, D.E. 1951. Biology of the elk calf, *Cervus canadensis nelsoni*. Journal of Wildlife Management. 15: 396-410.

Krzywinski, A.; Krzywinska, K.; Kisza, J. [and others]. 1980. Milk composition, lactation, and the artificial rearing of red deer. Acta Theriologica. 25: 341-347.

Leckenby, D.A. 1984. Elk use and availability of cover and forage components in the Blue Mountains, northeast Oregon, 1976-1982. Wildl. Res. Rep. 14. Portland, OR: Oregon Department of Fish and Wildlife. 40 p.

Neil, P.H.; Woodard, T.N.; Baker, D.L. 1979. Procedures for rearing wild ruminants in captivity. Game Info. Leafl. 106. Denver: Colorado Division of Wildlife. 4 p.

Parker, K.L.; Wong, B. 1987. Raising black-tailed deer fawns at natural growth rates. Canadian Journal of Zoology. 65: 20-23.

Pedersen, Richard J. 1977. Big game collar-transmitter package. Journal of Wildlife Management. 41(3): 578-579.

Pedersen, Richard J. 1985. Elk use of an unlogged and logged forest environment. Moscow, ID: University of Idaho. 142 p. Ph.D. dissertation.

Pekins, Peter J.; Mautz, William W. 1985. A new fawn feeding schedule. Wildlife Society Bulletin. 13: 174-176.

Reichert, D.W. 1972. Rearing and training deer for food habits studies. Res. Note RM-208. Fort Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 7 p.

Robbins, Charles T.; Oftedal, O.T.; O'Rourke, K.I. 1987. Lactation, early nutrition, and hand-rearing of wild ungulates, with special reference to deer. In: Wemmer, C.M., ed. Biology and management of the Cervidae: Research symposia of the National Zoological Park; [Date of meeting unknown]; [Location of meeting unknown]. Washington, DC: Smithsonian Institution Press: 429-442.

Robbins, Charles T.; Podbielancik-Norman, Roberta S.; Wilson, David L.; Mould, Eric D. 1981. Growth and nutrient consumption of elk calves compared to other ungulate species. Journal of Wildlife Management. 45(1): 172-186.

Sadlier, R.M.R.S. 1980. Energy and protein intake in relation to growth of suckling black-tailed deer fawns. Canadian Journal of Zoology. 58: 1347-1354.

Schwartz, Charles C.; Nagy, Julius G.; Kerr, Stephen M. 1976. Rearing and training pronghorns for ecological studies. Journal of Wildlife Management. 40(3): 464-468.

Skovlin, J.M. 1991. Fifty years of research progress: a historical document of the Starkey Experimental Forest and Range. Gen. Tech. Rep. PNW-GTR-266. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 58 p.

Smits, J.E.G. 1991. A brief review of infectious and parasitic diseases of wapiti, with emphasis on western Canada and the northwestern United States. Canadian Veterinary Journal. 32: 471-479.

Strickler, G.S. 1965. The flora of the Starkey Experimental Forest and Range. Special Res. Pap. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Forest and Range Experiment Station. 85 p.

Thomas, J.W. 1989. The Starkey research project: interactions of elk, deer, cattle, and timber management in fully managed National Forests of the future. In: Proceedings of the 1989 national convention of the Society of American Foresters; 1989 September 24-27; Spokane, WA. Bethesda, MD: Society of American Foresters: 141-143.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1989. Animal welfare; final rules. In: U.S. Federal Register, 9 CFR Parts 1, 2, and 3. Vol. 54, No. 168. August 31, 1989: 36112-36163.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1991. Licensing and registration under the Animal Welfare Act: guidelines for dealers, exhibitors, transporters, and researchers. Fact Sheet. Washington, DC. 9 p.

U.S. Laws, Statutes, etc.; Public Law 99-198. Animal Welfare Act; Sections 1751-1759 of the Food Security Act of 1985. Act of Dec. 23, 1985. 7 U.S.C. 2131-2157.

Wisdom, Michael. 1992. The Starkey Project: new technologies chase old questions about deer and elk management. Western Wildlands. Missoula, MT: Montana Forestry and Conservation Experiment Station, University of Montana; 18: 32-38.

Wood, A.J.; Nordan, H.C.; Cowan, I.M. 1961. The care and management of wild ungulates for experimental purposes. Journal of Wildlife Management. 25(3): 295-302.

Youngson, R.W. 1970. Rearing red deer calves in captivity. Journal of Wildlife Management. 34(2): 467-470.

Appendix 1: Justification and Protocols for Studies on Forest Cover-Elk Energetics Relations in the Blue Mountains of Northeast Oregon

Background

The FS and National Council of the Paper Industry for Air and Stream Improvement (NCASI), a private nonprofit research organization, have begun a study about relations between forest cover and elk condition in the Blue Mountains of northeast Oregon. The primary objectives of the study are to (1) determine if thermal cover sufficiently mediates weather conditions to influence energetics of elk, and (2) validate the component of FS models now used to evaluate forest cover conditions for big game habitats (Thomas and others 1988, Wisdom and others 1986).

Justification

The practical value of thermal cover to free-ranging ungulates has been long debated among wildlife biologists and other scientists (Edge and others 1990; Peek and others 1982; Riggs and others, in press). On one hand, forest cover reduces wind, intercepts precipitation, moderates winter temperatures, and emits thermal radiation (Nyberg and others 1986, Reifsnyder and Lull 1965, Schwab and others 1987), and thus may provide important energetic benefits to big game (Black and others 1976, Nelson and Leege 1982, Parker 1987, Thomas and others 1988). Observations indicate free-ranging ungulates select areas providing thermal cover (Beall 1976, Leckenby 1984, Zahn 1985), thereby leading many biologists to conclude that thermal cover is a critical component of big game winter ranges. Based on forest cover-weather relations and results of these mensurative studies, wildlife biologists have developed habitat models that include forest cover as an evaluation criterion for big game ranges (Thomas and others 1988, Wisdom and others 1986). These models are now used to direct forest planning in the Pacific Northwest (Edge and others 1990).

In contrast, other biologists refute or question the hypothesis that thermal cover contributes energetic benefits (Freddy 1984, 1986; Hobbs 1989; Peek and others 1982; Robinson 1960). The alternative hypothesis is that the combination of insulating qualities of pelage, behavioral adjustments to reduce activity and thermal stress, heat increment of digestion, and other physiological adaptations render cover a negligible role in big game energetics. Controlled studies with deer moreover have failed to identify significant relations between cover and measures of animal condition (Freddy 1984, 1986; Gilbert and Bateman 1983; Robinson 1960).

This study will carefully examine thermal cover benefits to big game, thereby helping to clarify these issues. Physiological assessments of elk condition will provide sensitive measures of animal response to forest cover, at a level of sensitivity not included in earlier controlled studies with deer (Freddy 1984, 1986; Gilbert and Bateman 1983; Robinson 1960). In addition, no previous attempt has been made to evaluate the suitability of definitions of cover quality, included in habitat evaluation models, under controlled experimental conditions. This study will provide the first assessment of this model component.

General Study Design

About 30 tame female elk will be maintained in holding pens in habitat treatment units that have been either clearcut, partially cut, or left uncut (fig. 16). Three replicates of each treatment with two to three elk in each replicate will be included in the study. In a fourth treatment, also with three replicates and two to three elk per replicate, both uncut forests and clearcuts will be available to elk (fig. 16). Body composition, urine and blood chemistry, weight, and activity profiles of elk will be measured and compared among treatments. Experimental trials, each about 4 months long, will be conducted during summer and winter beginning in late November 1991 and ending in March 1994. Assessments will be conducted on calves, yearlings, and adults sequentially during the 3 years of the study.

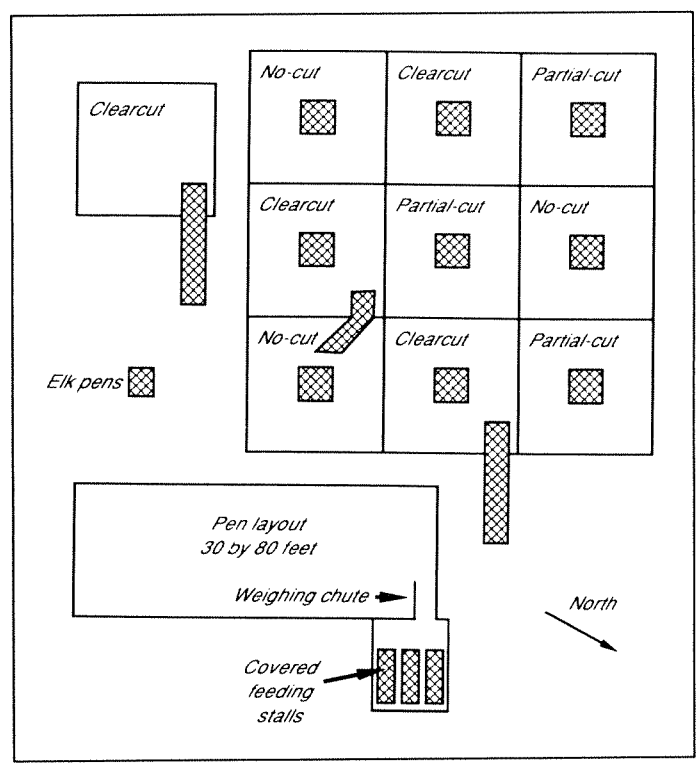


Figure 16—Tame elk are kept in nine forest cover treatment units as part of the forest cover study near Kamela, Oregon.

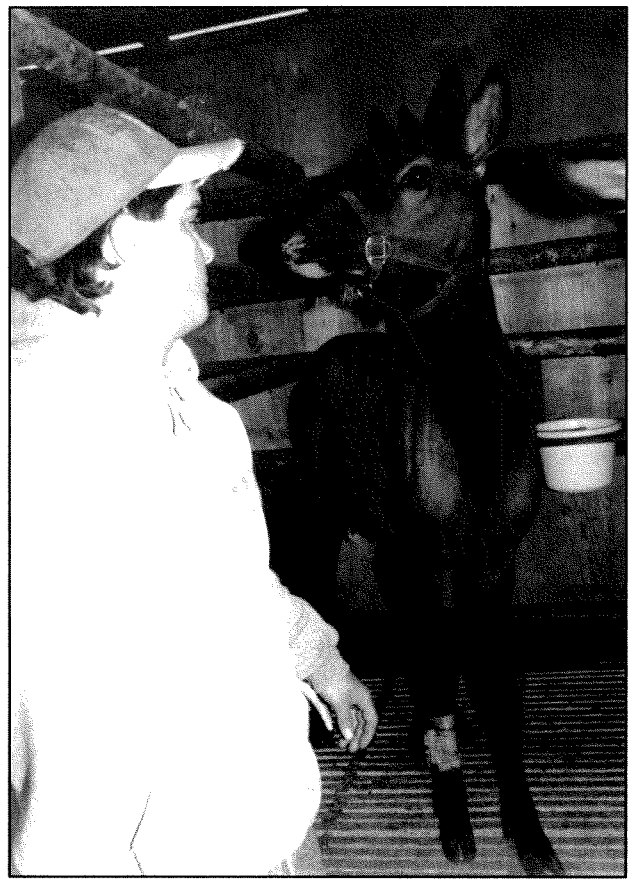


Figure 17—Covered sheds allow segregation of elk for individualized feeding in the forest cover study at Kamela; note transmitter mounted on leg to monitor daily activities.

Location and Facilities

This study will be conducted on corporate timberlands owned by Boise Cascade Corporation near Kamela, Oregon, about 15 miles north of the Starkey Experimental Forest and Range. Severe defoliation of forest canopies by spruce budworms within the Starkey fence made the area unsuitable for this experiment. The study site was chosen based on absence of forest-canopy defoliation, accessibility, and other factors discussed by Irwin and others (see footnote 3).

Elk will be maintained in pens measuring 30 by 80 feet (fig. 16). No artificial cover is provided. A small enclosed shed with three separate stalls is beside each pen (figs. 16 and 17). The sheds will permit individualized feeding of elk and collection of urine through grated floors in each stall. Animals will be allowed inside only for feeding and urine collection. No facilities to alter ambient temperatures or humidity in the sheds are provided. A weighing chute is attached to each shed, through which elk must pass each day to be fed (fig. 16). This design allows weighing without changes in daily elk-handling routines, thereby minimizing stress.

Feed and Water

Two rations have been developed, one each for winter and summer trials. Rations have been developed based on the National Research Council (NRC) guidelines for sheep and cattle (NRC 1984, 1985), with assistance of an animal nutritionist (Dr. Mike Mehrens, Hermiston, OR). Crude protein levels of the winter and summer ration average 10 percent and 15 percent, respectively, providing protein requirements slightly above seasonal requirements (NRC 1984, 1985) for nonreproducing ruminants. Energy levels of rations were designed to differ more substantially between seasons, with the summer ration averaging 70 percent Total Digestible Nutrients (TDN) and the winter ration averaging 60 percent TDN. Both rations have proven palatable to elk in previous experiments. Vitamins and minerals are added to the ration. Alfalfa hay, fed at 25 to 40 percent of total daily dry matter intake, will provide roughage. No natural forage will be available to the elk.

Feeding rates will be adjusted to mimic feeding conditions normally encountered by free-ranging elk. During winter, volume of food for calves will be slightly sub-maintenance, resulting in minor (about 5 percent) losses in weight. Volume of food for yearlings and adults will be more deficient, so that they will lose 10 to 15 percent of their body weight. We collected data during winter 1991-92 to determine maintenance feeding levels for elk calves. Feeding levels for yearlings and adults will be based on published data for livestock and deer. During summer, the higher quality ration will be fed so that growth rates are normal. Daily feeding rates in summer also will be based on published data for livestock and deer.

Elk will be fed twice each day. The primary ration will be fed during the morning in the sheds, and alfalfa hay will be fed late in the day to mimic early morning and early evening feeding patterns often observed in the wild. Covered hay feeders are provided in each pen.

Fresh water will be provided *ad libitum* except during winter when snow has accumulated within the pens. Requiring elk to consume snow during winter mimics normal winter range conditions, when unfrozen water is generally unavailable to elk and deer.

Protocols

This study will measure four variables of tame elk response to forest cover: body composition (DelGiudice and others 1990; Torbit and others 1985a, 1985b), urine and blood chemistry (DelGiudice and others 1987, 1990), weight, and activity. Protocols for each will impose relatively minor stress on experimental animals.

Body composition—Body composition will be determined for each animal at the beginning, middle, and end of each trial. Animals will be anesthetized with xylazine hydrochloride administered intramuscularly, and injected with deuterium intravenously. After equilibrium of deuterium with body fluids (about 5 hours), blood will be collected via the jugular, and analyzed to determine deuterium concentration. Yohimbine hydrochloride will be used to reverse immobilization effects of xylazine.

Urine and blood chemistry—Urine and blood samples will be collected five times during each summer and winter trial. Urine samples will be collected in the individual stalls equipped with floor grates permitting passage of urine into collection pans under the grates. Results from winter 1991-1992 indicated that elk will urinate within 2 to 4 hours after placement in the stalls, so retention time will be short. Blood samples will be collected via the jugular. Animals will be mildly sedated with a light dose of xylazine to permit blood sampling, and effects of immobilization will be reversed with yohimbine. Collecting blood samples generally requires about 15 minutes.

Weight—Weight will be measured twice each week during each trial. Weighing chutes leading into the barns will be equipped with portable electronic scales. Animals will be weighed when brought into the barn for feeding. No sedation or abnormal handling procedures will be required to weigh animals.

Activity profiles—Twenty-four-hour activity profiles will be obtained from pulse rates of leg-mounted, motion-sensitive radio transmitters (fig. 17; Riggs and others 1990). Specially designed transmitters will be mounted on the shank just above the fetlock on the right front leg. The shank will be wrapped with gauze and vet-wrap before placement of the transmitters. These transmitters are generally ignored by the experimental animals and cause no physical damage if time of attachment is restricted to less than 15 days. Attachment of transmitters generally can be done without sedation.

General immobilization procedures—Our decision to use xylazine hydrochloride reflects its high therapeutic index, availability of an antidote (yohimbine hydrochloride), and nontoxic effects in humans. We have used xylazine extensively with tame calf and yearling elk, with good results at dosages ranging from 0.05 to 0.2 milligrams per pound of body weight (1/20 to 1/5 recommended levels). And we have found yohimbine to be a satisfactory antidote, particularly when animals are lightly sedated.

Virtually all immobilizations will require light dosages of xylazine (0.05 to 0.1 milligrams per pound) and short handling times (less than 15 minutes) before the antidote is administered. Probability of complications such as hypothermia or hyperthermia is low. Animals will be monitored, and yohimbine will be administered if complications are observed. During the summer, immobilizations will be restricted to early morning hours to avoid high ambient temperatures.

Treatment of Disease

Treatment of sick animals is similar to that described under "Raising and care of tame elk."

Literature Cited in Appendix 1

Beall, Robert C. 1976. Elk habitat selection in relation to thermal radiation. In: Hieb, S.R., ed. Elk-logging-roads: Proceedings of a symposium. Moscow, ID: University of Idaho: 97-100.

Black, H., Jr.; Scherzinger, R.J; Thomas, J.W. 1976. Relationships of Rocky Mountain elk and Rocky Mountain mule deer habitat to timber management in the Blue Mountains of Oregon and Washington. In: Hieb, S.R., ed. Elk-logging-roads: Proceedings of a symposium. Moscow, ID: University of Idaho: 11-31.

DelGiudice, Glenn D.; Mech, L. David; Seal, Ulysses S. 1990. Effects of winter undernutrition on body composition and physiological profiles of white-tailed deer. Journal of Wildlife Management. 54(4): 539-550.

DelGiudice, Glenn D.; Mech, L. David; Seal, Ulysses S.; Karns, Patrick D. 1987. Effects of winter fasting and refeeding on white-tailed deer blood profiles. Journal of Wildlife Management. 51(4): 865-873.

Edge, W. Daniel; Olson-Edge, Sally L.; Irwin, Larry L. 1990. Planning for wildlife in national forests: elk and mule deer habitats as an example. Wildlife Society Bulletin. 18: 87-98.

Freddy, D. J. 1984. Quantifying capacity of winter ranges to support deer-evaluation of thermal cover used by deer. Wildl. Res. Rep. Denver: Colorado Division of Wildlife: 21-24.

Freddy, D. J. 1986. Quantifying capacity of winter ranges to support deer-evaluation of thermal cover used by deer. Wildl. Res. Rep. Denver: Colorado Division of Wildlife: 9-18.

Gilbert, F.F.; Bateman, M.C. 1983. Some effects of winter shelter conditions on white-tailed deer, *Odocoileus virginianus.* Canadian Field Naturalist. 97: 391-400.

Hobbs, N. Thompson. 1989. Linking energy balance to survival in mule deer: development and test of a simulation model. Wildl. Monogr. 101. Bethesda, MD: The Wildlife Society. 39 p. Supplement to Journal of Wildlife Management. 53(2): 1989 April.

Leckenby, D.A. 1984. Elk use and availability of cover and forage habitat components in the Blue Mountains, northeast Oregon, 1976-1982. Wildl. Res. Rep. 14. Portland, OR: Oregon Department of Fish and Wildlife. 40 p.

National Research Council. 1984. Nutrient requirements of beef cattle. Washington, DC: National Academy Press. 90 p.

National Research Council. 1985. Nutrient requirements of sheep. Washington, DC: National Academy Press. 99 p.

Nelson, J.R.; Leege, T.A. 1982. Nutrition and food habits. In: Thomas, J.W.; Toweill, Dale, eds. Elk of North America—ecology and management. Harrisburg, PA: Stackpole Books: 323-369. Chapter 8.

Nyberg, J.B.; Bunnell, F.L.; Janz, D.W.; Ellis, R.M. 1986. Managing young forests as black-tailed deer winter ranges. Land Manage. Rep. No. 37. Victoria, BC: Information Services Branch, Ministry of Forests. 49 p.

Parker, K.L. 1987. Effects of heat, cold, and rain on coastal black-tailed deer. Canadian Journal of Zoology. 66: 2475-2483.

Peek, J.M.; Scott, M.D.; Nelson, L.J. [and others]. 1982. The role of cover in habitat management for big game in northwestern United States. Transactions, North American Wildlife Natural Resources Conference. 47: 363-373.

Reifsnyder, William E.; Lull, Howard W. 1965. Radiant energy in relation to forests. Tech. Bull. No. 1344. Washington, DC: U.S. Department of Agriculture, Forest Service. 111 p.

Riggs, Robert A.; Urness, Philip J.; Gonzalez, Karen A. 1990. Effects of domestic goats on deer wintering in Utah oakbrush. Journal of Range Management. 43(3): 229-234.

Riggs, R.A.; Cook, J.G.; Irwin, L.L. [In press]. Cover and timber management on interior northwest winter range: some thoughts on reducing conflict. In: Proceedings of the western states and provinces elk workshop; 1990 May 15; Arcata, CA. [Place of publication unknown]: [Publisher unknown].

Robinson, William L. 1960. Test of shelter requirements of penned white-tailed deer. Journal of Wildlife Management. 24(4): 364-371.

Schwab, Francis E.; Pitt, Michael D.; Schwab, Susan W. 1987. Browse burial related to snow depth and canopy cover in northcentral British Columbia. Journal of Wildlife Management. 51(2): 337-342.

Thomas, Jack Ward; Leckenby, Donavin A.; Henjum, Mark [and others] . 1988. Habitat-effectiveness index for elk on Blue Mountain winter ranges. Gen. Tech. Rep. PNW-GTR-218. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 28 p.

Torbit, Stephen C.; Carpenter, Len H.; Alldredge, A.W.; Swift, David M. 1985a. Mule deer body composition—a comparison of methods. Journal of Wildlife Management. 49(1): 86-91.

Torbit, Stephen C.; Carpenter, Len H.; Swift, David M.; Alldredge, A.W. 1985b. Differential loss of fat and protein by mule deer during winter. Journal of Wildlife Management. 49(1): 80-85.

Wisdom, M.J.; Bright, L.R.; Carey, C.G. [and others]. 1986. A model to evaluate elk habitat in western Oregon. Publ. No. R6-F&WL-216-1986. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Region. 35 p.

Zahn, H.M. 1985. Use of thermal cover by elk (*Cervus elaphus*) on a western Washington summer range. Seattle: University of Washington. 175 p. Ph.D. dissertation.

Appendix 2: Definition of Terms

Terms that are relevant to care and handling of animals at the Starkey Experimental Forest and Range are defined in the CFR Standards (USDA APHIS 1989) for implementing the Animal Welfare Act (U.S. Laws, Statutes, etc. 1985). Selected definitions are quoted in the following list.

Activity: Those elements of research, testing, or teaching procedures that involve the care and use of animals.

Animal: Any live or dead dog, cat, nonhuman primate, guinea pig, hamster, rabbit, or any other warm-blooded animal, which is being used, or is intended for use for research, teaching, testing, experimentation, or exhibition purposes, or as a pet. This term excludes: Birds, rats of the genus *Rattus* and mice of the genus *Mus* bred for use in research, and horses and other farm animals, such as, but not limited to, livestock and poultry, used or intended for use as food or fiber, or livestock or poultry used or intended for use for improving animal nutrition, breeding, management, or production efficiency, or for improving the quality of food or fiber. With respect to a dog, the term means all dogs including those used for hunting, security, or breeding purposes.

APHIS: The Animal and Plant Health Inspection Service, United States Department of Agriculture.

APHIS, Regulatory Enforcement and Animal Care, Animal Care Sector Supervisor: A veterinarian or his designee, employed by APHIS, who is assigned by the administrator to supervise and perform the official work of APHIS in a given State or States.

Attending Veterinarian: A person who has graduated from a veterinary school accredited by the American Veterinary Medical Association Council on Education, or has a certificate issued by the American Veterinary Medical Association's Education Commission for Foreign Veterinary Graduates, or has received equivalent formal education as determined by the Administrator; has received training or experience in the care and management of the species being attended; and who has direct or delegated authority for activities involving animals at a facility subject to the jurisdiction of the Secretary.

Committee: The Institutional Animal Care and Use Committee (IACUC) established under section 13(b) of the Act. It shall consist of at least three (3) members, one of whom is the attending veterinarian of the research facility and one of whom is not affiliated in any way with the facility other than as a member of the committee, however, if the research facility has more than one Doctor of Veterinary Medicine (DVM), another DVM with delegated program responsibility may serve. The research facility shall establish the Committee for the purpose of evaluating the care, treatment, housing, and use of animals, and for certifying compliance with the Act by the research facility.

Euthanasia: The humane destruction of an animal accomplished by a method that produces rapid unconsciousness and subsequent death without evidence of pain or distress, or a method that utilizes anesthesia produced by an agent that causes painless loss of consciousness and subsequent death.

Federal Research Facility: Each department, agency, or instrumentality of the United States which uses live animals for research or experimentation.

Field Study: Any study conducted on free-living wild animals in their natural habitat, which does not involve an invasive procedure, and which does not harm or materially alter the behavior of the animals under study.

Handling: Petting, feeding, watering, cleaning, manipulating, loading, crating, shifting, transferring, immobilizing, restraining, treating, training, working and moving, or any similar activity with respect to any animal.

Housing Facility: Any land, premises, shed, barn, building, trailer, or other structure or area housing or intended to house animals.

Indoor Housing Facility: Any structure or building with environmental controls housing or intended to house animals and meeting the following three requirements:

1. It must be capable of controlling the temperature within the building or structure within the limits set forth for that species of animal, of maintaining humidity levels of 30 to 70 percent and of rapidly eliminating odors from within the building; and
2. It must be an enclosure created by the continuous connection of a roof, floor, and walls (a shed or barn set on top of the ground does not have a continuous connection between the walls and the ground unless a foundation and floor are provided); and
3. It must have at least one door for entry and exit that can be opened and closed (any windows or openings which provide natural light must be covered with a transparent material such as glass or hard plastic).

Inspector: Any person employed by the Department who is authorized to perform a function under the Act and the regulations in 9 CFR parts 1, 2, and 3.

Institutional Official: The individual at a research facility who is authorized to legally commit on behalf of the research facility that the requirements of 9 CFR parts 1, 2, and 3 will be met.

Licensed Veterinarian: A person who has graduated from an accredited school of veterinary medicine or has received equivalent formal education as determined by the Administrator, and who has a valid license to practice veterinary medicine in some state.

Major Operative Procedure: Any surgical intervention that penetrates and exposes a body cavity or any procedure which produces permanent impairment of physical or physiological functions.

Outdoor Housing Facility: Any structure, building, land, or premise, housing or intended to house animals, which does not meet the definition of any other type of housing facility provided in the regulations, and in which temperatures cannot be controlled with set limits.

Painful Procedure: Any procedure that would reasonably be expected to cause more than slight or momentary pain or distress in a human being to which that procedure was applied; that is, pain in excess of that caused by injections or other minor procedures.

Paralytic Drug: A drug causing partial or complete loss of muscle contraction and which has no anesthetic or analgesic properties, so that the animal cannot move, but is completely aware of its surroundings and can feel pain.

Positive Physical Contact: Petting, stroking, or other touching, which is beneficial to the well-being of the animal.

Primary Enclosure: Any structure or device used to restrict an animal or animals to a limited amount of space, such as a room, pen, run, cage, compartment, pool, hutch, or tether. In the case of animals restrained by a tether (e.g., dogs on chains), it includes the shelter and the area within reach of the tether.

Principal Investigator: An employee of a research facility, or other person associated with a research facility, responsible for a proposal to conduct research and for the design and implementation of research involving animals.

Quorum: A majority of the Committee members.

Research Facility: Any school (except an elementary or secondary school), institution, organization, or person that uses or intends to use live animals in research, tests, or experiments, and that (1) purchases or transports live animals in commerce, or (2) receives funds under a grant, award, loan, or contract from a department, agency, or instrumentality of the United States for the purpose of carrying out research, tests, or experiments: Provided, That the Administrator may exept, by regulation, any such school, institution, organization, or person that does not use or intend to use live dogs or cats, except those schools, institutions, organizations, or persons, which use substantial numbers (as determined by the Administrator) of live animals the principal function of which schools, institutions, organizations, or persons, is biomedical research or testing, when in the judgment of the Administrator, any such exemption does not vitiate the purpose of the Act.

Sanitize: To make physically clean and to remove and destroy, to the maximum degree that is practical, agents injurious to health.

Sheltered Housing Facility: A housing facility which provides the animals with shelter; protection from the elements; and protection from temperature extremes at all times. A sheltered housing facility may consist of runs or pens totally enclosed in a barn or building, or of connecting inside/outside runs or pens with the inside pens in a totally enclosed building.

Standards: The requirements with respect to the humane housing, exhibition, handling, care, treatment, temperature, and transportation of animals by dealers, exhibitors, research facilities, carriers, intermediate handlers, and operators of auction sales.

Study Area: Any building room, area, enclosure, or other containment outside of a core facility or centrally designated or managed area in which animals are housed for more than 12 hours.

Wild Animal: Any animal which is now or historically has been found in the wild, or in the wild state, within the boundaries of the United States, its territories, or possessions. This term includes, but is not limited to animals such as: Deer, skunk, opossum, raccoon, mink, armadillo, coyote, squirrel, fox, wolf.

Wild State: Living in its original, natural condition, not domesticated.

*U.S. GOVERNMENT PRINTING OFFICE: 1993-790-301/80015

Wisdom, Michael J.; Cook, John G.; Rowland, Mary M.; Noyes, James H. 1993. Protocols for care and handling of deer and elk at the Starkey Experimental Forest and Range. Gen. Tech. Rep. PNW-GTR-311. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 49 p.

Several hundred Rocky Mountain elk (*Cervus elaphus nelsoni* V. Bailey) and Rocky Mountain mule deer (*Odocoileus hemionus hemionus* Rafinesque) inhabit a fenced, 25,000-acre enclosure at the Starkey Experimental Forest and Range in the Blue Mountains of northeast Oregon. Research there requires handling most of these animals each winter. In addition, 33 elk calves have been captured and raised for research. Protocols for care and handling of deer and elk are described. Legal requirements for the operation of facilities and research within the enclosure also are discussed.

Keywords: Elk, mule deer, animal welfare, Starkey Experimental Forest and Range, Blue Mountains (Oregon), tame elk.

The **Forest Service** of the U.S. Department of Agriculture is dedicated to the principle of multiple use management of the Nation's forest resources for sustained yields of wood, water, forage, wildlife, and recreation. Through forestry research, cooperation with the States and private forest owners, and management of the National Forests and National Grasslands, it strives—as directed by Congress—to provide increasingly greater service to a growing Nation.

The U.S. Department of Agriculture is an Equal Opportunity Employer. Applicants for all Department programs will be given equal consideration without regard to age, race, color, sex, religion, or national origin.

Pacific Northwest Research Station
333 S.W. First Avenue
P.O. Box 3890
Portland, Oregon 97208-3890

U.S. Department of Agriculture
Pacific Northwest Research Station
333 S.W. First Avenue
P.O. Box 3890
Portland, OR 97208

Official Business
Penality for Private Use, $300

do NOT detach label