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Humane and efficient capture and handling methods for carnivores

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To be effective, conservation and management programs for carnivores require a good understanding of the animals' biology, ecology, behavior, and habitat requirements. To gather scientific information essential to the development of such programs, it is often necessary to capture, handle, and mark animals, a controversial and complex activity (Proulx and Barrett 1989) requiring special skills to minimize negative effects on individuals and populations, and to maximize scientific gains. For this, researchers should use methods that are consistent with codes of ethics and guidelines published by professional societies and countries (Table 5.1). They should continuously improve capture procedures and equipment to work more effectively and more safely for both animals and people (Powell and Proulx 2003). Research design should minimize both potential short-term and long-term effects of capture (Seddon *et al.* 1999, Cattet *et al.* 2008a), and deal with non-random sampling that may affect population structures (Banci and Proulx 1999).

Here, we discuss trap types, sets, and efficiency, and describe humaneness criteria that we use in the selection of specific carnivore traps. We review use of drugs as a primary method of capture through chemical immobilization, but also as a means to support mechanical capture methods by reducing stress and pain. Our approach results in some redundancy but minimizes confusion because different techniques can be used for the same group of carnivores, similar traps and anesthetics may be used for different mammals, and methods that meet performance criteria for one species may not for others. Lastly, we summarize complications that can occur with capture and handling, methods of humane killing, and techniques for restraining and marking carnivores.

Table 5.1 *Non-exhaustive list of codes of ethics and guidelines on animal welfare (all websites were accessed in were accessed in August 2011).*

Code of ethics/Guidelines	Organization/ Country	Reference
Guidelines for the Capture, Handling, and Care of Mammals as Approved by the American Society of Mammalogists	ASM Animal Care and Use Committee 1998	http://www.mammalsociety.org/committees/animal-care-and-use (last accessed 1 July 2011)
Guidelines for the Treatment of Animals in Behavioral Research and Teaching	ASAB/ABS 2000	http://www.animalbehavior.org/ABSHandbook/animal-behavior-society-handbook/27ABSASABGuidelinesForTheTreatmentOfAnimalsInBehavioralResearchAndTeaching
CCAC Guidelines: the care and use of wildlife	CCAC 2011	http://www.ccac.ca/Documents/Standards/Guidelines/Wildlife.pdf
Directive 86/609/EEC on the Approximation of Laws, Regulations and Administrative Provisions of the Member States Regarding the Protection of Animals Used for Experimental and Other Scientific Purposes.	European Union 1986	http://eur-lex.europa.eu/LexUriServ/LexUriServ.do?uri=CELEX:31986L0609:EN:HTML
Animal (Scientific procedures) Act in the United Kingdom	HMSO 1986	http://www.archive.official-documents.co.uk/document/hoc/321/321-xa.htm
Ethical Principles and Guidelines for Experiments on Animals	Swiss Academy of Medical Sciences 2005	http://www.scnat.ch/downloads/Ethik_Tiervers_Nov05_e.pdf
Animal Welfare Act	United States Department of Agriculture	http://www.nal.usda.gov/awic/legislat/awa.htm
Ethical Principles and Guidelines for the Use of Animals	National Research Council of Thailand 1999	http://ird.sut.ac.th/newsite/Form/pet_ethic_eng.pdf
Endangered Species Act	United States Fish & Wildlife Service 1973	http://www.fws.gov/laws/lawsdigest/esact.html

5.1 Mechanical capture methods

5.1.1 Traps and sets

Restraining traps allow captured animals to be released and include cage traps, foothold traps, foot, neck and body snares, and nets (Appendix 5.1). Killing traps include neck snares, and snap, planar, rotating-jaw, and killing box traps, and submarine traps (Appendix 5.1).

Diverse sets exist to capture carnivores (Appendix 5.2). Trap design, preparation, and sets affect trapping efficiency (target captures/trap-night; Boggess *et al.* 1990), and selectivity (number of non-target species). The tripping force of the trigger must match the size of target animals. For example, by setting pan tension on foothold traps at 1.4–1.8 kg, kit foxes (*Vulpes macrotis*) may be excluded from traps set for coyotes (*Canis latrans*, Phillips and Gruver 1996). A trap with a light tripping force may capture carnivores of all sizes and not be efficient due to low selectivity. To increase trapping efficiency, parts of a trap may be modified to control access to the triggering system. For example, a bionic trap with a 6-cm high bait cone aperture will capture small carnivores, such as minks (*Neovison vison*, Proulx and Barrett 1991a), but will restrict access by larger carnivores, such as fishers (*Martes pennanti*, Proulx and Barrett 1993a). The shape and size of triggers can discourage some carnivores from entering a trap. For example, the efficiency of C120 Magnum rotating-jaw traps to capture American martens (*Martes americana*) is higher with one-way four prong triggers, where the central prongs are shorter than the outside ones (Barrett *et al.* 1989), than with pitchfork triggers with four long prongs of equal length (e.g. Naylor and Novak 1994) that interfere with martens' movements (Pawlina and Proulx 1999).

The position of traps in sets also may affect capture efficiency. For example, lynxes (*Lynx canadensis*) can be properly killed by a blow to the neck by placing rotating-jaw traps at least 23 cm above ground and centered in line with bait at the back of a cubby (Proulx *et al.* 1995). With traps set too low, lynxes try unsuccessfully to go over the trap or lose interest in the bait. With a trap set higher but not centered in line with the bait, a lynx may reach for the bait with a front paw, inadvertently firing the trap on its limb.

A trap must be sited carefully to capture carnivores efficiently without causing undue injury. An animal caught in an EGG trap set in a hole dug into a stream bank can injure itself by wrapping the trap anchor cable around something solid and pulling on the captured foot (Hubert *et al.* 1996). Injuries can also occur when foot-snared canids, felids, and ursids become entangled in surrounding vegetation (Mowat *et al.* 1994; Powell 2005).

Baited sets use food or scent to draw target animals to a trap, while trail (i.e. blind) sets are placed where target animals are expected to travel on their own (Powell and Proulx 2003). While baited traps may have higher capture rates for carnivores, they also attract non-target animals.

5.1.2 Trapping efficiency

Trap models and sets, baits and lures, trappers' experience, weather, and biological variables affect trap efficiency (Pawlina and Proulx 1999). Weakened springs (Gruver *et al.* 1996), distorted components (Warburton 1982), and poorly made traps (Linhart *et al.* 1986) affect trap performance. Traps of different generations or manufacturers may have different components. For example, even though the Novak and the Fremont foot snares are similar in design, the latter is markedly more efficient in capturing coyotes (Skinner and Todd 1990). Red foxes (*Vulpes vulpes*) may smell rusty or oily traps, discover traps that move when a fox steps on jaws or springs, and shy from a set that does not provide a clear view (Krause 1989).

Whether baits and lures increase capture efficiency is either unknown or variable for many conditions. Baits compete with odors of natural foods to attract carnivores (Linhart and Knowlton 1975; Humphrey and Zinn 1982). Scent lures may mimic pheromones (Carde and Elkinton 1984) or stimulate curiosity. Their effectiveness is affected by weather, as well as the physiological condition of target carnivores and the animal that is the source of the scent (Pawlina and Proulx 1999).

Trapping efficiency changes with a trapper's experience. Trappers may require a 1-year acclimatization period before becoming proficient with new trapping devices (Skinner and Todd 1990; Pawlina and Proulx 1999).

Weather may interfere with, or enhance, trap operation and affect the behavior of the target species. For example, frozen soil may affect rubber-padded foothold traps set for coyotes more than unpadded ones (Linhart *et al.* 1986) and wind direction affects food detection by dingos (*Canis familiaris dingo,* Joly and Joly 1992).

Finally, biological variables affect capture efficiency. If traps are located diffusely over large areas, they may be absent from small home-ranges (Gehrt and Fritzell 1996). If males and females have home ranges of different size, trap density will affect the sex ratio of captured animals (King and Powell 2007), and when changing resources lead to changes in the sizes of home ranges, capture efficiency changes (Smith *et al.* 1994). Also, animals of different sex often behave differently towards traps and sets (Gehrt and Fritzell 1996). Some animals become trap-shy after initial capture, while others become trap-happy (Pawlina and Proulx 1999). Resident or dominant individuals may intimidate intruders or subordinates with their scent marks, affecting capture rate (Pawlina and Proulx 1999). Finally, life-history condition may affect capture. For example, adult coyotes may be captured more often when rearing pups (Sacks *et al.* 1999).

5.1.3 Humaneness

Killing and restraining traps used to capture carnivores should be humane and either cause unconsciousness as quickly as possible or hold animals with minimal injury and stress.

For state-of-the-art killing traps, we adopt the following criterion, established by Proulx and Barrett (1994):

> Criterion I: at a 95% confidence level, humane killing traps should render $\geq$ 70% of target animals irreversibly unconscious in $\leq$ 3 minutes.

Powell and Proulx (2003) showed that, despite solid technical advances in trap research and development that meet Proulx and Barrett's (1994) criterion, recently developed standards (CGSB 1996, European Community *et al.* 1997) had not completely incorporated those technical advances. Also, instead of adhering to humane trapping standards, the United States developed its own best-management practices on the basis of technical, economical, and social criteria (International Association of Fish and Wildlife Agencies 1997). Nevertheless, Proulx and Barrett's (1994) criterion for killing traps still is the best-defined, objective, and published criterion consistent with state-of-the-art technological development.

For restraining traps, Tullar (1984), Olsen *et al.* (1986), Hubert *et al.* (1996), and others (summarized by Proulx 1999a) developed injury-scoring systems, most of which correspond with pathological changes in captured animals. Over the years, the number of injury classes has increased and, while early scores were based solely on the injuries of captured limbs, more recent injury-scoring systems also include whole-body trap-related trauma (Proulx *et al.* 1993a; Hubert *et al.* 1996). In all systems, injuries that have the potential to decrease the survival of released animals were identified and a 50-point threshold was used to separate humane restraining devices from unacceptable ones (Proulx 1999a). Although captured animals experience behavioral and physiological changes (Kreeger *et al.* 1990b; Proulx *et al.* 1993a; Seddon *et al.* 1999; Cattet *et al.* 2003, 2008a), to date no objective scoring system for restraining traps integrate these changes with physical injuries (Proulx 1999a), at least in part because interpreting such responses is not straightforward (Dawkins 1998). On the basis of Proulx *et al.*'s (1993a) live-trapping tests with raccoons (*Procyon lotor*) in enclosures, and Powell and Proulx's (2003) humane criterion, which does not specify a maximum time of restraint, we adopt the following standard for restraining traps:

Criterion II: at a 95% confidence level, humane restraining traps should hold $\geq$ 70% of animals for $\leq$ 24 hours with $\leq$ 50 points scored for physical injury.

We recommend that this standard be used for the live capture of carnivores, because it will exceed recent national and international standards, which are not as rigorous and fail to integrate state-of-the-art technological advancements.

All killing and restraining traps should be monitored within a 24-hour period to minimize pain and discomfort. Reducing the time that animals spend in foothold traps greatly reduces injuries (Proulx *et al.* 1994). Unless traps can be visited easily, in person, and multiple times daily, they should be equipped with a monitor (Nolan *et al.* 1984; Marks 1996; Larkin *et al.* 2003; Ó Néill *et al.* 2007) that allows false positives but not false negatives, and that notifies a researcher when battery power is low or when a trap has misfired (Powell and Proulx 2003). Remotely monitored traps must, nonetheless, be visited regularly for maintenance; animals avoiding capture may disturb a trap site and render the set ineffective. The mere fact that animals are dead when kill-traps are checked is not evidence that traps are humane, especially if traps are checked only once every 24 hours (Proulx and Barrett 1989). Without knowing a priori whether traps generate enough energy to kill target animals, whether traps consistently strike animals in appropriate locations for a quick kill, and how long trapped animals remain alive, assuming that traps are humane, can lead to undue suffering.

5.1.4 Traps and sets for specific carnivores

Both restraining and killing traps can contribute significantly to research on evolution, ecology, animal behavior, physiology, parasitology, genetics, and other disciplines. The choice of restraining vs. killing traps depends, at the least, on research hypotheses and goals, research design, and study site (Powell and Proulx 2003). Because restraining traps allow the release of trapped animals, they should be used when species-at-risk and pets may be captured. When non-target captures are unlikely, using a restraining trap to capture a target carnivore, only to kill it later (to collect a sample, for example), may be less humane than using a quick-killing trap (Powell and Proulx 2003). Keeping animals alive may be required, however, to avoid freezing or decomposition of tissues to be sampled (Kreeger *et al.* 1990b).

Common sense dictates choosing traps that maximize both selectivity and efficiency (Pawlina and Proulx 1999). Selective, efficient traps minimize the capture of non-target species or individuals, thereby increasing the rate of data collection and reducing the overall impact of the research on the ecological community in the study area. Thus, within the constraints of research design, choose traps based on selectivity, efficiency, and state-of-the-art trapping technology based on

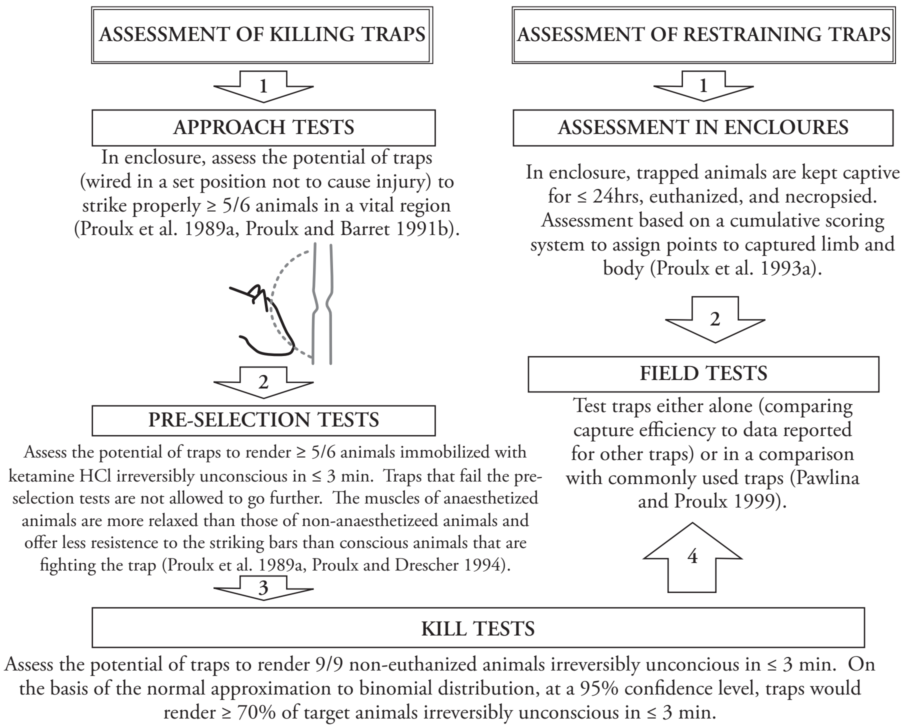


Fig. 5.1 Sequential series of biological tests used to assess the humaneness of killing and restraining traps (after Proulx and Barrett 1991b; Proulx *et al.* 1993a).

humaneness. Both efficiency and humaneness must be properly evaluated through sound, scientific protocols (Proulx 1999a), preferably peer-reviewed and published. We evaluate traps here on the basis of published data about capture efficiency and humaneness.

Proulx and Barrett (1991b) described a sequence of biological tests to develop and to evaluate killing traps (Figure 5.1). These tests were carried out in simulated environments (along with mechanical evaluations of trap properties), and they led to the development of most of the state-of-the-art killing traps identified here. Proulx *et al.* (1993a) developed a protocol to assess restraining traps (Figure 5.1), where animals are left in the trap for $\leq$ 24 hours, unless there is evidence of serious trauma. Enclosure tests must be followed by field tests to assess humaneness and capture efficiency fully (Pawlina and Proulx 1999).

The performance of a trap in the field depends on how the trap is set and monitored.

When using killing or restraining traps that are efficient and humane, follow these rules:

1. Do not modify trap size, shape, components, materials, or power, which are essential to achieve a humane kill.
2. Do not modify trigger shape or operation, which affect both humaneness and capture efficiency.
3. Replicate sets that have been used in the assessment of humane traps.
4. Visit traps <24 hours (but preferably <12 hours) after setting them (a) to kill animals that may be seriously injured but are still alive in a killing trap or (b) to release animals captured in restraining traps. No matter how humane a restraining trap is, if it is not visited at short intervals, animals will be injured.

Responsible professionals must strive continuously to improve traps to work more efficiently, more selectively, more humanely, and more safely for both animals and people. Changing the properties of traps, however, may affect the humaneness and capture efficiency of models that meet our criteria. Therefore, modified traps should be re-evaluated.

Finally, safety to the researcher should be kept in mind when developing and assessing traps. In most cases, a locking device can be installed to stop springs from firing or trap jaws from closing. If a trap cannot be safely handled without some safety device, it should not be used.

For this chapter, we reviewed traps on a species-specific basis to describe how and when they have been used, and to determine their advantages and limitations (Appendix 5.3). Wherever possible, we provide examples for all carnivore families, but information about humaneness and capture efficiency for trapping devices is often lacking. On the other hand, by matching size and behaviors of carnivores, one can often predict which trapping device is most likely to be effective for the capture of a species for which little information exists. In general, humane and capture-efficient killing traps and cage traps are available for small- and medium-sized (<5 kg) carnivores. Large carnivores must be captured in rubber-padded foothold traps or cage/box/log traps. Raccoons should be captured in EGG or cage traps. For the majority of canids, foothold traps, foot snares or neck snares are the only devices that are efficient and humane. Small felids (cat, bobcat, lynx, and others) may be captured in cage traps. Foothold traps and foot snares can be effective and humane for all felids and ursids.

Injury caused by cage traps has not been adequately evaluated (Proulx 1999a). In general, cage traps meet Criterion II for the capture of all carnivore species, as they appear to cause less trauma than other restraining techniques (White *et al.* 1991). Their capture efficiency varies among species, and is often lower than other restraining traps with canids (e.g. Muñoz-Igualada *et al.* 2002; Shivik *et al.* 2005). Regardless of the species being trapped, special precautions must be taken to ensure the well-being of captured animals. Carnivores may break teeth and cut their mouths while biting wire-mesh walls (Rust 1968; Belant 1992). Cages with small mesh holes or with solid walls usually are superior to mesh with large holes that allow animals to catch their muzzles. Cage traps without insulated nest boxes and bedding should not be used when temperatures drop below –20°C, or if researchers cannot check traps daily. Warm, dry bedding (e.g. raw wool with natural lanolin) in live-traps can reduce mortalities (Powell and Proulx 2003). Traps should be concealed and covered with vegetation to protect carnivores from direct sunlight, rain, and large predators.

5.2 Use of drugs for capture and restraint of carnivores

Drugs are powerful tools used for capture and restraint of carnivores, and to relieve pain and stress.

5.2.1 Drug access, storage, and handling

Regulations for drugs vary considerably from one country (and states within countries) to the next. Information can usually be obtained by consulting with a local veterinarian working with wildlife or zoo animals.

Although some studies have shown that potency and safety persists well past expiration dates for some drugs (Kreeger *et al.* 1990a; Kreeger and Arnemo 2007), we strongly recommend using non-expired drugs for capture of free-ranging carnivores, to minimize unpredictable variations in drug response and to be fully compliant with regulations. Drug manufacturers provide instructions for appropriate storage of their products with attention to factors such as temperature, humidity, and light exposure.

Storing drugs in a secure and safe place is important to prevent theft for illicit use (Woodward 2005), and to enable accurate inventory in order to know when to order fresh stocks and to dispose of old ones. A running inventory record should have standard information for purchases and use (Cattet *et al.* 2005). Capture records that document drug use and animal response on a case-by-case basis should be maintained to evaluate the effectiveness and safety of drug protocols.

The handling of drugs for use with carnivores requires training and experience. Pay particular attention to human safety to prevent personnel from being exposed inadvertently to veterinary drugs through contact with skin or mucous membranes (eyes and mouth; Kreeger and Arnemo 2007). Persons involved in the capture of carnivores should complete a creditable course in wildlife chemical immobilization, and have current training in basic first aid and cardiopulmonary resuscitation, prior to working with drugs. Personnel who use drugs and associated equipment for the capture of carnivores must have a clearly written emergency action plan in case of human exposure to drugs or capture-related injuries to personnel (Nielsen 1999; Cattet *et al.* 2005). Most physicians are unfamiliar with drugs used in wildlife capture, so communication beforehand will save valuable time in an emergency.

5.2.2 Selection of drugs for use in carnivores

More than one drug may be effective in a given species and availability of drugs changes over time as new products are released and old products are discontinued. In addition, use of drugs in wild animals is often "extra-label" or "off-label" (i.e. use of a specific drug does not follow conditions specified on the label, including specified species, dose, and method of administration). Conditions for extra-label use may be obtained from a wildlife or zoo veterinarian or found in peer-reviewed scientific literature. Regardless of source, the efficacy and safety of a drug based on empirical evidence in a target species should be the primary consideration in selecting a drug protocol. Many published reports describe the effectiveness of a specific drug for capture of a given species, but fewer reports evaluate safety based on the physiological responses of a species to a drug (e.g. vital rates, blood gases, adverse effects). Safety should not be ignored.

Drug effectiveness and safety must be considered when selecting an injectable drug for chemical immobilization (Table 5.2). Because no single drug meets all considerations, different drugs are often combined to attain many desired characteristics, while at the same time eliminating undesired effects (Grimm and Lamont 2007; Kreeger and Arnemo 2007). Many of these combinations include anesthetic drugs (e.g. ketamine, tiletamine), which cause loss of consciousness, with sedatives or tranquilizers (e.g. xylazine, medetomidine, zolazepam, acepromazine), which improve anesthesia in various ways including increased muscle relaxation and pain control. Some immobilizing drugs are used in conjunction with an antagonist drug that is administered to counteract the effects of anesthesia at the conclusion of handling or if complications arise during immobilization. Immobilizing drugs with antagonists are generally preferred because removing effects of anesthesia (1) permits mitigation of anesthesia-related physiological complications, (2) reduces likelihood of injury or death during recovery, and (3) decreases time spent by personnel monitoring recovery (Kreeger and Arnemo 2007).

Adjunctive drugs are used to support immobilization and are generally administered following capture and immobilization. These drugs include the dissociative anesthetic ketamine, which is often administered as a "top-up" to maintain immobilization, even when it is not a component of the immobilizing drug. The

Table 5.2 *Characteristics of ideal drugs for anaesthesia and euthanasia.*

Anaesthesia	
Advantages for the animals	Advantages for the users
Mixes safely with other drugs (i.e. no loss of potency or formation of by-products), and does not react with dart material.	Has low toxicity in humans should accidental exposure occur.
Is safe for pregnant and lactating animals, and nonirritating following intramuscular or intravenous injection.	Rapidly degrades *in vivo* to inactive, non-toxic metabolites (i.e. no harmful effects to humans consuming meat from drugged animals).
Is effective in small volumes (i.e. high potency), and has a wide margin of safety between effective and toxic doses (i.e. accidental overdosage is unlikely to have harmful effects).	Has low potential for human abuse as a recreational drug.
Causes rapid immobilization and loss of consciousness with minimal fear or memory of capture.	Is readily available on the market (i.e. commercial supplier exists, and access is not limited by regulation).
Causes minimal depression of cardiovascular and respiratory function, and produces muscle relaxation.	Is reasonably priced.
Causes minimal inhibition of swallowing reflex.	Is highly water soluble and stable in solution.
Causes good control of pain (analgesia) at immobilizing dosages.	Has a long shelf life.
Is reversible by administering an antagonist drug.	
Causes behavioral effects during induction, immobilization, and recovery that are predictable and safe.	
Rapidly degrades *in vivo* to inactive, non-toxic metabolites (i.e. no harmful effects to drugged animals, predators or scavengers).	
Euthanasia	
Causes rapid loss of consciousness and death without causing pain, distress, or anxiety.	Reliability.
Effects cannot be reversed.	Has low toxicity in humans should accidental exposure occur.
Widely compatible with different species, age, health status, and numbers of animals.	Safe to use in different environments, e.g. urban setting vs. remote field location.
Safe for predators or scavengers that consume the drugged carcass.	Compatibility with subsequent evaluation, examination, or use of tissue.
	Is readily available on the market.

advantage of ketamine for this purpose is that it is metabolized quickly and is less likely to prolong recovery, than will administering more of the initial immobilizing drugs (Cattet *et al.* 2005). Although generally not regarded as a drug, medical-grade oxygen is also a valuable adjunctive "drug" that can be used to prevent and treat several common complications (e.g. hypoxia, hyperthermia) associated with capture and anesthesia (Read *et al.* 2001; Arnemo and Caulkett 2007). Oxygen can be administered intranasally in the field without much difficulty and with minimal training using a lightweight aluminum cylinder (D- or E-type), a pressure regulator, and silastic tubing.

Drugs to relieve pain and stress should be considered for use with carnivores captured either with or without chemical immobilization (CCAC 2003). Aside from obvious concern for the welfare of captured animals, pain and stress can affect their behavior in ways that affect research results (Powell and Proulx 2003; Cattet *et al.* 2008a). Many drugs are available to provide pain relief (analgesia) and to reduce stress for wildlife. These include local anesthetic drugs, opioids, and non-steroidal anti-inflammatory drugs for pain relief (Machin 2007), and sedatives or tranquilizers for reducing stress (Arnemo and Caulkett 2007). Long-acting tranquilizers can be valuable for reducing stress in wildlife that must be translocated or maintained in captivity (Read 2002; Flick *et al.* 2007).

Table 5.3 lists some of the commonly used immobilizing drugs for different carnivore families. Detailed information on specific protocols, including dosages, can be found in extensive reference lists compiled by Kreeger and Arnemo (2007), and West *et al.* (2007).

5.2.3 Methods to administer drugs

Drugs can be delivered to wild carnivores via a variety of methods and equipment (Appendix 5.4) and no one method is suitable for all animals at all times. The choice of delivery method should be based on the behavior of the target species, the circumstances for drug administration, and the user's experience. The goal is to administer drugs in a safe (for personnel and animal alike) and effective manner (Cattet *et al.* 2005). Researchers seeking detailed information on use of equipment and on equipment manufacturers should review books by Nielsen (1999), Kreeger and Arnemo (2007), West *et al.* (2007), and Fowler (2008).

5.2.4 The value of knowledge and experience

Beyond knowledge of drugs and methods of administration, capture personnel must have knowledge of, and experience with, the target species to ensure that captures are effective, consistent, and safe (Nielsen 1999; Fowler 2008). For chemical immobilization, one must be able to visualize where thick, superficial

Table 5.3 *Immobilizing drugs for use with carnivores. "H" denotes relative use of drug is high compared to other drugs used for animals in the Family indicated; "M" use is moderate; "L" use is low.*

Family	*Immobilizing drugs with antagonists*[1, 2] *(Drug/antagonist)*	*Immobilizing drugs lacking antagonists*[1, 3]
Ailuridae (2)[4] (giant panda, lesser panda)	H: Ketamine-xylazine/yohimbine M: Ketamine-medetomidine/ atipamezole	H: Tiletamine-zolazepam
Canidae (28) (dogs, foxes, jackals, wolves)	M: Ketamine-xylazine/yohimbine M: Ketamine-medetomidine/ atipamezole L: Butorphanol-medetomidine/ naltrexone and atipamezole L: etorphine-promazine/ diprenorphine L: fentanyl-xylazine/naltrexone and yohimbine	H: Tiletamine-zolazepam H: ketamine-acepromazine L: Ketamine-promazine L: ketamine-midazolam
Eupleridae (2) (Malagasy civet, Malagasy ring-tailed mongoose)		H: Tiletamine-zolazepam
Felidae (27) (cats, lions, leopards)	H: Ketamine-xylazine/yohimbine M: Ketamine-medetomidine/ atipamezole L: Ketamine-medetomidine- butorphanol/atipamezole and naloxone L: tiletamine-zolazepam- medetomidine/atipamezole L: tiletamine-zolazepam-xylazine/ yohimbine or atipamezole	H: Tiletamine-zolazepam L: Ketamine L: ketamine-acepromazine
Herpestidae (3) (mongooses)	L: Ketamine-xylazine/yohimbine	H: Tiletamine-zolazepam M: Ketamine L: Ketamine-acepromazine
Hyaenidae (4) (aardwolf, hyenas)	M: Ketamine-xylazine/yohimbine L: Etorphine-xylazine/diprenorphine and yohimbine	H: Tiletamine-zolazepam L: Ketamine-acepromazine
Mephitidae (4) (skunks)	M: Ketamine-xylazine/yohimbine	H: Tiletamine-zolazepam H: ketamine-acepromazine
Mustelidae (22) (badgers, ferrets, otters, weasels)	H: Ketamine-medetomidine/ atipamezole H: ketamine-xylazine/yohimbine L: Ketamine-medetomidine- butorphanol/atipamezole and naloxone	H: Tiletamine-zolazepam M: Ketamine M: ketamine-acepromazine L: Ketamine-diazepam L: ketamine-midazolam

Family	*Immobilizing drugs with antagonists[1, 2] (Drug/antagonist)*	*Immobilizing drugs lacking antagonists[1, 3]*
	L: tiletamine-zolazepam-xylazine/ yohimbine or atipamezole L: etorphine-xylazine/diprenorphine and yohimbine L: fentanyl-xylazine/naltrexone and yohimbine L: fentanyl-diazepam/naltrexone	
Procyonidae (5) (coatimundi, raccoon, kinkajou)	M: Ketamine-xylazine/yohimbine M: ketamine-medetomidine/ atipamezole L: Tiletamine-zolazepam-xylazine/ yohimbine or atipamezole	H: Tiletamine-zolazepam H: ketamine-acepromazine L: Ketamine-acepromazine
Ursidae (7) (bears)	H: Tiletamine-zolazepam-medetomidine/atipamezole M: Tiletamine-zolazepam-xylazine/ yohimbine or atipamezole M: ketamine-xylazine/yohimbine M: ketamine-medetomidine/ atipamezole L: Etorphine/diprenorphine	H: Tiletamine-zolazepam
Viverridae (11) (civets, genets, binturongs)	M: Ketamine-xylazine/yohimbine L: Ketamine-medetomidine-butorphanol/atipamezole and naloxone	H: Tiletamine-zolazepam L: Ketamine-acepromazine

[1] *Kreeger and Arnemo (2007) and West* et al. *(2007) provide information on species-specific drug use including dosages, cautionary comments, and appropriate references.*
[2] *Immobilizing drugs are typically combined prior to injection, hence "ketamine-xylazine." Antagonist drugs are generally administered separately, hence "atipamezole and naloxone."*
[3] *Although antagonists are available for diazepam, midazolam, and zolazepam, they are seldom used because of their short duration of effect.*
[4] *In parentheses is number of species for which drug use is reported in scientific literature.*

muscles lie beneath skin and fur, because drugs are typically administered to wild carnivores by injection, often using remote drug-delivery equipment (blowpipes, modified pistols or rifles, and darts; Kreeger and Arnemo 2007). Injection into other tissues, such as fat or bone, will likely prolong or prevent capture and increase the potential for complications. One must be able to distinguish between normal and drug-induced behavior to monitor the effectiveness of a given drug dose and, if necessary, to determine if more drug is required. Once an animal is anesthetized, it is necessary to monitor its vital signs, as well as its level (or depth) of anesthesia, and to recognize when adverse physiological responses are developing (Cattet *et al.* 2005; West *et al.* 2007). Much of this knowledge can only be gleaned through extensive hands-on experience, not through the pages of books and reports. Nonetheless, attention to current literature and participation in creditable courses in wildlife chemical immobilization will help improve the value of field experiences.

5.3 Identification, prevention, and treatment of medical emergencies associated with capture

Wildlife capture is often unpredictable and relatively uncontrolled. As a result, the potential for medical emergencies is ever present, whether it is injury sustained during capture or adverse physiological response to drugs or restraint (Appendix 5.5). Emergencies or complications can develop at any time between capture and release, and sometimes days to weeks following release (Cattet *et al.* 2005). During capture, an animal can be injured by the trap (Powell 2005; Cattet *et al.* 2008a), through impact or injection by darts (Valkenburg *et al.* 1999; Cattet *et al.* 2006), or while being pursued (Cattet *et al.* 2003). While restrained in a trap, animals can injure themselves while attempting to escape (Proulx *et al.* 1993a; Powell 2005), be injured by other animals (Hooven *et al.* 1979; Craft 2008a), or develop adverse physiological conditions as a consequence of stress, extreme ambient temperatures, or lack of water (Cattet *et al.* 2003). With chemical immobilization, emergencies can arise with inappropriate use of drugs or failure to monitor physiological function (vital signs) of anesthetized animals (Cattet *et al.* 2005). Following release, animals may develop complications as a delayed effect of their response to capture (e.g. exertional myopathy, Cattet *et al.* 2008b).

5.3.1 Homeostasis, stress, distress, and treatment of medical emergencies

Preventing medical emergencies is better and easier than treating them. Effective prevention, however, depends on a sound knowledge of factors that can cause complications and how animals respond to them (Appendix 5.5). Normally, animals actively maintain a relatively constant internal environment (i.e. body temperature, acid–base balance, body water content, etc.) in the face of changing external conditions, such as weather, food availability, and activity. This homeostasis is an essential requisite for many biological processes, including reproduction, growth and development, and immunity. Factors that threaten or disturb homeostasis are called stressors, and the behavioral and physiological responses required to maintain homeostasis are collectively termed the stress response (Figure 5.2) (Hofer and East 1998; Moberg and Mench 2000). If the stress response is effective, homeostasis is maintained and biological processes continue unabated.

When biological processes are disrupted, however, as a result of a prolonged or excessive stress response, the resulting state is termed distress. Manifestations of distress include impaired reproduction, suppression of immune function, stunted growth, and reduced ability to mount an effective stress response in future. At its extreme, distress results in death. Reducing the occurrence and intensity of potential stressors in capture and handling will help prevent distress. Treating medical emergencies in free-ranging wild animals is often difficult, and sometimes impossible. Wild animals are not compliant patients, thus drugs are required to ensure that an animal remains immobilized, or at least sedated, during treatment. Further, effective treatment may require follow-up care over a period following

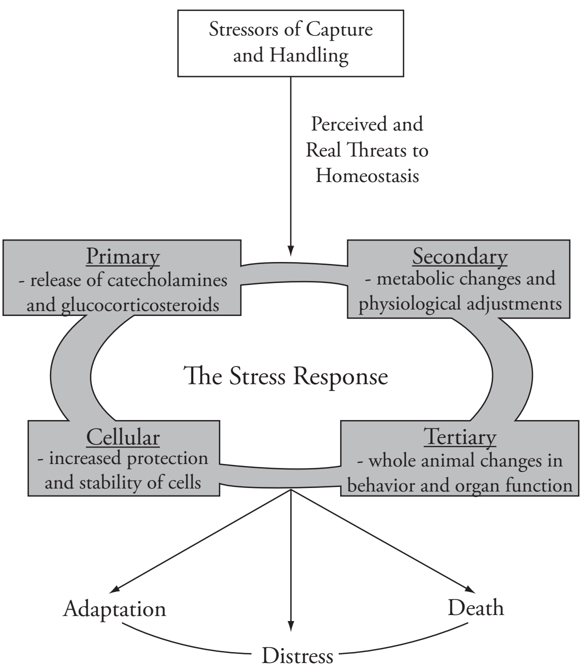


Fig. 5.2 Diagram illustrating the stress response that follows when an animal perceives a threat to homeostasis. The perception may be psychological, physical, physiological, or a combination of types. The overall effectiveness of the stress response—adaptation, distress, or death—is affected by the number, intensity, and duration of stressors.

initial treatment (e.g. to change bandages, to remove sutures or to administer medication). With non-captive, wild animals, follow-up care is not an option. Typically, an animal is released, possibly never to be seen again, and one hopes for the best. These difficulties underscore the importance of placing emphasis on prevention rather than treatment (Appendix 5.5). One must be able to recognize emergencies, to have proper treatment materials on hand, and to have appropriate training and skills required to provide treatment (Appendix 5.5). Researchers seeking more detailed information on treatment of medical emergencies should review books or technical manuals by Nielsen (1999), Cattet *et al.* (2005), Kreeger and Arnemo (2007), and Fowler (2008).

5.3.2 Necropsy

Animals that die during or following capture should be necropsied (Chapter 13; Cattet *et al.* 2005). If an animal dies as a direct result of capture procedures, the capture and handling protocol should be reviewed carefully and minutely, and possibly revised, to ensure similar deaths do not occur in future. If an animal dies as a consequence of concurrent disease combined with the stress of capture, necropsy findings will help to assure continued confidence in the capture protocol and may provide new information regarding the health of the species.

In the field, appropriate tissue samples should be collected and frozen or fixed in 10% buffered formalin for submission to a veterinary pathology facility (Chapters 4, 6, and 13). Appropriate tissue samples should include brain, lung, heart, liver, kidney, spleen, lymph nodes, and muscle. Capture personnel should refer to a wildlife necropsy manual for details regarding required equipment, techniques, and sampling procedures (Chapter 13; Munson 1999; CCWHC 2010). Documentation should include a detailed history and digital images of the field necropsy to assist the veterinary pathologist diagnosing the cause of death. Alternatively, under some circumstances, it may be desirable to arrange shipment of the entire carcass to a veterinary pathology facility for detailed necropsy (Chapter 13).

5.4 Euthanasia

Euthanasia is the humane killing of animals, characterized by minimal pain and distress (AVMA 2007). Minimal pain means the dying animal experiences little sensation of pain because its cerebral cortex, the area of the brain that controls thought, memory, sensation, and voluntary movement, has been rendered non-functional by drugs, concussion, or oxygen deficiency (hypoxia). Minimal distress means the dying animal has not had the opportunity to respond to its situation in a way that is harmful to itself. In the context of wildlife capture, untreatable pain and distress may provide the basis for deciding to kill an animal, so euthanasia by strict definition may not be possible (Drew 2006). Nonetheless, the killing should be as humane as possible.

In addition to severe untreatable injury, other situations can arise during capture and handling when researchers must consider euthanizing an animal; for example, when an animal poses an immediate threat to capture personnel, to the public, to other wildlife (e.g. risk of spreading a serious infectious disease) or to the environment (e.g. an invading species). Consequently, capture personnel must be familiar with acceptable methods of humanely killing wild animals, and must have appropriate equipment (including drugs) on hand to perform a kill quickly. Killing an animal humanely requires appropriate training and experience with the required techniques, restraint of the animal to be killed, and selection of proper drugs (Table 5.2) using criteria that consider humaneness and user safety (AVMA 2007). Specific attention must be given to how best to restrain a wild carnivore prior to killing it (AAZV 2006). Use of "gentle restraint" methods advocated for domestic animals are likely to be ineffective and dangerous with wild animals that are injured or already distressed by being captured. Sedative- or anesthetic-type drugs should be used for these situations, and drugs should be administered by a method that is safe for personnel and minimizes distress in the animal.

Specific consideration should also be given to the human psychological response to killing an animal (AVMA 2007). The decision to kill an animal is sometimes difficult. Uncertainty or differences in opinion often arise regarding the potential impact of a severe injury on an animal's future welfare. Furthermore, a euthanasia decision determined by peripheral factors (e.g. policies or regulations), rather than the condition of the animal, may be controversial. Decision criteria and methods for euthanasia should be discussed and understood by all team members prior to starting trapping.

Appendix 5.6 provides information on acceptable methods of euthanasia for wild carnivores. Some methods, such as exsanguination (bleeding out) or intravenous administration of potassium chloride, are only regarded as acceptable if the animal is killed while deeply anesthetized. Methods regarded as unacceptable, regardless of circumstances, include a blow to the head for animals >1 kg body weight, carbon monoxide, chest compression, drowning, hypothermia (or rapid freezing), and use of neuromuscular blocking agents, such as succinylcholine (AAZV 2006, AVMA 2007).

Table 5.4 *Restraining techniques to handle captured animals.*

Technique	Description	Examples
By hand with a catching pole (pole snare) or a forked stick	Wear gloves—grasp the animal firmly at the base of the neck with one hand, and the hips with the other hand. Hands and arms should be kept above the back of the mammal to avoid claws (Jones *et al.* 1996).	Wolves—Kolenosky and Johnston 1967 Jackals—Rowe-Rowe and Green 1981 Red foxes—Henry 2004; Craft 2008b Pumas—Davis *et al.* 1996 Badgers—Proulx, unpubl. data
Portable cushion	Use cushion to break the fall of anaesthetized animals.	Pumas—McCrown *et al.* 1990
Squeeze cage	This is a cage equipped with a squeeze panel (wire mesh, wood, netting, compact cloth) to hold an animal firmly against the side of the cage for anaesthesia.	Fishers—Buck 1982; Frost and Krohn 1994 River otters—McCullough *et al.* 1986
Wire mesh cone	This is used to handle animals <1500 g (Taber and Cowan 1969; Powell, unpubl. data; Proulx, unpubl. data).	American marten—Bull *et al.* (1996) Long-tailed weasel—Proulx, unpubl.data
Cloth, mesh or heavy plastic bags	May be used during anaesthesia.	Red fox—Zabel and Taggart 1989 Polecats—Forman and Williamson 2005

Carcasses of animals killed while anesthetized, or killed by barbiturate overdose, should be disposed of by deep burial or incineration to prevent secondary toxicity of scavengers (AAZV 2006).

5.5 Restraining and marking techniques

In the absence of, before or immediately after, anesthesia, captured animals must be restrained safely, so as to minimize physical injury and stress (Table 5.4).

Temporary or permanent marks should be as painless as possible and should not affect the animals' behavior or health (ASM Animal Care and Use Committee 1998). Marks must be matched to research objectives and must be appropriate for a carnivore's sizes, body shape, future growth, and behavior (Powell and Proulx 2003). Many short-term, long-term, and permanent markers have been developed for mammals, but few have been tested with carnivores (Appendix 5.7).

5.6 Designing effective trapping programs for carnivores

Carnivores have cognitive maps of where they live, and they do not use space within their home ranges randomly (Chapter 9; Peters 1978; Powell 2000; Proulx 2005). Therefore, setting traps randomly or uniformly across the landscape will likely be less productive than setting traps at special habitat features. Aside from trap and set characteristics, a variety of biotic and abiotic factors affect capture success. To develop an effective trapping program, one must know how a carnivore is associated with the vegetative and physical structures of a study area, and the sizes of home ranges of males and females, adults and juveniles, females with or without young, and dominant and subordinate animals. Traps may be spread so that each individual of a population has one trap within its home range (to trap as many different individuals as possible) or so that each individual has many traps within its home range (to recapture each individual many times; Powell and Proulx 2003). Home range and population sizes may be related to food patches (Macdonald 1983; Fuller *et al.* 1992), but also to intra- and interspecific competition (Rosenzweig 1966; Marker and Dickman 2005; Moorcroft *et al.* 2006). Finally, weather change can affect carnivore activity (Zielinski 2000). Understanding species-specific factors that may affect capture success dictates how and when traps should be set in the field to meet a program objective. The distribution of traps will vary from one species to another but, in all cases, trapping programs should be developed with spatio-temporal schemes that are compatible with the biology of animals.

5.7 Animal welfare

While animals have been captured for centuries by human populations evolving with their environments, today, capturing and handling carnivores is specialized work and must be conducted with scientifically sound protocols and high standards of animal use and welfare.

Researchers should apply Russell and Burch's (1959) "3 Rs," *Replacement, Reduction, Refinement*, to the use of animals in field research. Although the 3 Rs are well-established principles in the field of laboratory animal science, many wildlife researchers are unfamiliar with them and their implementation in wildlife research. This unfamiliarity may be explained, in part, because the goals of wildlife research often value the welfare or needs of populations, communities or ecosystems over the welfare of individual animals (CCAC 2008). Nonetheless, the welfare of individual wild animals is of concern because:

1. Animals (target or non-target species) may be injured during capture or handling.
2. Sampling or marking of captured animals may involve invasive procedures.
3. Wild animals are likely to be intensely stressed during capture because they are not conditioned to human handling.
4. Wild animals may conceal capture-related injuries (from researchers) that could have serious consequences for their long-term survival.
5. Welfare indicators are deficient for many wild species.
6. Peer-reviewed reports on the welfare and research implications of wild animal studies are lacking.

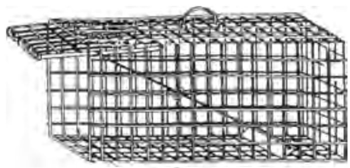
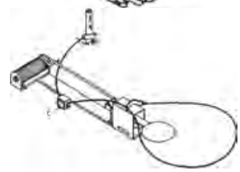
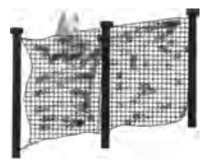
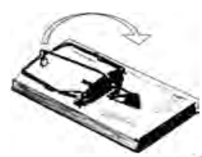
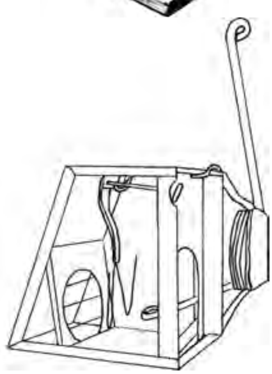
Researchers must consider and implement the 3 Rs to balance the needs of wildlife research and wild animals in accordance with the following definitions (CCAC 2008):

- *Replacement*—Researchers should use animals only if they are unable to find a replacement by which to obtain the required information. Replacement strategies include noninvasive sampling (Chapter 4), collation and use of information already gained, population meta-analyses, population and habitat suitability simulations, and archived tissue samples.
- *Reduction*—Researchers should use the fewest animals needed to provide valid information and statistical inference (Chapter 8). Sample size can be minimized by (1) designing research that yields data appropriate for statistical tests needing small or remotely collected samples (Chapters 4 and 8); (2) using factorial design to explore the effects of several variables in one experiment; (3) using sequential and multivariate statistical methods; or (4) using repeated measured designs (McConway 1992). Reduction also can be applied without compromising animal welfare by maximizing the information obtained per animal (e.g. collection of biological and genetic samples for archiving, Chapter 6), thereby limiting or avoiding, the subsequent use of additional animals. When trapping carnivores, reduction can be applied by designing trapping programs that minimize the likelihood of capturing non-target animals.
- *Refinement*—Researchers should use the most humane, least invasive techniques to minimize pain and distress (Chapter 4). This is the easiest of the 3 Rs to apply in wildlife research. Possible strategies include: (1) assessing and reducing potential sources of harm to captured animals; (2) avoiding methods that raised questions of animal welfare in other studies; (3) using drugs (analgesics) to control pain in invasive procedures (e.g. biopsy, tooth extraction); (4) using noninvasive sampling (Chapter 4) and other sampling not requiring capture to collect biological and genetic samples (e.g. skin samples

by remote biopsy darting; Spong and Creel 2001); (5) minimizing disturbances that can lead animals to abandon home ranges, can pre-empt feeding, can disrupt social structure, and can alter predator–prey relationships; (6) using a minimal (but safe) restraint and the shortest possible handling time; (7) collaborating with manufacturers to produce research equipment least likely to cause pain and distress or to disrupt an animal's normal way of life; and (8) publishing descriptions of refined techniques in the peer-reviewed scientific literature (CCAC 2008).

Researchers and managers can implement and promote the 3 Rs by ensuring that all personnel involved in their capture programs are trained appropriately in field procedures and have undertaken formal training in the concept and implementation of the 3 Rs, and by collaborating in the development and dissemination of training courses, guidelines, and protocols for various species and types of wildlife research (CCAC 2008; Norecopa 2008).

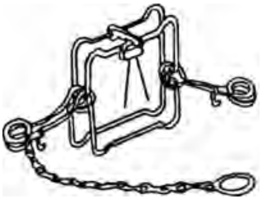
Appendix 5.1 *Trap types used in the capture of carnivores.*

Traps	*Material*	
Restraining devices Cage or box traps	Wire mesh, solid wood, metal, or plastic walls	
Foothold (leghold) traps	Metal clamping jaws that can be rubber-padded or offset	
Foot (leg), and neck snares	Metal cable of a single or multiple strands	
Nets	Nylon mesh	
Killing traps Snap trap (mousetrap)	Metal striking bars mounted on flat surfaces	
Planar trap	Metal bar	

Mode of action	*Species*	*References*
One or two entrances that close when animals step on a treadle or move a triggering device.	Small cages to capture small carnivores to huge structures made of logs or road culverts to trap wolverine- (*Gulo gulo*) to bear-sized (*Ursus* spp.) carnivores.	Powell and Proulx 2003
Jaws open to 180° in their set positions and clamp together to capture animals by a paw or a leg. Some traps have a housing that completely encases a captured limb. All traps are powered by either coil or leaf springs when sprung.	Foothold traps for canids and felids. Traps such as the EGG tap are used for small- and medium-sized carnivores that manipulate and explore with their paws (e.g. raccoon, *Procyon lotor*).	Proulx *et al.* 1993a Proulx 1999a Hubert *et al.* 1999
The energy to tighten the noose around an animal's limb or body is provided by the captured animal or a spring. The cable is equipped with a locking mechanism to prevent the loop from loosening.	Medium-sized carnivores. Neck snares are used to live-trap canids. They hold animals by their necks as if restrained with a leash; a stop prevents the loop from choking animals.	Nellis 1968 Bjorge and Gunson 1989 Proulx 1999a Woodroffe *et al.* 2005b Gese 2006
Drive nets, stretched loosely between two solid objects and supported by poles or branches, to capture carnivores driven by battue and fladry lines, or by helicopters. Hand-held net guns fired from helicopters or all-terrain vehicles.	Medium- and large-sized carnivores.	Beasom *et al.* 1980 Gese *et al.* 1987 Okarma and Jedrzejewski 1997
U-shaped jaw, as in common mouse and rat traps, or a straight bar that closes from 180° onto a flat surface.	Small carnivores.	Powell and Proulx 2003
The spring forms the killing bar and closes in the same plan.	Small carnivores.	Proulx 1999a

(continued)

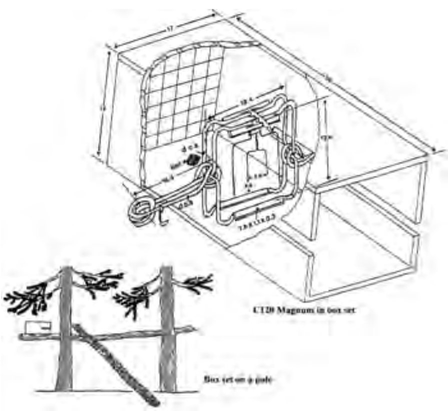
Appendix 5.1 *Continued*

Traps	*Material*	
Rotating-jaw (body-gripping) traps	Square-shaped metal bars	
Killing box trap	Metal striking jaw or cable	
Killing snares	Wire nooses set on land or underwater	
Submarine traps	Cage, rotating-jaw or foothold traps	

Mode of action	*Species*	*References*
Rotating-jaw traps have two metallic, circular, square, or rectangular frames that are hinged at their center point to operate in a scissor-like action, and are equipped with two torsion springs. Frames rotate and close on the animals upon firing.	Small- and medium- sized carnivores.	Proulx 1999a
Striking jaw or cable set within a box or pipe that is driven by a spring to strike an animal ventrally when the trigger is released.	Although these traps are used mainly for the capture of rodents, they can capture small carnivores such as weasels (*Mustela* spp.).	Proulx 1997, 1999a, unpubl. data.
In manual snares, an animal provides the energy to tighten the noose around its neck. In power snares one or more springs tighten the noose.	Medium-sized carnivores.	Proulx 1999a
Traps are set underwater, or slide underwater. The captured animal may drown or be killed by the trap itself.	Semi-aquatic (e.g. mink, *Neovison vison*) and riparian (e.g. raccoon) carnivores.	Proulx 1999a

Appendix 5.2 *Trap sets that are commonly used for the capture of carnivores.*

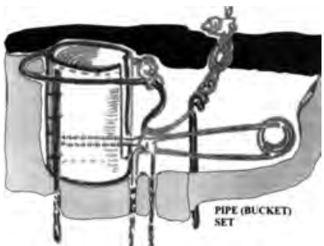
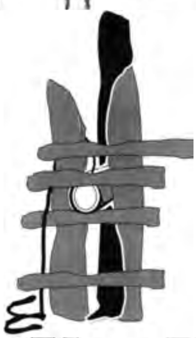
Set type	*Description*
Slide wire (drowning)	Foothold trap set in such a way on land, at the edge of water, or in a shallow rill entering a large body of water that it slides into the water upon capture of an animal. A lock stops the trap from coming back up, and the animal is submerged with the trap and drowns.
Channel	A rotating-jaw trap set at the bottom of water channel to capture predators such as minks and otters.
Running pole	A killing trap is set on a pole leaning on a tree trunk. Vegetation placed on top of the trap discourages animals from stepping over the trap to reach the bait, which is between the trap and the trunk. Bait covered with vegetation is less obvious to birds.
Box	A killing trap is inserted and secured in a wire, wooden, or plastic box with one end open and the other covered with wire mesh. Bait is placed behind the trap, at the back of the box near wire mesh. The box may be placed on the ground, on a stump, or on a running pole. Traps set in small boxes with openings at both ends will capture weasels.
Cubby	Teepee-like construction made of logs and branches, a hole dug into a bank, or a rock pile that encloses the trap and bait. The trap is set at the mouth of the funnel-like entrance, which channels the animal toward the bait. For bears (*Ursus* spp.), the back of the cubby should be a large rock or tree that forces the animal to enter the cubby to reach the bait. Large logs should be set on each side of a cubby to direct a bear towards the trap.

	References
	Boggess and Loegering 1985 Hubert *et al*. 1996
	Boggess and Loegering 1985
	Boggess and Loegering 1985
	Boggess and Loegering 1985 Proulx 1999a Proulx and Barrett 1993b
	Boggess and Loegering 1985 Proulx *et al*. 1995

(continued)

Appendix 5.2 *Continued*

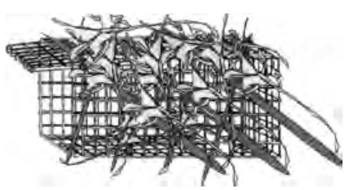
Set type	*Description*
Trail	A foothold trap or footsnare is set on a game trail. Setting the trap on one side of a log set across the trail forces a target animal to step over the log and land with its full weight on the trap trigger. A two-trap blind set, where a small stick is placed between the traps and at either approach, increases capture rates of some species (e.g. cougar, *Felis concolor*). For bears, a leg snare should be set under a footprint on the bear trail.
Pipe (bucket)	This is a set specifically for bears. The noose of an Aldrich snare is set around a 23-cm long stove pipe or bucket (13-cm diameter) inserted in a 23-cm deep hole in the ground. One side of the pipe has a 6.5-cm long and 2.5-cm wide slot to accommodate the spring throw arm of the snare so the trigger extends through the slot into the center of the pipe. Bait is placed at the bottom of the pipe, below the trigger. The cable loop and the spring throw arm are covered with soil, grass and leaves. When the snare fires, a bear's paw is below the rim of the noose and the snare captures the bear by the leg rather than by the paw.
Tube trap	A rubber-padded snare is placed within a PVC pipe that is 85 cm from the ground between three trees forming a triangle. When a bear pulls on the trigger to reach the bait placed at the back of the pipe, the snare tightens on the leg.
Snare	A manual or power snare is set across a game trail, without bait or scent, or set at the entrance of a baited enclosure (see pen set below). Depending on the size and height of the cable loop, medium- or large-sized carnivores may be captured selectively.
EGG trap	An EGG trap may be anchored to a tree above ground or set in a hole dug into a stream bank within 25 cm of the waterline.

	References
	Young and Goldman 1946 Provencher 1969
	PESCOF 1988 Hygnstrom 1994 Huber *et al*. 1996
	Lemieux and Czetwertynski 2006
	PESCOF 1988
	Proulx *et al*. 1993a Hubert *et al*. 1996

(continued)

Appendix 5.2 *Continued*

Set type	*Description*
Scent post	A scent or lure is placed on a stump, a stick or another prominent object to entice an animal to approach and rub the object. A foothold trap or foot snare set near the base of the stump captures the carnivore.
Dirt hole	A foothold trap or foot snare is set in front of a 10-cm diameter and 20-cm deep hole dug at a 45–60° angle at the base of clump of weeds, small stump or other backstop, in a relatively open area where visibility is good on all sides. Bait is placed at the bottom of the hole and covered with dirt.
Cage trap	A cage trap set uses the trap itself as a self-contained cubby for carnivores that will enter enclosed spaces. Traps should be concealed and covered with vegetation to protect animals from sunlight, precipitations and predators. Bait should be placed behind the treadle or trigger to force the animal to enter the trap and step on the trigger.
Pen	A pen set uses a pen with a single entrance constructed around a burrow system inhabited by a target carnivore. Bait is located outside the pen, in line with the entrance and cage, foothold trap, foot snare, or killing trap is set at the entrance, between the pen and the bait.

	References
	Boggess and Loegering 1985
	Boggess and Loegering 1985 PESCOF 1988 Krause 1989
	Boggess and Loegering 1985 Powell and Proulx 2003
	Currie and Robertson 1992Proulx, unpubl. data

Appendix 5.3 *Assessment of killing and restraining traps. For details on appropriate trap design and sets, consult the cited references. Criterion I: at a 95% confidence level, humane killing traps should render ≥70% of target animals irreversibly unconscious in ≤3 minutes. Criterion II: at a 95% confidence level, humane restraining traps should hold ≥70% of animals with ≤50 points scored for physical injury.*

Species	*Trap model*	*Performance*	*Meets Criterion I*		*Meets Criterion II*	
			Yes	*No*	*Yes*	*No*
CANIDS						
Coyote (*Canis latrans*)	Manual neck snare and neck snares with Kelly lock, and Gregerson or Denver Wildlife Research Center locks.[1]	Do not meet Criterion I (Guthery and Beasom 1978; Phillips 1996).		√		
	No.3—steel-jawed[2], laminated steel-jawed Sterling MJ600[3], Northwoods[2], and Bridger[4] traps	Do not meet Criterion II (Olsen *et al.* 1986; Phillips *et al.* 1996; Hubert *et al.* 1997).				√
	Novak foot snare[5]	Does not meet Criterion II (Onderka *et al.* 1990).				√
	Nos. 3 and 3½ EZ grip padded foothold traps[6]	Meet Criterion II (Olsen *et al.* 1988; Phillips *et al.* 1996). Can be equipped with tranquilizer tabs (Balser 1965; Berger and Gese 2007).			√	
	Fremont footsnare[7]	Meets Criterion II (Onderka *et al.* 1990). Effective snare cable lock required to minimize escapes (Skinner and Todd 1990).			√	
	0.32-cm diameter cable foot snare with twist-link chain between cam-lock and cable	Based on low frequency of major injuries in 17 animals (Darrow *et al.* 2009), likely meets Criterion II.			√	
	0.32-cm diameter standard cable foot snare with cam-lock, with or without a plastic tube sleeve	Based on mean injury scores and high frequency of major injuries (Darrow *et al.* 2009), does not meet Criterion II,				√
	Collarum neck snare[8]	Meets Criterion II (Shivik *et al.* 2005).			√	

	Cam-Loc[9] neck snare with tranquilizing tab	With 3.2-mm cable, lock stop set at 27 cm, 4-mg diazepam tab meets Criterion II (Pruss *et al.* 2002). Winter capture efficiency comparable or superior to standard neck snares or foothold trapping devices (Pruss *et al.* 2002).		√	
	Net gun[10]	With 8-cm mesh and 13-cm mesh, meets Criterion II (Barrett *et al.* 1982, Gese *et al.* 1987). Aerial net gunning common (e.g. Kitchen *et al.* 1999).		√	
	Cage traps[11] 66 × 51 × 152 cm, 66 × 51 × 183 cm	Used where padded foothold taps and snares illegal, but they are not capture-efficient. Expensive, frequent non-target species, many months of pre-baiting required; because of frequent tooth injuries, may not meet Criterion II (Way *et al.* 2002).			√
	Multicapture, wooden box trap 35 × 35 × 105 cm	Placed at den entrance, meets Criterion II for capture of pups (Foreyt and Rubenser 1980).		√	
Dogs, wild; *dingoes (Canis familiaris dingo)*, feral domestic *(C. familiaris)*, dog hybrids	No. 1½ Victor Soft-Catch™ foothold trap[2]	Meets Criterion II with or without tranquilizer trap device (Fleming *et al.* 1998; Marks *et al.* 2004). As capture efficient as other foothold traps used by trappers.		√	
	No. 3 Victor Soft-Catch™ foothold trap[2]	Meets Criterion II (Meek *et al.* 1995).		√	
	Treadle snare[12]	Meets Criterion II (Meek *et al.* 1995, Fleming *et al.* 1998). More expensive, bulky and awkward than Soft-Catch™ traps. Less capture efficient than commonly used traps. Wire snares must be replaced after each capture.		√	
	Cage trap of metal bars and wire fences, two guillotine doors 1.7 × 1.2 × 0.7 m	Meets Criterion II (Curi and Talamoni 2006). Highly selective for canids.		√	
Fox, arctic (*Vulpes lagopus*)	Sauvageau 2001–08[13]	With baited offset trigger, set on post in frozen ground, portable three-sided wire mesh (or snow) cubby, meets Criterion I (Proulx *et al.* 1993b, 1994).	√		

(continued)

Appendix 5.3 *Continued*

Species	*Trap model*	*Performance*	*Meets Criterion I*		*Meets Criterion II*	
			Yes	*No*	*Yes*	*No*
CANIDS						
	No. 1½ long spring steel-jawed foothold trap[2]	If checked daily, meets Criterion II (Proulx *et al.* 1994).			√	
Manual snares, 0.15 cm cable	If hung directly above dens to capture pups running through the den area, animals processed upon capture, meets Criterion II (Zabel and Taggart 1989).			√		
Fox, crab-eating (*Canis thous*)	Cage trap of metal bars and wire fences, two guillotine doors 1.7 × 1.2 × 0.7 m	Meets Criterion II (Curi and Talamoni 2006). Highly selective for canids.			√	
Fox, grey (*Urocyon cinereoargenteus*)	No. 1½ steel-jawed foothold trap[2]	Does not meet Criterion II (Berchielli and Tullar 1980; Olsen *et al.* 1988).				√
Fox, kit or swift (*Vulpes velox*)	Tomahawk wire cage traps[11] 39 × 39 × 109 cm double-door 31 × 31 × 83 cm single-door Havahart wire cage traps[14] 26 × 26 × 83 cm	If lined with a 3-mm hard board, meet Criterion II (Moehrenschlager *et al.* 2003). Two reverse, double-set traps more capture-efficient than single trap (Kamler *et al.* 2002).			√	
Single-gate, clover-type trap 46 × 46 × 122 cm	Trap is set at the entrance of an enclosure placed over den holes (Zoellick and Smith 1986). Meets Criterion II for the capture of pups.			√		
Fox, red (*Vulpes vulpes*)	Manual snares[15]	Does not meet Criterion I (FPCHT 1981).		√		
	Power snares[15]	Does not meet Criterion I (Proulx and Barrett 1990).		√		
	No. 1½ steel-jawed and No. 3 Victor Soft-Catch™ foothold traps[2]	Do not meet Criterion II (Olsen *et al.* 1988, Kern *et al.* 1994. Seddon *et al.* 1999).				√
	No. 1½ Soft-Catch foothold trap[2]	Meets Criterion II (Olsen *et al.* 1988; Kern *et al.* 1994; Fleming *et al.* 1998; Kreeger *et al.*			√	

		1990b). As capture efficient as equivalent, unpadded traps (Tullar 1984; Linscombe and Wright 1988).		
	Åberg (Swedish) foot snare[16]	Meets Criterion II (Englund 1982).	√	
	Novak foot snare[5]	With blackened cable, appears to meet Criterion II (Novak (1981).	√	
	Belisle foot snare[17]	Meets Criterion II (Muñoz-Igualada *et al.* 2002). Less capture efficient than Collarum foot snare. More selective than cage traps.	√	
	Collarum neck snare[8]	Meets Criterion II (Muñoz-Igualada *et al.* 2002). More capture efficient than Belisle foot snare. More selective than cage traps.	√	
Fox, Ruppell's (*Vulpes rueppellii*)	No. 3 Victor Soft-Catch™ foothold trap[2]	Meets Criterion II, possibly because animals lay relatively quietly in foothold traps (Seddon *et al.* 1999).	√	
Jackals (*Canis mesomelas, C. adustus, C. aureus*)	No. 3 Oneida Jump trap[1] padded with two layers of cotton mutton cloth	Meets Criterion II when traps checked daily after dawn (Rowe-Rowe and Green 1981).	√	
	Victor Soft-Catch™ No. 1½ foothold trap[2]	Meets Criterion II if animals handled upon capture (Craft 2008b).	√	
Wolf, grey or timber (*Canis lupus*)	No. 4 double long spring, No. 4 jaws offset 2 mm, No. 14 toothed, No. 14 double long spring steel-jawed foothold traps[2]	Commonly used to capture wolves in North America but cause extensive injuries and do not meet Criterion II (Van Ballenberghe 1984; Kuehn *et al.* 1986; Sahr and Knowlton 2000).		√
	Modified Newhouse No.4[18] foothold trap	With one spring only, meets Criterion II (Kolenosky and Johnston 1967).	√	
	Newhouse 14 with 1.8-cm offset toothed jaw[18] foothold trap	May meet Criterion II (Kuehn *et al.* 1986).		

(continued)

Appendix 5.3 *Continued*

Species	*Trap model*	*Performance*	*Meets Criterion I* Yes	*Meets Criterion I* No	*Meets Criterion II* Yes	*Meets Criterion II* No
CANIDS						
	No. 4 foothold trap with offset steel-jaws[1] with tranquilizer tabs				-	
		With propiopromazine hydrochloride tabs meet Criterion II (Sahr and Knowlton 2000, Chavez and Gese 2006). For use with adults and pups.			√	
	No. 7 EZ Grip Trap[6]	May meet Criterion II (Frame and Meier 2007).			-	
	Aldrich foot snare[19]	Meets Criterion II (Van Ballenberghe 1984; Okarma *et al.* 1998). Difficult to conceal, not capture efficient for trap-shy wolves.			√	
	Drive net	With nylon mesh nets stretched loosely and supported by poles or branches, meets Criterion II (Okarma and Jedrzejewski 1997; Okarma *et al.* 1998; Theuerkauf *et al.* 2003).			√	
	Net gun[10]	Meets Criterion II (Walton *et al.* 2001b; Chavez and Gese 2006). Used to capture adults and pups.			√	
Wolf, maned (*Canis brachyurus*)	Cage trap of metal bars and wire fences, two guillotine doors 1.7 × 1.2 × 0.7 m	Meets Criterion II (Curi and Talamoni 2006). Highly selective for canids.			√	
FELIDS						
Bobcat (*Lynx rufus*)	No. 3 Soft-Catch™ foothold trap[2]	Meets Criterion II (Olsen *et al.* 1988). Can be modified for optimal use (Earle *et al.* 2003).			√	
	Cage trap 38 × 38 × 90 cm	Meets Criterion II (Woolf and Nielsen 2002). More capture-efficient than No. 3 Soft-Catch foothold traps.			√	
Cat, feral domestic (*Felis catus*)	No. 1½ Soft-Catch™ foothold trap[1]	Meets Criterion II (Molsher 2001).			√	
	Cage trap[20] 40 × 40 × 60 cm	Meets Criterion II (Molsher 2001).			√	
Cougar (*Puma concolor*)	Schimetz-Aldrich foot snare[21]				√	

		Meets Criterion II (Logan *et al.* 1999). May cause fewer deaths than treeing animals with trained dogs.				
Lynx, North American (*Lynx canadensis*)	Conibear 330[1]	Used by trappers in North America (CGSB 1996) but does not meet Criterion I (Proulx *et al.* 1995).		√		
	Conibear 330[2] with clamping bars and a one-way four-prong trigger	In simulated natural environments met Criterion I; never field-tested (Proulx *et al.* 1995).	-			
	No. 3 Soft-Catch™ foothold trap[2]	Does not meet Criterion II even with weakened springs (Mowat *et al.* 1994).				√
	Fremont foothold snare[7]	Meets Criterion II (Mowat *et al.* 1994), especially if loosely attached to trees or drag poles and modified to reduce tangling.			√	
	Tomahawk wire-cage traps[11] 66 × 51 × 122 cm, 51 × 38 × 114 cm Home-made wire cage trap 99 × 102 × 122 cm	Meet Criterion II (Mowat *et al.* 1994; Kolbe *et al.* 2003). Large traps may have higher capture efficiency.			√	
Lion (*Panthera leo*)	Aldrich foothold snare[19]	Set in a baited thornbush enclosure, meets Criterion II (Frank *et al.* 2003). After brief struggle, lions lie down.			√	
Tiger, Amur (Siberian) (*Panthera tigris altaica*)	Aldrich foothold snare[19]	Meets Criterion II (Goodrich *et al.* 2001). Effort/capture high.			√	
HYAENIDS						
Hyena, brown (*Hyaena brunnea*)	Aldrich foot snare[19]	Meets Criterion II (Frank *et al.* 2003).			√	
MEPHITIDS						
Skunks, striped, spotted, hog-nosed (*Mephitis mephitis, M. macroura, Spilogale putorius, S.* gracilis, Conepatus mesoleucus)	Foothold traps	Used (Allen 1939; Verts 1960) but limb injuries range from cut skin to fractures to chewed feet; do not meet Criterion II (Novak 1981; Rosatte 1987).				√
	Foothold snares	Do not meet Criterion II (Novak 1981). Poor capture efficiency.				√

(continued)

Appendix 5.3 *Continued*

Species	*Trap model*	*Performance*	*Meets Criterion I* *Yes*	*No*	*Meets Criterion II* *Yes*	*No*
MEPHITIDS						
	Wire cage traps	If covered with wood, plastic or cartons meet Criterion II (Larivière and Messier 1999). Opaque-sided traps recommended to reduce stress, facilitate handling (Larivière and Messier 1999).			√	
Wooden box traps	Meet Criterion II (Crabb 1941; Knight 1983).			√		
MUSTELIDS						
Badgers (*Meles meles*, *Taxidea taxus*)	Tomahawk wire cage trap[11] 76 × 25 × 30 cm	Meet Criterion II (Woodroffe *et al.* 2005b). May be less efficient than foothold traps to capture Eurasian badgers (*Meles meles*) (Loureiro *et al.* 2007) but recommended to cull populations in Europe (MAFF 1983).			√	
	No. 1½ Soft-Catch™ foothold trap[2]	Meets Criterion II (Kinley and Newhouse 2008).			√	
	No. 3 Soft-Catch™ foothold trap[2]	Meets Criterion II (Schemnitz 2005).			√	
Fisher (*Martes pennanti*)	Conibear 220[2]	Commonly used by trappers but does not meet Criterion I (Proulx and Barrett 1993b).		√		
	Bionic[22]	With 10 cm aperture cone met Criterion I in simulated natural environments (Proulx and Barrett 1993a). Never field-tested (Proulx 1999a).	-			
	Coon-getter cage trap[23] 37.5 × 37.5 × 90 cm	Meets Criterion II. Relatively efficient, particularly when equipped with radio monitors (Arthur 1988; Frost and Krohn 1994). Serious tooth injuries minimized if no openings larger than 2.5 cm^2 and hard surfaces covered with wood (Arthur 1988).			√	

Marten, American (*Martes americana*)	No. 3 and 4[2] foothold traps in pole box sets	Strike martens in the chest but do not cause major traumatic lesions; do not meet Criterion I (Barrett *et al.* 1989). Energy levels too low (Proulx and Barrett 1994).		√	
	Conibear 120[2]	Commonly used by trappers but does not meet Criterion I (Proulx *et al.* 1989a)		√	
	C120 Magnum[22] with one-way, four-prong pitchfork trigger (central prongs shorter than outside ones)	If in a pole box set, animals single-struck in the head-neck region or double-struck in the head-neck and thorax regions; meets Criterion I (Barrett *et al.* 1989; Proulx *et al.* 1989b). Capture efficiency same as standard Conibear traps.	√		
	Bionic[24]	Judged suitable from work with minks and fishers (Proulx and Barrett 1991a, 1993a). In pole sets, trauma associated with quick loss of consciousness; capture efficiency similar to control traps used by trappers (Proulx 1999b).	√		
	Tomahawk wire cage traps[11] 15 × 15 × 71 cm 20 × 20 × 51 cm	If wrapped in black plastic or connected to wooden nest boxes, meet Criterion II (Bull *et al.* 1996). Relatively capture efficient.			√
Mink (*Neovison vison*)	No. 1½ foothold trap[2] in drowning sets	Does not meet Criterion I (Proulx's 1999a review of Gilbert and Gofton 1982). Note that drowning is not considered to be an acceptable euthanasia method (Ludders *et al.* 1999, 2001).		√	
	Conibear 120[2]	Commonly used by trappers in North America but even upgraded version lacks necessary power; does not meet Criterion I (Gilbert 1981).		√	
	C120 Magnum[22] with pan trigger	The only rotating-jaw trap of its category with the potential to meet Criterion I when it strikes in the head-neck and thorax regions simultaneously (Proulx *et al.* 1990). Captures efficiency similar to traps commonly used by trappers (Proulx and Barrett 1993c).	√		

(continued)

Appendix 5.3 *Continued*

Species	*Trap model*	*Performance*	*Meets Criterion I* *Yes*	*No*	*Meets Criterion II* *Yes*	*No*
MUSTELIDS						
Otters (*Hydrictis maculicollis, Lontra canadensis, Lutra lutra*)	Hancock[25] 45 × 59 × 95 cm	Used to capture North American otters (Northcott and Slade 1976; Melquist and Hornocker 1979) but tooth damage, particularly canines, can be excessive and otters may become trap shy (Blundell *et al.* 1999).				√
	No. 1½ Soft -Catch™ foothold trap[2]	Fewer limb injuries than for modified Victor No. 11 double spring trap but no difference in dental injuries (Serfass *et al.* 1996). More research and development needed to minimize serious trauma and to meet Criterion II.				√
	Havahart wire cage traps[14] 40 × 40 × 100 cm unknown 80 × 80 × 140 cm	Recommended by Maxfield *et al.* (2005) for North American otters, and by Perrin and Carranza (1999) for the spotted-necked otters but no data on capture efficiency or tooth injuries.			-	
	No. 11 Victor and Sleepy Creek foothold traps[2, 26]	Meet Criterion II (Blundell *et al.* 1999, Shirley *et al.* 1983).			√	
	No. 3 Victor Soft-Catch™ foothold trap[2] equipped with a trap alarm	Functioning alarms and rapid response (ca. ½ h) necessary to meet Criterion II (Ó Néill *et al.* 2007).			√	
	Nos. 1 and 1½ Victor Soft-Catch™ foothold traps[1] with one spring replaced by a # 2 spring	Set in shallow water, anchored to solid objects, 1-m long chain, monitored daily early morning. Meet Criterion II (Fernández-Morán *et al.* 2002).			√	
Polecat (*Mustela putorius*)	Wire cage traps[6] 15 × 15 × 76 cm	If covered with dry hay for animal welfare so polecats pull at hay instead of trap, meets Criterion II (Birks *et al.* 1994).			√	

Weasel, short-tailed, or stoat (*Mustela erminea*)	Victor No. 1½ steel-jawed foothold trap[2]	Does not meet Criterion I (Trap Effectiveness Research Team 1995).		√		
	Fenn trap[27]	Does not meet Criterion I (Warburton *et al.* 2008). Efficient, used to control stoats and weasels (King 1981).		√		
	DOC traps[28] Models 150, 200, 250	Meet Criterion I (Warburton *et al.* 2008). Conceptually identical to Fenn trap but have six parallel strike bars powered by two coil springs. Set in wooden tunnels, capture as efficiently as Fenn traps (New Zealand Department of Conservation 2008).	√			
Wolverine (*Gulo gulo*)	Foothold No. 4[2] Wooden and metal cage traps	Used to capture wolverines (Hornocker and Hash 1981, Banci 1987, and others) but do not meet Criterion II. Capture efficiency low (Lofroth *et al.* 2008).				√
	Log trap	If bait properly attached, meets Criterion II (Copeland *et al.* 1995; Lofroth *et al.* 2008). Capture efficiency of portable wooden traps and modified round log traps markedly greater than for foothold and metal cage traps (Hornocker and Hash 1981; Banci 1987).			√	
PROCYONIDS						
Raccoon (*Procyon lotor*)	Sauvageau 2001–8[13] Conibear 220[1]	Used by trappers in North America but lack power to meet Criterion I, especially the Conibear 220 (Proulx and Drescher 1994).		√		
	Conibear 160[1]	Used by trappers in North America but lacks power to meet Criterion I (Sabean and Mills 1994).		√		
	Foothold traps with steel or padded jaws[2]	Do not meet Criterion II (Olsen *et al.* 1988; Berchielli and Tullar 1980; Tullar 1984; Proulx *et al.* 1993a; Hubert *et al.* 1996).				√

(continued)

Appendix 5.3 *Continued*

Species	*Trap model*	*Performance*	*Meets Criterion I*		*Meets Criterion II*	
			Yes	*No*	*Yes*	*No*
PROCYONIDS						
	EGG trap[29]	If anchored to a tree above ground, meets Criterion II (Proulx 1991; Proulx *et al.* 1993a). As capture efficient as rotating-jaw traps, avoids non-target species (Proulx 1995). More capture efficient than cage traps (Austin *et al.* 2004). Used in holes in stream banks, more injurious but still more humane than foothold traps in similar sets (Hubert *et al.* 1996).			√	
URSIDS						
Bear, black (*Ursus americanus*), Grizzly bear (*U. arctos*), Malayan sun (*Helarctos malayanus*)	Culvert and barrel trap	If constructed correctly, meet Criterion II (Powell and Proulx 2003). Efficient but cumbersome (Powell and Proulx 2003). May cause less distress and muscle injuries than foot snares (Schroeder 1987; Wong *et al.* 2004; Powell 2005; Cattet *et al.* 2008a).			√	
	Aldrich snare[18]	If set properly and modified with shock-absorbing spring, meets Criterion II (Johnson and Pelton 1980; Kaczensky *et al.* 2002; Powell 2005).			√	
	RL04 tube trap[30] with rubber-padded snare	If set properly, meets Criterion II (Lemieux and Czetwertynski 2006). Use solid anchor for grizzly bears.			√	

VIVERIDS				
Civet. brown palm (*Paradoxurus jerdoni*)	Havahart wire-cage trap[14] 33 × 28 × 107 cm	For animals processed within 30 min after capture, meets Criterion II (Mudappa and Chellam (2001).		√

[1]*Kelley locks: D. Amberg, Morris, Minnesota, USA; Gregerson locks: K. Gregerson, Roundup, Montana, USA; Denver Wildlife Research Center, Denver, Colorado, USA.*
[2]*Woodstream Co., Lititz, Pennsylvania, USA. Current manufacturer: Oneida Victor, Inc., Euclid, Ohio, USA.*
[3]*Sterling MJ600: Glen Sterling, Faith, South Dakota, USA.*
[4]*Montgomery Fur Co., Ogden, Utah, USA.*
[5]*E. R. Steel Products, Barrie, Ontario, Canada.*
[6]*Livestock Protection Co., Alpine, Texas, USA.*
[7]*Fremont Humane Traps, Beaumont, Alberta, Canada.*
[8]*Wildlife Control Supplies, East Branby, Connecticut, USA.*
[9]*Halford Hide & Leather Co., Ltd, Edmonton, Alberta, Canada.*
[10]*Mountain Helicopters Ltd., Taupo, New Zealand.*
[11]*Tomahawk Live Trap Co., Tomahawk, Wisconsin, USA.*
[12]*Glenburn Motors, Yea, Victoria, Australia.*
[13]*Les Pièges du Québec, Enr., St-Hyacinthe, Quebec, Canada.*
[14]*Havahart Woodstream Co., Lititz, Pennsylvania, USA.*
[15]*King snare: Western Creative Services, Ltd., Winnipeg, Manitoba, Canada; Mosher snare: W. C. Mosher, Mayerthorpe, Alberta, Canada.*
[16]*Nordic Sports AB, Kanalgatan 73, S-931 00 Skellefteå, Sweden.*
[17]*Edouard Belisle, Sainte-Veronique, Quebec, Canada.*
[18]*Kirsh Foundry, Inc., Beaver Dam, Wisconsin, USA.*
[19]*Aldrich Animal Trap Co., Clallam Bay, Washington, USA.*
[20]*Manufacturer not identified.*
[21]*Schimetz-Aldrich Spring Activated Animal Care, Sekiu, Washington, USA. Now available from Margo Supplies, Ltd., High River, Alberta, Canada.*
[22]*Originally produced by Woodstream Co. Current manufacturer: Les Pièges du Québec, Enr., St-Hyacinthe, Quebec, Canada.*
[23]*Coon Getter Traps, Miller, South Dakota, USA.*
[24]*Originally produced by W. Gabry, Vavenby, British Columbia, Canada. Current manufacturer: Les Pièges du Québec, Enr., St-Hyacinthe, Quebec, Canada.*
[25]*Hancock Trap Co., Custer, South Dakota, USA.*
[26]*Sleepy Creek Manufacturing, Inc., Berkeley Springs, West Virginia, USA.*
[27]*A. Fenn & Co., Hoopers Lane, Astwood bank, Redditch, Worchestershire, UK.*
[28]*Department of Conservation, Wellington, New Zealand.*
[29]*EGG Trapp Co., Butte, North DAKOTA, USA.*
[30]*Rolland Lemieux, St.-Emile, Quebec, Canada.*

Appendix 5.4 *Methods for administering drugs to wild carnivores.*

Method	Distance from Target Animal	Description	Specific References[1]
Inhalation anesthesia (gas anesthesia)	Close contact	Vaporized drug (volatile anesthetic) delivered directly into the lungs of an animal for absorption by blood and delivery to the brain. Advantages over injectable drugs: finer level of operator control, rapid induction and recovery. Disadvantages: requires field-durable delivery system and dedicated operator to monitor delivery system and animal continuously. Although use in field research is limited, has proven effective and safe with marine and terrestrial carnivores (Heath *et al.* 1996; Mathews *et al.* 2002; Lewis 2004; Potvin *et al.* 2004).	Heath *et al.* 1996 Mathews *et al.* 2002 Lewis 2004 Potvin *et al.* 2004
Hand injection (syringe and needle)	Close contact	Used to deliver drug to restrained or anesthetized animals, to transfer drug to other delivery devices, and to collect blood samples. Syringes available in many sizes (1–60 ml).Needles available in many lengths (16–75 mm) and gauges (14–25 ga).One-time use of disposable syringes and needles strongly recommended to avoid contamination of drugs, blood samples, and animals. Safe practices essential for handling needles and other sharps to prevent "needlestick injury" (the most common cause of accidental human exposure to drugs and animal fluids; Weese and Jack 2008).	See general references for instructions on use of syringe and needle.
Nasal delivery	Close contact	Intranasal drug delivery (drug sprayed into the nostrils) may provide an alternative to intravenous drug delivery in select cases.	Cattet *et al.* 2004 Wolfe and Bernstone 2004

		Advantages: quick and painless, nasal route is immediately available (relative to venipuncture), time to drug effect as rapid as by intravenous delivery (Wolfe and Bernstone 2004). Although use in carnivores not reported, used with xylazine (a sedative-type drug) to reduce stress in wild elk (*Cervus elaphus*) captured by net gun (Cattet *et al.* 2004).	
Oral delivery	Remote	Delivery of drug into the mouth by "tranquilizer tablet" or by drug-laced food (bait). Tranquilizer tablets used with mixed success to calm carnivores captured in foothold traps and snares (Balser 1965; Sahr and Knowlton 2000; Pruss *et al.* 2002) Drug-laced bait used to immobilize captive black bears and brown bears (Ramsay *et al.* 1995; Mortenson and Bechert 2001). Difficulties for drug delivery to free-ranging animals include poor bait acceptance, drug instability, inability to control dosage, risk for non-target animals (Conover 2002). Programs for mass vaccination of wild carnivores with vaccine-laced bait successful (Bachmann *et al.* 1990; Ballesteros *et al.* 2007). Carcasses laced with midazolam used to sedate lions (*Panthera leo*; Stander and Morkel 1991).	Bachmann *et al.* 1990 Stander and Morkel 1991 Ramsay *et al.* 1995 Sahr and Knowlton 2000 Mortenson and Bechert 2001 Conover 2002 Pruss *et al.* 2002 Ballesteros *et al.* 2007
Pole syringe(jab pole)	≤3 m	Extends the distance of operation for injection by syringe and needle. Used to deliver drug to trapped or restrained animals or to give additional drug to animals not completely immobilized but safe to approach. Effective volumes generally limited to <10 ml because animals react quickly to injection. Requires quick delivery through short (≤2.5 cm), large gauge (14–18 ga) needle.	Beltran and Tewes 1995 Grassman *et al.* 2006 Lofroth *et al.* 2008

(continued)

Appendix 5.4 *Continued*

Method	Distance from Target Animal	Description	Specific References[1]
Darts	Determined by type of projector	Used for remote delivery of drug (1–20 ml) to a specific target animal using blowpipe, bow, or dart gun. Many different designs, but basic components are a needle, body (or syringe), plunger, and tailpiece. Discharge of drug by expanding gas from an explosive powder charge, by compressed air, by butane, or by chemical reaction. Some designs and discharge mechanisms more likely to cause injury to target animals (Cattet *et al.* 2006). Needles of different sizes and port configurations have smooth shaft, wire barb, or metal (or gelatin) collar. Barb or collar essential to retain dart securely in free-ranging animal. Can be equipped with radio transmitters to enable location of lost darts or animals (Kilpatrick *et al.* 1996). Can be fitted with skin cutting heads for remote biopsy sampling (Spong and Creel 2001).	Kilpatrick *et al.* 1996 Spong and Creel 2001 Kreeger 2002 Cattet *et al.* 2006
Blowpipe(blow gun)	≤15 m	Effective for restrained animals or free-ranging animals that can be approached closely. Used to deliver small volumes of drug (≤3 ml). Darts propelled quietly and cause minimal trauma. Precise delivery location usually possible. Discrete appearance unlikely to attract public attention when used in urban settings. Long pipes increase accuracy. Risk of drug exposure by mouth. Use in some localities prohibited or requires legal authorization.	Brockelman and Kobayashi 1971 Haigh and Hopf 1976 Ryser *et al.* 2005

		"Powered" blowpipes using compressed air or gas can project darts up to 30 m. Remote-controlled, "powered" blowpipe used to capture Eurasian lynxes (*Lynx lynx*; Ryser *et al.* 2005).	
Long bows and cross bows	≤50 m	Used to deliver ≤5 ml of drug to large animals. Commercially-available darts mountable on arrow shafts using adapter. Can project darts long distances, but difficult to prevent trauma caused by high velocity impact (Hawkins *et al.* 1967). Of limited use given wide availability of accurate dart guns that cause less trauma.Modified long bow used to capture lions and leopards (*Panthera pardus*; Stander *et al.* 1996).	Short and King 1964 Hawkins *et al.* 1967 Stander *et al.* 1996
Dart guns	≤90 m	Modified pistols, shotguns, rifles, and custom-designed projectors for propelling drug-filled darts into specific target animals.	Crockford *et al.* 1957 Smuts *et al.* 1977
		Used to deliver drug to trapped carnivores and to capture free-ranging carnivores (≥20 kg) from ground or helicopter. Darts propelled by gas generated from a .22-caliber blank cartridge, by compressed CO_2 or N_2, or by compressed air. Can deliver 1–10 ml of drug for compressed gas/air-powered guns and up to 20 ml for .22-caliber-powered guns. Effective range less for pistols than other types of projectors, and less for compressed gas/air-powered guns than for .22-caliber-powered guns. Darts fired at high velocity can cause injury or death (Valkenburg *et al.* 1999; Cattet *et al.* 2006).	Ballard *et al.* 1982 Kreeger 1999 Valkenburg *et al.* 1999 Cattet *et al.* 2003 Holekamp and Sisk 2003 Cattet *et al.* 2006 Fahlman *et al.* 2008
Remote injection collars	≤25 000 m	Used to recapture animals by remote-controlled injection of drug contained in a syringe mounted on a collar.	Mech *et al.* 1984 Mech and Gese 1992 Jessup 1993 Federoff 2001 Powell 2005

(continued)

Appendix 5.4 *Continued*

Method	Distance from Target Animal	Description	Specific References[1]
		Useful for repeated immobilization of free-ranging animals difficult to dart. Communication with collar greater by air (<25 km) than by ground (<3 km). Used by to recapture wolves and black bears (Mech and Gese 1992; Powell 2005). No longer available commercially.	

[1] *Specific references often chosen as representative examples from a broader selection of literature. Readers seeking additional references or details on use of equipment and manufacturer information should review comprehensive works by Nielsen (1999), Kreeger and Arnemo (2007), West* et al. *(2007), and Fowler (2008).*

Appendix 5.5 *Medical emergencies and complications that can occur with capture and handling of carnivores. Comments provide a definition or description (D) of the emergency or complication and brief summary information concerning clinical signs (S), causes (C), prevention (P), and treatment (T).*

Emergency or Complication[1]	Comments[2]
Physical injury	D: Abrasions (scrapes), contusions (bruises), concussion, dislocation or fracture of bones, lacerations (cuts), and punctures. S: Depends on type of injury—may see blood, reduced mobility, limb irregularities • Animal may vocalize frequently in response to pain. C: Darts • Marking techniques • Prolonged physical exertion (from pursuit or resisting restraint) • Prolonged induction of anesthesia • Rough handling • Sample collection • Self-infliction • Surgery • Telemetry devices (collar, ear tag, implantable) • Traps. P: Animal care approval of capture and handling protocol • Appropriate selection of capture method and equipment for target species • Appropriate training and expertise in capture and handling techniques • Check traps frequently • Perform capture and handling procedures quickly and efficiently. T: Basic first aid for minor injuries • Veterinary care, or possibly humane killing, for major injuries.
Thermal stress (hyperthermia)	D: Increase in body temperature to point where oxygen uptake is insufficient to meet need of oxygen for cellular metabolism. S: Rise in body temperature to >40ºC (104ºF) • Rapid, shallow breathing (panting) • Rapid heart rate • Warm extremities • Convulsions • Death. C: Concurrent disease • Drug effect (inhibition of thermoregulation) • External heat absorption (solar radiation) • Fear • Improper trap site prevents behavioral thermoregulation • Prolonged physical exertion (from pursuit or resisting restraint) • Species-specific factors (mass, surface-to-volume ratio, insulation) • Weather (high ambient temperature, no wind). P: Avoid capturing animals on hot days • Avoid prolonged pursuit • Check traps frequently • Minimize stress (consider providing sedation) • Monitor rectal temperature frequently, e.g. every 5–10 min • Protect animals/traps from direct exposure to sun • Use immobilizing drugs with effects that can be terminated by administering appropriate antagonist drugs. T: Administer antagonist drug • Administer cold water enema • Administer cold lactated Ringers by intravenous route • Administer oxygen • Apply external cold sources to areas of greatest heat exchange • Immerse animal in cold water • Provide adequate ventilation, e.g. circulate air around animal with a fan • Spray body surface with cold water.

(continued)

Appendix 5.5 *Continued*

Emergency or Complication[1]	Comments[2]
Thermal stress (hypothermia and frostbite)	D: Decrease in body temperature to point where cellular death occurs due to decreased metabolism and/or freezing of tissue. S: Decrease in body temperature to <35ºC (95ºF) • Shivering • Cold extremities (also firm with frostbite) • Dullness or lack of behavioral responsiveness to stimuli • Decreased heart rate • Shock • Coma • Death. C: Drug effect (inhibition of thermoregulation, decrease in metabolism) • Improper trap site prevents behavioral thermoregulation • Loss of insulation (poor body condition, wet fur) • Prolonged immobility • Prolonged restraint on a cold surface • Species-specific factors (mass, surface-to-volume ratio, insulation) • Weather (low ambient temperature, high wind, precipitation). P: Apply external heat sources, e.g. hot water bottles, warming blankets, chemical heat packs • Avoid capturing animals on cold days • Check traps frequently • Insulate animals from cold, wet surfaces • Monitor rectal temperature frequently, e.g. every 5–10 min • Protect extremities of anesthetized animals from frostbite • Shelter animal from wind and precipitation • Use immobilizing drugs with effects that can be terminated by administering appropriate antagonist drugs. T: Administer antagonist drug *before* animal becomes hypothermic or after it is re-warmed, but not when it is hypothermic • Administer warm water enema • Administer warm physiological saline by intravenous route • Apply external heat sources, e.g. hot water bottles, warming blankets, chemical heat packs • Dry wet fur.
Dehydration	D: Excessive loss of body water. S: Depends on severity of dehydration—may see dryness of mouth (including gums), loss of skin elasticity, sunken eyes, fever, weak pulse, shock, coma, death. C: Decreased water intake • Fever (due to pre-existing illness) • Hyperthermia • Increased water loss (due to panting or persistent vomiting, diarrhea, urination, or bleeding). P: Avoid trapping on hot days • Avoid prolonged pursuit • Check traps frequently • Minimize stress (consider providing sedation) • Protect animals/traps from direct exposure to sun. T: Estimate amount of fluid lost • Administer fluids (lactated Ringer's solution or physiological saline) by intravenous, subcutaneous, or intraperitoneal routes.
Hypoxia (hypoxemia)	D: Decreased availability of oxygen in blood (hypoxemia) or more generally in body tissues (hypoxia) • A common complication of anesthetized animals, less likely to occur in non-anesthetized animals.

	S: Labored or difficult breathing • Blue (cyanotic) mucous membranes • Hemoglobin oxygen saturation (measured by pulse oximeter) <80% for more than 1 min • Rapid pulse • Unconsciousness • Convulsions • Death. C: Concurrent respiratory disease • Drug-induced depression of respiratory function • Excessive pressure applied to the thoracic cavity (chest) • Obstruction of airways, including nostrils • Regurgitation and aspiration of stomach content. P: Administration of appropriate drug dose • Monitor mucous membrane color and hemoglobin oxygen saturation frequently, e.g. every 5–10 min • Position anesthetized animals correctly • Use proper restraint and handling techniques. T: Administer antagonist drug • Administer oxygen • Correct mechanism causing hypoxia, e.g. reposition body, remove pressure from chest, etc.
Acidosis	D: Disturbance of normal acid-base balance resulting in a blood pH <7.35. S: Rapid breathing • Confusion • Convulsions • Coma • Death. C: Intense or prolonged physical exertion resulting in excessive accumulation of lactic acid (metabolic acidosis) • Obstruction of airways resulting in excessive accumulation of carbon dioxide (respiratory acidosis). P: Avoid prolonged pursuit • Check traps frequently • Position anesthetized animals correctly • Use proper restraint and handling techniques. T: *For metabolic acidosis:* Administer sodium bicarbonate by intravenous route in conjunction with other fluids (physiological saline or dextrose). *For respiratory acidosis:* Assist respiration by artificial ventilation • Establish open airways • Re-position anesthetized animal correctly.
Regurgitation/vomiting and aspiration	D: Passive flow (regurgitation) or forceful ejection (vomiting) of stomach content into the mouth followed by inhalation of regurgitated material into the airways (aspiration). S: Presence of stomach contents in nostrils or mouth • Respiratory distress (gagging, retching, gurgling) • Fever • Death. C: Drug-induced relaxation of stomach sphincter • Improper restraint, handling, or positioning • Recent feeding followed by intense exertion • Stress. P: Avoid pursuing feeding animals • Minimize stress • Position anesthetized animals correctly • Use proper restraint and handling techniques. T: Avoid prolonged immobilization with animal lying on its side • Quickly clear mouth and airway of regurgitated or vomited material • If aspiration has occurred, seek veterinary assistance or consider humane killing.

(continued)

Appendix 5.5 *Continued*

Emergency or Complication[1]	Comments[2]
Shock	D: Failure of blood circulation resulting in ineffective perfusion of tissues. S: Rapid heart rate • Low blood pressure (capillary refill time >2 s) • Shallow, rapid breathing • Bluish pale mucous membranes. C: Concurrent illness • Prolonged physical exertion • Prolonged stress • Severe blood loss. P: Avoid prolonged pursuit • Check traps frequently • Minimize stress (consider providing sedation) • Monitor cardiovascular function (heart rate, pulse, capillary refill time, mucous membrane color, and hemoglobin oxygen saturation) frequently, e.g. every 5–10 min. T: Administer antagonist drug • Administer corticosteroids (dexamethasone) intravenously • Administer fluids (lactated Ringer's solution or physiological saline) intravenously to increase blood volume and blood pressure • Maintain an open airway and provide oxygen • Monitor rectal temperature and keep animal warm.
Seizures/convulsions	D: Disturbance of brain function characterized by involuntary, violent contractions of skeletal muscles. S: Rigid extension of the limbs • Uncontrolled muscle spasms (may be focal or involve whole body) • Increasing body temperature (associated with intense muscular contractions). C: Drug-induced effect, e.g. side-effect of ketamine • Hyperthermia • Metabolic disturbances incited by capture and stress, e.g. hypocalcemia (low blood calcium), hypoglycemia (low blood glucose) • Trauma. P: Administration of appropriate drugs at appropriate doses • Apply same measures as taken to prevent hyperthermia. T: Administer benzodiazepine sedative (diazepam or midazolam) intravenously slowly • Monitor rectal temperature and take appropriate steps to prevent hyperthermia.
Respiratory arrest	D: Cessation of breathing. S: Slow, shallow breathing or cessation of breathing • Blue (cyanotic) mucous membranes • Hemoglobin oxygen saturation (measured by pulse oximeter) <80% for more than 1 min or downward trend in saturation values • Rapid pulse • Unconsciousness • Convulsions • Death. C: Drug-induced depression of respiratory function (possibly as a result of a severe overdose) • Excessive pressure applied to the thoracic cavity (chest) • Obstruction of airways, including nostrils. P: Administration of appropriate drug dose • Monitor respiratory function (respiratory rate and depth, respiratory sounds, mucous membrane color, and hemoglobin oxygen saturation) frequently, e.g. every 5–10 min • Position anesthetized animals correctly • Use proper restraint and handling techniques.

	T: Administer antagonist drug • Establish open airway • Provide artificial ventilation • Administer oxygen • Administer doxapram intravenously.
Cardiac arrest	D: Loss of effective heart function. S: Increased respiratory rate or cessation of breathing • Weak or absent heart sounds or pulse • Low blood pressure (capillary refill time >2 s) • Blue (cyanotic) mucous membranes • Dilated pupils • Cold skin • Unconsciousness • Death. C: Acid-base imbalance • Drug-induced depression of cardiovascular function • Electrolyte imbalance • Hypothermia • Respiratory arrest. P: Administration of appropriate drug dose • Avoid prolonged pursuit • Check traps frequently • Minimize stress (consider providing sedation) • Monitor cardiovascular function (heart rate, pulse, capillary refill time, mucous membrane color, and hemoglobin oxygen saturation). frequently, e.g. every 5–10 min. T: Ensure animal is breathing and, if not, establish open airway and provide artificial ventilation • External cardiac massage (60–100 cycles per min) • Administer epinephrine intravenously.
Exertional myopathy (capture myopathy)	D: A noninfectious disease characterized by degenerative or necrotizing damage to skeletal and cardiac muscles. S: Weakness and loss of muscle coordination • Hyperthermia • Rapid breathing • Rapid heart rate • Dark, brownish urine (myoglobinuria) • Sudden death • Delayed death occurring days or weeks following capture. C: Prolonged physical exertion • Prolonged stress. P: Avoid prolonged pursuit • Check traps frequently • Minimize stress (consider providing sedation) • Use drugs that induce muscle relaxation. T: Administration of sodium bicarbonate and fluids intravenously is sometimes recommended, but treatment is often unsuccessful.

[1] *The likelihood of some emergencies occurring may be determined in part by the method of capture. For example, hypoxia or aspiration of stomach content is much more likely to occur in anesthetized animals than trapped animals.*

[2] *Sources: Nielsen (1999), Cattet* et al. *(2005), Kreeger and Arnemo (2007), and Fowler (2008).*

Appendix 5.6 *Acceptable methods used to humanely kill wild carnivores.*

Method	Comments[1]
Gunshot	Can be used to kill captive, restrained, anesthetized, or free-ranging carnivores. Shooter must be able to make a clean killing shot, using the appropriate firearm and ammunition. Personnel should stand behind the shooter and away from the animal. Large, heavy, slow-moving bullets (e.g. shotgun slugs) are more effective and safer than high-power rifle bullets. Captive, restrained, and anesthetized animals should be shot in the brain, either from the front or side, or in the neck through the vertebral column if the brain needs to be preserved for disease diagnoses, e.g. rabies. For accurate bullet placement, preferable to place the barrel of the gun right on and perpendicular to the skull or neck. For human safety, best location for shooting free-ranging animals is the heart/lung region, rather than the head. Remove lead-contaminated carcasses or body parts from sites where consumption by scavengers can lead to secondary lead toxicity.
Penetrating captive bolt	Can be used to kill anesthetized carnivores. Less risk of injury to bystanders and nearby animals than gunshot. Safe use requires full immobilization of the animal's head, accurate placement of the captive bolt against the skull, equipment that is in good working order, and administration by fully trained personnel. Animals should be exsanguinated (bled out) after the use of a penetrating captive bolt to ensure death. Non-penetrating captive bolts are *not* recommended for humane killing.
Exsanguination (bleeding to death)	Acceptable *only* if animal has been rendered unconscious by drugs or stunning. Can be done quickly and effectively by severing the major arteries leading from the heart by inserting a long-bladed knife into the junction of the base of the neck and shoulder and slicing inwards and downward. Severing of the jugular or femoral veins may also be effective, but will take longer because of slower blood flow. Placing body on incline with head downward may help improve blood flow.
T-61	Can be used to kill anesthetized carnivores. Must be administered intravenously. Mixture of three drugs: embutramide (a general anesthetic), mebezonium iodide (neuromuscular blocker), and tetracaine hydrochloride (a local anesthetic). Not available in all countries, including the United States.
Barbiturates	Can be used to kill anesthetized carnivores. Several euthanasia products contain a barbituric acid derivative (usually sodium pentobarbital) often mixed with local anesthetic agents. Should be administered intravenously, but may also be administered by intraperitoneal or intrathoracic injection in small- to medium-sized carnivores (<50 kg). These drugs are controlled substances in many countries.

Method	Comments[1]
	Effective volume needed to euthanize large carnivores (>150 kg) can be high (>50 ml). Animals killed with any barbiturate must be incinerated or buried because of potential secondary toxicity to potential scavengers.
Potassium chloride	Can be used to kill deeply anesthetized carnivores. Must be administered intravenously quickly. Kills by increasing concentration of circulating potassium in the blood which directly influences the electrical activity of the heart resulting in cardiac arrest. Potassium chloride solution is made by adding 300 mg potassium chloride per ml of water (tap or distilled) and shaking vigorously prior to injection. Administer at a dosage of at least 50 mg per kg body weight.

[1] *Sources: AAZV (2006), Kreeger and Arnemo (2007), and AVMA (2007).*

Appendix 5.7 *Temporary and permanent marking techniques used to identify carnivores.*

Mark	Technique	General References
Temporary Short-term markers		
Fur clipping and dyeing	Shaving in patterns to reveal underfur of animals. Clipping does not affect body condition, but must be cautiously applied with individuals in poor condition or in cold climates. Hair can be dyed to mark carnivores of all sizes and is particularly useful for mammals with light pelage. Rhodamine B has been used on coyotes (Johns and Pan 1981). Whether clipping or dying fur affects responses to the environment (e.g. physical protection or thermoregulation) or increases visibility of small carnivores to larger predators is unknown.	Ramsay and Stirling 1986 Stewart and Macdonald 1997 Macdonald *et al.* 2004a
Body attachments	Streamers and colored disks of different lengths and color codes attached to a carnivore's body or to ear tags allow identification from distance.	Lentfer 1968 Powell and Proulx 2003
Temporary long-term markers		
Tags	Tags made from metals or plastics of all shapes, sizes, and colors, and stamped with letters, numbers, and short messages, have been affixed to ears, and less frequently to other body parts, such as interdigital webbing (Gorman *et al.* 2006). Ear tags vary in size to accommodate different carnivores of all sizes. Ear tags can be pulled out by animals grooming each other (Stirling 1989) or may catch on vegetation (Hubert *et al.* 1976). Turning metal, crimping ear tags in a mammal's ear so that the clasp is outermost appears to minimize loss (Powell, unpubl. data). Retention of tags varies among species and habitats, but putting ear tags in both ears of an animal and using redundant marking to overcome identification problems are recommended. Ear tags should be lose enough not to interfere with blood circulation and punctures should be treated appropriately to prevent infection and to ensure healing.	Nietfeld *et al.* 1994 Powell and Proulx 2003

Collars and harnesses	Conspicuous metal (copper or brass non-corrosive alloy) or plastic collars with printed instructions, numbered tags, or color codes. Self-collaring collars may be placed like snares along animal trails. Collars may need replacement at regular intervals. Neck collars outfitted with radio transmitters allow researchers to identify animals, to follow their movements, and to investigate population structures and dynamics (Chapter 6).	Sheldon 1949 Zabel and Taggart 1989 De Luca and Ginsberg 2001
Passive integrated transponder (PIT tag)	A PIT tags consists of an electromagnetic coil and custom-designed microchip that emits an analog signal when excited by electromagnetic energy from a scanning wand. The transponder chip is uniquely programmed with an alpha or numeric code, and $>$34 billion combinations are available. Once inserted under a mammal's skin with a large bore syringe, a PIT tag (2 mm diameter $\times$ 10 mm length) can be read by a scanner. PIT tags are relatively expensive and require a specific scanner matched to the tag type to read the identification. They must be read within 10 cm of a wand. A new generation of PIT tags with antennas may be read at greater (up to 0.5 m) distance (e.g. Kurth *et al.* 2007). PIT tags may wander under an animal's skin, especially on large mammals but they are advantageous alternative to ear tags that may be lost during long-term studies. PIT tags have been successfully used with skunks (Neiswenter and Dowler 2007), Eurasian badgers (Rogers *et al.* 2002), ferrets (*Mustela fero*, *M. nigripes*) (Fagerstone and Jones 1987), Iberian lynxes (*Lynx pardinus*) (Palomares *et al.* 2001), and black bears (Leigh 2007).	Nietfeld *et al.* 1994 Powell and Proulx 2003 Jones *et al.* 2004
Radioactive and chemical markers	A variety of mammals have been marked with radioisotopes as inert implants, as external attachments, and as metabolizable radionucleoides, all of which can be detected in tissues, feces or urine. Rhodamine B is a systemic fluorescent marker that can be detected in hair, whiskers, claws, and other tissues, and it has been used as a qualitative marker of bait consumption in	Nellis *et al.* 1967 Pelton and Marcum 1975 Linn 1978 Savarie *et al.* 1992 Jones *et al.* 2004

(continued)

Appendix 5.7 *Continued*

Mark	Technique	General References
	coyotes (Johns and Pan 1981). Iophenoxic acid is an organic iodine chemical that binds to proteins in the blood, elevating the level of protein-bound iodine. It has been found an effective marker for ferrets, raccoons, coyotes, arctic foxes (*Vulpes lagopus*), and red foxes (Larson *et al.* 1981, Follman *et al.* 1987; Hadidian *et al.* 1989; Ogilvie and Eason 1998).	
Betalights	A betalight is a phosphor-coated glass capsule containing a small quantity of mildly radioactive tritium gas. When the phosphor is struck by low-level beta radiation from tritium, it produces visible light of a characteristic color. It may be used with other markers. They have been successfully used with Eurasian badgers (Cheeseman and Mallinson 1980; Tuyttens *et al.* 2000). They appear to pose no appreciable health hazard to radiation and may function for years.	Rudran 1996
Permanent markers		
Freeze branding	Cryo-branding has been used to mark carnivores. Freeze-branding applies a copper branding iron that is super cooled in liquid nitrogen, or a mixture of dry ice and alcohol, or a commercial refrigerant to a shaved area of the body. It kills the pigment-producing melanocytes of the skin but not the hair follicles, so the hair and skin that regrow in the branded area are permanently white.	Hadow 1972 Day *et al.* 1980 Rood and Nellis 1980 Sasaki and Ono 1994
Tattoos	Tattoos are applied with special pliers or an electric tattooing pencil. Any body part that is relatively free of hair and remains fairly clean can be tattooed, such as upper lips and the groin.	Powell *et al.* 1997 Diefenbach and Alt 1998 Walton *et al.* 2001b Powell and Proulx 2003

Chemical markers	Tetracycline antibiotics chelate with calcium ions in bones and teeth to produce characteristics patterns of fluorescence under UV light. They have been used to mark mustelids, procyonids, canids and ursids.	Linhart and Kennelly 1967 Nelson and Linder 1972 Bachmann *et al.* 1990 Garshelis and Visser 1997
Mutilations	Toe clipping, where the claw and first joint of the toe are removed with dissecting scissors, has been used to mark long-tailed weasels (*Mustela frenata*; DeVan 1982), arctic foxes (Roth 2002), and coyotes (Andelt and Gipson 1980). Toe-clipping could modify the behavior of animals due to pain or reduced function, and could, potentially, reduce success in foraging and competition, resulting in differential mortality. Even though toe-clipping is inexpensive and rapidly applied to very small carnivores, it should be considered only when no other marking method is appropriate. Debrot (1984) and Santos-Reis *et al.* (2004) clipped ears in unique patterns to mark mustelids and viverrids.	Henshaw 1981 ASM Care and Use Committee 1998