41 Otariid Seals

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INTRODUCTION

The family Otariidae (sea lions and fur seals) within the order Pinnipedia is composed of 14 species. Otariids bear weight on all four flippers, climb, locomote quickly, and are more adept on land than phocid seals. However, their aquatic adaptations are less developed and they generally do not dive as deep or for as long as phocids. Anatomical and physiological adaptations for diving (e.g., large venous sinuses and dive response) therefore, are not as extreme (Elsner, 1999; Pabst, Rommel, and McLellan, 1999). Some of these differences make otariids more difficult to physically or mechanically restrain than phocids of the same weight. Additionally, they are less sensitive to immobilization drugs and anesthetic regimens are similar to those of terrestrial carnivores.

PRE-ANESTHETIC CONSIDERATIONS

Planning

As with any species, successful otariid anesthesia is dependent upon adequate planning and availability of the proper equipment. The animal's size, species, sex, and physiological status are important considerations in choosing the best immobilization method. The site (captive facility versus free-living animals in the field), experience of the personnel, and availability of equipment and drugs often dictate the method chosen. Finally, the degree of invasiveness and expected duration of the procedure affect decisions.

Translocation without Restraint

Many otariid species are easily trained to follow trainers, and voluntarily enter different housing units or transport cages. Training of permanent captives is essential for enrichment, minimizing stress and facilitating medical procedures. Animals captive for only short periods of time, as in a wildlife rehabilitation center or research facility, require the use of protective equipment by handlers. Depending on size and species, animals can be safely moved with herding boards, chutes, and mobile fencing. They can be herded into cages or transport containers for longer travel. Free-living pups from a variety of species are safely herded into temporary corrals using long poles. Depending on the local topography, moving healthy adult otariids is very challenging and requires well-trained, experienced personnel.

Physical Restraint

Physical restraint is primarily limited by the animal's size and the experience of personnel. Training captive animals for a variety of behaviors minimizes the requirement for physical restraint. Towels, protective gloves, or sedative drugs may facilitate restraint.

Multiple personnel are required to restrain animals greater than or equal to 20 kg. Mechanical or drug assisted restraint is strongly recommended for healthy untrained animals greater than or equal to 90 kg. Larger debilitated animals, such as those encountered at rehabilitation centers, may be restrained using only physical methods. The limitations of physical restraint in otariids include short allowable duration of procedures, poor accessibility to various parts of the animal, lack of analgesia, risk of injury to personnel, and undue stress to the animal as a result of prolonged struggling.

Otariids are quick, agile, and strong and use their large carnivorous teeth in defense. Controlling the head is essential. Sea lions have tremendous power in their forelimb muscles. If they gain a purchase, and are able to lift their thorax off the ground, they will easily throw off a person. Care is taken to control the front flippers, raising them slightly and holding them against the side of the animal. This is particularly important in animals greater than or equal to 20 kg. Animal safety concerns include ensuring airway integrity and the head is not turned at an awkward angle or held over an edge that may collapse the trachea. Too much weight on the

thorax inhibits ventilation. Musculoskeletal problems arise from excessive restraint on the extremities. Care must be taken to avoid soft-tissue injuries, including abrasions and scrapes, if restraint is on a rough substrate.

Mechanically Assisted Restraint

There are several commercially available large carnivore squeeze cages that have been used successfully in a variety of otariid species. Although many designs exist, squeeze cages that entrap the animal's neck in a vise or noose put pressure on the trachea and should be avoided. The otariid trachea easily collapses because of incomplete tracheal rings. When using squeeze cages, it is important to closely monitor the respiratory excursions and mucous membrane color of the animal. Care is taken to avoid pinching extremities, especially as the cage begins to squeeze. The cage bars may be padded to avoid soft-tissue injury. Biting the bars results in tooth fracture and should be avoided. Captive animals can be trained to enter a cage and be squeezed to allow sample collection (e.g., for venipuncture). Some animals have been trained to accept delivery of an anesthetic gas through a mask without further restraint. As with physical restraint, sedative agents may decrease the degree of struggling and facilitate restraint.

There are a variety of commercially available nets custom-designed for use in otariids. Nets aid in capturing animals, can be used for restraint, and facilitate administration of chemical immobilization drugs. Well-designed nets are somewhat tubular to keep the pectoral limbs against the side and prevent the animal from lifting itself up. Nets should be wide enough to easily capture the animal, but taper to a point so the head can be easily controlled. In the field, care must be taken to monitor an animal in a net for hyperthermia and exhaustion after capture. Animals have overheated in tightly fitting nets left in the sun while other animals are being sampled or processed. Some nets have small openings at the tip that allow exposure of the nares and rostrum but are tight enough to prevent the animal from biting personnel. These openings aid in masking an animal with an inhalant anesthetic after capture.

Recently, a method of capturing free-ranging Steller sea lions (*Eumetopias jubatus*) in the water was developed using a team of divers, baited nooses attached to floats, and a surface capture team in boats (Raum-Suryan et al., 2004). This capture method requires a tremendous amount of training and planning and should only be carried out by highly experienced staff.

Although most methods of mechanical restraint enhance safety for personnel, training and experience are required to ensure adequate and safe restraint. Most mechanical devices, including nets, limit access to the animal for some procedures.

Chemical Restraint

Venous access in many species is difficult to maintain for intravenous (IV) injection. This is particularly true for difficult to restrain animals and under field conditions (Work et al., 1993). Most anesthetic drugs evaluated for use, therefore, have been limited to those that can be administered IM (Bester, 1988; Loughlin and Spraker, 1989; Heard and Beusse, 1993; Heath et al., 1996) and inhalant anesthetics (Work et al., 1993; Heath et al., 1997; Yamaya et al., 2006).

Intramuscular drugs are injected into the large muscle masses overlying the lower lumbar spine (Bester, 1988; Loughlin and Spraker, 1989), tibia and hips (Loughlin and Spraker, 1989; Heard and Beusse, 1993; Sepulveda, Ochua-Acuna, and McLaughlin, 1994; Haulena et al., 2000) as well as the shoulders (Loughlin and Spraker, 1989). Immobilizing intramuscular (IM) agents have been hand-injected or delivered by dart. Physical or mechanical restraint may facilitate accurate hand injection of anesthetic agents. Delivery by dart has been used in captive animals (Haulena et al., 2000) as well as in the field (Heath et al., 1996). In free-living animals, delivery by dart poses some risk to animals that may escape to the water or an inaccessible area as the anesthetic drug begins to take effect prior to complete immobilization (Heath et al., 1996). Some authors report a decrease in the reliability of anesthesia when darts are employed, in comparison to hand injection (Haulena et al., 2000).

Inhalation anesthetics (e.g., isoflurane) appear to be the safest method for anesthetizing otariids because of the ability to titrate the level of drug to effect. The main limitation is the availability and portability of equipment to safely and reliably deliver the anesthetic in the field. In addition, delivery of a gas for a sufficient period of time to induce anesthesia may be difficult in a fractious, unrestrained animal. Most public display facilities, rehabilitation centers, and research facilities are equipped for delivery of inhalant anesthetics. A recent publication describes the use of an induction chamber for delivery of inhalant anesthesia to sea lions (Yamaya et al., 2006). The development of safe, portable gas anesthesia machines for field work has greatly increased the use of gas anesthesia in free-living species. A comprehensive animal training program, adequate physical or mechanical restraint, or the use of chemical sedative and immobilizing agents (Heard and Beusse, 1993; Heath et al., 1996; Haulena et al., 2000; Haulena and Gulland, 2001) facilitates the use of inhalation anesthesia.

Although chemical immobilization is the safest method of restraint for personnel, allowing complete access to the entire animal, there is some risk to the animal being anesthetized. This risk is minimized by adequate preparation, use of experienced anesthetists, use of safe and efficacious immobilization agents, careful monitoring, and physiological support of the animal.

Immobilization Location

As with phocid seals, otariid immobilization success is greatly enhanced by good planning, proper equipment, experienced staff, and a well-prepared space. The ability

to safely handle, restrain, and deliver anesthetic agents to the animal is essential. A surgery or procedure area that includes an accessible immobilization and recovery pen for monitoring or emergencies is recommended. The ability to control temperature, noise, and light will help induction and physiological support of the patient. Immobilization of larger animals may that require procedures be carried out in the animal's pen because of space and transport limitations. Adequate pre-procedure planning and availability of emergency equipment are essential for a safe procedure.

Immobilization of animals in the field presents significant challenges. However, portable gas anesthesia machines, battery-operated monitoring equipment, and emergency equipment are all available (Gales and Mattlin, 1998). If injectable chemical immobilization is required prior to adequate physical or mechanical restraint, it is essential that animals selected for capture are as far as possible from water or other hazards. It is also important to choose animals that are relatively calm and have the least risk of escaping into the water or inaccessible areas (Heath et al., 1996). This will help minimize the risk of drowning or falling from large heights.

Assessment of Physiological Status

Pre-anesthetic assessment is important for selection of the optimal method of immobilization and to decrease the incidence of adverse side effects. It is often very difficult to obtain a complete medical history for an individual animal, particularly for free-living animals or those undergoing rehabilitation. A medical history for captive animals may be available. However, even for these animals, the purpose of the immobilization procedure may be to gather information and samples to diagnose an unknown condition. Therefore, it is important to try to select animals that are in good body condition and of known health status for elective procedures or for field studies. In addition, it is important to have some knowledge of the common medical conditions that affect both captive and free-living animals in order to facilitate pre-procedure planning.

Some problems are specific to species, sex, age, season, or geographic location. For example, most stranded animals entering a rehabilitation program are dehydrated, malnourished, and may have infectious disease conditions that should be stabilized prior to an anesthetic procedure. Particular attention to these potential problems will lead to better intra-procedure physiological support. Free-living animals, particularly juveniles, may have high parasite loads. Parasitic pneumonia caused by *Parafilaroides decorus* in species such as the California sea lion (*Zalophus californianus*) (Gage et al., 1993) may exacerbate ventilation problems encountered during an anesthetic procedure. Young animals such as northern fur seals (*Callorhinus ursinus*) may be affected by hookworm (*Uncinaria* sp.), which can cause anemia (Lyons et al., 2000). These animals are prone to hypoxemia and vascular compromise. Dehydrated juvenile and subadult California sea lions stranded during the late summer and early fall are often affected by leptospirosis, which may cause renal failure (Gulland et al., 1996). Poor renal function can significantly alter the excretion of some parenteral anesthetics. Animals with clinical leptospirosis are poor anesthetic candidates. Animals in captivity may live longer than free-living animals and may be prone to developing progressive organ failure similar to domestic species.

MONITORING

An anesthetic plan is developed based on available history, knowledge of the species, and any available laboratory data. Monitoring physiological variables (especially cardiopulmonary) should be begun as soon as possible after induction and throughout the procedure. Most commonly used monitors can be adapted for use in the otariid (see Chapter 7). Variables that are commonly measured include heart and respiratory rates, capillary perfusion, response to painful stimuli, body temperature, hemoglobin saturation (relative SpO_2), end-tidal carbon dioxide ($ETCO_2$), blood pressure, and blood gas levels. For otariids, the trends in the measured variables are more important than the point measurements.

Heart rate is one of the most important variables to monitor. It is determined by either palpation or observation of thoracic wall movement over the heart, just caudal to the axilla. Chest auscultation can be used, but thoracic noise is muffled compared to terrestrial mammals. Heart rate can also be determined using electrocardiogram (ECG) leads placed externally (Heard and Beusse, 1993) or attached within an esophageal probe (Haulena and Heath, 2001). Pulse oximeters also generate a pulse wave that can be used to calculate heart rate. However, the pulse wave does not reflect adequacy of tissue perfusion (see Chapter 7). The pulse oximeter probes can be clipped to the tongue (Heard and Beusse, 1993; Heath et al., 1996, 1997; Haulena et al., 2000), but the clips tend to slip off the short, thick tongue. Probes are placed on the nasal septum of larger animals. Reflectance probes can be placed rectally, vaginally, or along the buccal or gingival mucosa (Heath et al., 1996). Sudden or progressive bradycardia may be an early indication of initiation of the dive reflex. Some drugs, particularly the α_2-agonists, also cause bradycardia.

Respiratory rate is measured by observing thoracic movement, opening of the nares, or chest auscultation. Capnography and respiratory monitors can be used to electronically calculate respiratory rate. Apnea is common in anesthetized otariids (Sedgwick, 1999) and may result from excessive anesthetic, the immobilizing drug used, or the dive reflex.

Anesthetic depth is assessed using response to various stimuli such as noise and deep pain (interdigital web pinch, ear pinch, or surgical stimulation), presence or absence of the palpebral and pupillary reflexes,

Table 41.1. Blood gas variables from the caudal gluteal vein in ten physically restrained California sea lions (*Zalophus californianus*).

Variable	Mean ± SD	Range
Na	149 ± 3	146–152
K	4.3 ± 0.5	3.6–5.3
TCO_2	23 ± 5	17–29
iCa (mmol/L)	1.20 ± 0.07	1.07–1.31
Hct (%)	45 ± 5	37–51
Hb (g/dl)	16 ± 2	13–17
pH	7.31 ± 0.05	7.22–7.38
PCO_2 (mmHg)	43.9 ± 4.9	38.4–53.8
PO_2 (mmHg)	74 ± 21	45–103
HCO_3 (mmol/L)	22 ± 4	16–27
BE	–4 ± 5	–11–2
SO_2 (%)	91 ± 7	77–97

respiratory rate rhythm, tidal volume, and jaw tone (Work et al., 1993; Heath et al., 1996, 1997).

Peripheral body temperature measurement does not accurately reflect core because of the thick layers of insulating blubber. Flexible temperature probes are inserted at least 10 cm into the rectum of the animal (Bester, 1988; Loughlin and Spraker, 1989; Ferreira and Bester, 1999). Alternatively, esophageal probes inserted to the level of the heart (Heath et al., 1997) may give accurate core temperature readings.

Mucosal membranes (oral, rectal, and vaginal) are used for monitoring color and capillary refill time as indicators of perfusion and oxygenation (Work et al., 1993; Heath et al., 1997). Capnometer probes are attached to the endotracheal tube via filter line (Heard and Beusse, 1993; Haulena and Gulland, 2001). Elevations or sudden decreases in $ETCO_2$ levels may indicate ventilation and perfusion problems.

Non-invasive, oscillometric blood pressure monitoring is performed by attaching cuffs to the proximal portion of the limbs or the base of the tail. Venous blood gas samples are collected from the caudal gluteal, interdigital, or common jugular veins. Blood gas values obtained from 10 healthy California sea lions under physical restraint (Table 41.1) indicate that arterial blood is sometimes obtained from the area of the caudal gluteal vein (Haulena et al., 2001).

SUPPORTIVE CARE

Antimuscurinics

Atropine (0.02 mg/kg IM) has been recommended 10 minutes prior to immobilization to prevent bradycardia associated with the dive reflex in anesthetized otariids (Gage, 1993; Heath et al., 1996). Atropine has also been administered after injection of sedatives to control airway and oral secretion and prevent bradycardia (Spelman, 2004). However, α_2-agonists such as medetomidine cause bradycardia. Use of atropine with medetomidine is contraindicated in terrestrial mammals (Cullen, 1996).

Endotracheal Intubation

Endotracheal intubation is strongly recommended for any prolonged procedure that requires a surgical plane of anesthesia (Work et al., 1993; Sedgwick, 1999). Otariid intubation is easier than in phocids, resembling intubation of terrestrial carnivores. In general, endotracheal tubes are of similar diameter to those that would be used on terrestrial carnivores of the equivalent mass. Care is taken to ensure endotracheal tubes do not extend past the pre-thoracic bifurcation of the trachea, resulting in unilateral lung intubation (McGrath et al., 1981). The mouth is opened with soft nylon straps or rope. The head and neck are held straight and in a slightly hyperextended (opisthotonic) position. Ensure table edges or other equipment does not compress the trachea and interfere with passage of the endotracheal tube (Lynch, Tahmindjis, and Gardner, 1999). Gentle manipulation is used to prevent trauma to the larynx. Standard laryngoscopes facilitate visualization of the airway (Heard and Beusse, 1993; Haulena et al., 2000). Very large adults can be intubated by manual palpation of the epiglottis (Heath et al., 1996). Cuffed endotracheal tubes are used to prevent aspiration. It is important not to cause tracheal injury by over-inflating the cuff. Endotracheal tubes can be secured over the maxilla or mandibles using rolled gauze, rope, or tape passed caudal to the canine teeth.

Thermoregulation

Body temperature fluctuations resulting in either hypothermia or hyperthermia can occur in anesthetized otariids. It is particularly important to monitor temperature in field conditions in which control of the environment is more difficult. Temperature changes are influenced by the drugs used, the species, size, geographic location, and physiological status of the animal. For example, larger animals have a greater tendency to hyperthermia than smaller animals (Work et al., 1993). Some drugs (e.g., ketamine) may cause hyperthermia (Sepulveda, Ochua-Acuna, and McLaughlin, 1994), whereas others (e.g., isoflurane) cause vasodilation that promotes hypothermia (Loughlin and Spraker, 1989; Work et al., 1993). Profound hyperthermia was seen in a late-term pregnant California sea lion that was anesthetized using medetomidine and ketamine. Shelter from rain, wind, and sun, although maintaining adequate ventilation, is recommended to prevent temperature irregularities. A variety of commercially available heating blankets, heated surgical tables, hot water bottles, wraps, and insulating pads can be used to prevent hypothermia (Work et al., 1993; Sepulveda, Ochua-Acuna, and McLaughlin, 1994; Heath et al., 1997). Ice or cold water applied to extremities is used to treat hyperthermia.

Vascular Access

Vascular access for placement of catheters for fluid and emergency drug administration is difficult in small animals, hypothermic individuals, or otariids anesthetized with certain drugs (e.g., α_2-agonists). Some species have readily accessible interdigital veins in the pelvic limbs. However, these are not accessible in some species (e.g., California sea lion). Other accessible veins for catheterization include the cephalic, jugular, subclavian, and vessels running along the digits of the hind flipper. An indwelling catheter was maintained by the author for 4 days in a juvenile California sea lion using the common jugular vein.

Ventilation

Low SpO_2 values (less than 85%) have been reported in sea lions immobilized with zolazepam/tiletamine (Heath et al., 1996) and medetomidine/ketamine (Haulena et al., 2000). This effect is greater in animals not intubated and provided supplemental oxygen. Conversely, sea lion pups maintained with isoflurane in oxygen maintained higher SpO_2 values (Heath et al., 1997). This may be result from the drugs used, anesthetic depth, or animal's physiology. Low SpO_2 levels indicate the anesthetist should be prepared to intubate, provide oxygen therapy, and assist ventilation.

High $ETCO_2$ (greater than 70 mmHg) levels (Heard and Beusse, 1993; Haulena and Gulland, 2001) associated with acidemia (pH less than 7.15) in anesthetized California sea lions support the need for assisted mechanical ventilation in some animals (Haulena, Heath, and Gulland, 2001). Some drugs are more commonly associated with hypoventilation and hypercapnia. Animals anesthetized for prolonged periods, maintained at deep anesthetic planes and positioned in a manner that interferes with normal thoracic expansion, are particularly prone to developing hypercapnia. Conversely, hyperventilation of California sea lions has resulted in alkalemia (pH greater than 7.5).

In anesthetized otariids, mechanical ventilation is recommended at a starting tidal volume of 15 ml/kg and a rate of 8 to 10 breaths per minute (Haulena, Heath, and Gulland, 2001). Capnometry is essential with mechanically assisted ventilation to adjust tidal volume and rate to maintain normocapnia.

SEDATION

The use of a variety of IM sedative drugs may facilitate physical or mechanical restraint and aid induction with other drugs (e.g., isoflurane) (Gales, 1989). Oral diazepam (0.1–0.2 mg/kg) used prior to transport aids physical restraint of some animals. More reliable sedation is achieved with midazolam in California sea lions (0.15–0.2 mg/kg IM) and fur seals (0.25–0.35 mg/kg IM) (Lynch, Tahmindjis, and Gardner, 1999). Benzodiazepines can be reversed with flumazenil (Karesh et al., 1997). Butorphanol (0.05–0.2 mg/kg IM) has been used for mild sedation and analgesia. Combination of midazolam and butorphanol results in an increased level of sedation. Medetomidine (70 µg/kg IM) is recommended for sedation of sea lions for electroencephalography because of its apparent lack of interference with brain wave patterns (Dennison et al., 2005). Although sedation was variable, placement of multiple percutaneous leads for recordings was accomplished for more than 30 minutes.

CHEMICAL IMMOBILIZATION

Drugs commonly used to immobilize otariids are discussed in the following section. Table 41.2 summarizes drug dosages from recent studies. Several reviews have been written and should be referred to for a complete list of pinniped immobilization methods (Gales, 1989; Williams, Williams, and Stoskopf, 1990; Lynch, Tahmindjis, and Gardner, 1999; Haulena and Heath, 2001).

Zolazepam/Tiletamine

The advantages of this combination include small injection volume, low cost, and dependable deep sedation and immobilization. However, some studies report significant mortality, prolonged recovery, and a narrow margin of safety (Heath et al., 1996; Dabin, Beauplet, and Guinet, 2002). Zolazepam/tiletamine (1.7 mg/kg IM) in California sea lions is administered 10 minutes after atropine (0.02 mg/kg IM) (Gage, 1993). A slightly lower dosage (0.9–1.3 mg/kg IM) is recommended in subantarctic fur seals (*Arctocephalus tropicalis*). Additional "top-up" doses to increase anesthetic depth have been associated with increased mortality (Heath et al., 1996). However, additional ketamine has been used successfully without mortality (Karesh et al., 1997). The combination is partially reversed with flumazenil (Karesh et al., 1997).

Medetomidine/Ketamine

In California sea lions, the combination of medetomidine (140 µg/kg IM) and ketamine (2.5 mg/kg IM) provides effective and safe immobilization that is reversed by atipamezole (0.2 mg/kg IM). Animals were premedicated with atropine (0.02 mg/kg IM). Disadvantages of this combination in sea lions include moderately variable anesthetic depth, large injection volume when commercially available products are used, and high cost (Haulena et al., 2000).

Medetomidine/Zolazepam/Tiletamine

The combination of medetomidine (70 µg/kg IM) and zolazepam/tiletamine (1 mg/kg IM) produced reversible (atipamezole 0.2 mg/kg IM), reliable anesthesia (Haulena and Gulland, 2001). Injection volume and cost were much less than for medetomidine/ketamine.

Table 41.2. Parenteral and inhalant anesthetic drug dosages in otariids.

Species	*N*	Drug(s)	Dosage	Route	Mortality	Comments	Reference
Arctocephalus australis South American fur seal	32	Tiletamine/Zolazepam	1.43 mg/kg	IM dart	0%	Partial reversal with flumazenil	Karesh et al., 1997
Arctocephalus australis South American fur seal	4	Tiletamine/Zolazepam, Ketamine	1.43 mg/kg 0.81 mg/kg	IM dart IM	0%	Supplemental ketamine given owing to insufficient sedation; partial reversal with flumazenil	Karesh et al., 1997
Arctocephalus australis South American fur seal	8	Tiletamine/Zolazepam, Ketamine	1.15 mg/kg 0.27 mg/kg	IM dart	0%	All administered together; partial reversal with flumazenil	Karesh et al., 1997
Arctocephalus australis South American fur seal	1	Ketamine/Midazolam	1 mg/kg 0.1 mg/kg	IM dart	0%		Karesh et al., 1997
Arctocephalus forsteri New Zealand fur seal	5	Isoflurane	1.2%–4.0%	IH	0%		Gales and Mattlin, 1998
Arctocephalus gazella Antarctic fur seal	172	Tiletamine/Zolazepam	1.2–1.7 mg/kg	IM dart	3%	Respiratory depression	Boyd et al., 1990
Arctocephalus gazella Antarctic fur seal	30	Ketamine	6.9 ± 0.1 mg/kg	IM dart	0%	Muscle tremors	Boyd et al., 1990
Arctocephalus gazella Antarctic fur seal	23	Ketamine/Diazepam		IM dart IM dart	4%		Boyd et al., 1990
Arctocephalus gazella Antarctic fur seal	45	Ketamine/Xylazine	7.3 ± 0.3 mg/kg 0.6 ± 0.02 mg/kg	IM dart IM dart	7%		Boyd et al., 1990
Arctocephalus gazella Antarctic fur seal	14	Ketamine/Xylazine	3.8–10.8 mg/kg 0.7–2.0 mg/kg	IM IM	14%	Poor sedation with Ketamine ≤5.6 mg/kg	Bester, 1988
Arctocephalus gazella Antarctic fur seal	7	Ketamine/Xylazine	5.6–7.8 mg/kg 0.5–1.3 mg/kg	IM dart	0%		Ferreira and Bester, 1999
Arctocephalus phillipi Juan Fernández fur seal	12	Ketamine/Diazepam	2.16–6.76 mg/kg 0.04–0.28 mg/kg	IM	17%	Decreased induction and recovery times than when used IV; variable plane of anesthesia	Sepulveda et al., 1994
Arctocephalus phillipi Juan Fernández fur seal	10	Ketamine/Diazepam	2.16–6.76 mg/kg 0.04–0.28 mg/kg	IV	0%	Deeper immobilization compared with IM	Sepulveda et al., 1994
Arctocephalus pusillus pusillus South African fur seal	27	Ketamine	4.3–7.8 mg/kg	IM dart	19%	Variable anesthesia	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	7	Ketamine/Xylazine	4.2–5.2 mg/kg 0.6–0.9 mg/kg	IM dart IM dart	29%	Xylazine dosage estimated	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	5	Carfentanil/Xylazine	6–18 µg/kg NA	IM dart IM dart	NA	20% of animals given combination with carfentanil died; apnea, muscle convulsions, variable plane of anesthesia	David et al., 1988

Arctocephalus pusillus pusillus South African fur seal	7	Carfentanil/Xylazine/ Azaperone/	6–18 µg/kg NA NA	IM dart IM dart IM dart	na	20% of animals given combinationwith carfentanil died; apnea, muscle convulsions, variable plane of anesthesia	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	2	Carfentanil/Xylazine/ Azaperone/Ketamine	6–18 µg/kg NA NA NA	IM dart IM dart IM dart IM dart	na	20% of animals given combination with carfentanil died; apnea, muscle convulsions, variable plane of anesthesia	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	2	Carfentanil/Xylazine/ Ketamine	6–18 µg/kg NA NA	IM dart IM dart IM dart	na	20% of animals given combination with carfentanil died; apnea, muscle convulsions, variable plane of anesthesia	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	15	Xylazine/Zzaperone	0.57–2.0 mg/kg 0.57–2.0 mg/kg	IM dart IM dart	7%	Sufficient for branding, short immobilization time	David et al., 1988
Arctocephalus pusillus pusillus South African fur seal	2	Droperidol	NA	IM	50%		David et al., 1988
Arctocephalus tropicalis Subantarctic fur seal	58	Ketamine	1.9–2.8 mg/kg	IM	0%	Sufficient for tooth extraction, some tremors noted	Dabin et al., 2002
Arctocephalus tropicalis Subantarctic fur seal	32	Ketamine/Xylazine	3.1–11.4 mg/kg 0.3–1.7 mg/kg	IM dart	13%	Variable anesthesia	Ferreira and Bester, 1999
Arctocephalus tropicalis Subantarctic fur seal	49	Tiletamine/Zolazepam	0.7–1.9 mg/kg	IM	4%	Prolonged recovery; apnea requiring artificial respiration	Dabin et al., 2002
Eumetopias jubatus Steller's sea lion	29	Tiletamine/Zolazepam	1.8–8.1 mg/kg	IM dart	21%	Best results; 1.8–2.5 mg/kg	Loughlin and Spraker, 1989
Eumetopias jubatus Steller's (northern) sea lion	51	Tiletamine/Zolazepam, Isoflurane	1.6–3.3 mg/kg	IM dart IM dart IH	10%		Heath et al., 1996
Otaria byronia South American sea lion	13	Tiletamine/Zolazepam	2.75 mg/kg	IM	0%	Flumazenil 1 mg for every 20–25 mg of tiletamine and zolazepam for reversal	Karesh et al., 1997
Otaria byronia South American sea lion	7	Isoflurane	NA	IH	0%		Karesh et al., 1997
Phocarctos hookeri Hooker's (New Zealand) sea lion	29	Isoflurane	0.8%–4.0%	IH	0%		Gales and Mattlin, 1998
Zalophus californianus California sea lions	4	Detomidine/Ketamine, Isoflurane	40–55 µg/kg 2.0–4.3 mg/kg 1%–5% in oxygen	IM IM IH	0%		Heard and Beusse, 1993
Zalophus californianus California sea lions	60	Tiletamine/Zolazepam	1.7 mg/kg	IM	0%	Apnea	Gage, 1993
Zalophus californianus California sea lion	115	Isoflurane	0.75%–3%	IH	0%		Heath et al., 1997
Zalophus californianus California sea lion	30	Halothane	0.75%–5%	IH	3%		Work et al., 1993

(*Continued*)

Table 41.2. (*Continued*)

Species	*N*	Drug(s)	Dosage	Route	Mortality	Comments	Reference
Zalophus californianus California sea lion	35	Medetomidine/Ketamine	140 µg/kg 2.5 mg/kg	IM IM	0%	Variable anesthesia; reversal with atipamezole	Haulena et al., 2000
Zalophus californianus California sea lion	16	Medetomidine/Ketamine, Isoflurane	140 µg/kg 2.5 mg/kg 1%–5%	IM IM IH	0%	Reversal with atipamezole	Haulena et al., 2000
Zalophus californianus California sea lion	17	Medeomidine, Tiletamine/Zolazepam	70 µg/kg 1 mg/kg	IM IM	6%	Reliable anesthesia; reversal with atipamezole; ataxia and disorientation during recovery in some animals	Haulena and Gulland, 2001
Zalophus californianus California sea lion	22	Medetomidine Tiletamine/Zolazepam, Isoflurane	70 µg/kg 1 mg/kg 1%–5%	IM IM IH	0%	Reliable anesthesia; reversal with atipamezole; ataxia and disorientation during recovery in some animals	Haulena and Gulland, 2001
Zalophus californianus California sea lion	2	Medetomidine/ Midazolam/Butorphanol/ Isoflurane	10–13 µg/kg 0.2–0.26 mg/kg 0.2–0.4 mg/kg 0.5%–2.0%	IM IM IM IH	0%	Two animals anesthetized 13 times Reversal with, atipamezole, flumazenil, naltrexone	Spelman, 2004

Adverse effects observed during recovery included tremors, ataxia, and disorientation. These were less than with zolazepam/tiletamine alone, but occur more often than with medetomidine/ketamine. Animals were premedicated with atropine (0.02 mg/kg IM). One mortality did occur in the study. Consequently, a lower medetomidine dosage (40 µg/kg) in combination with zolazepam/tiletamine is recommended.

Medetomidine/Butorphanol/Midazolam

A combination of medetomidine (10–13 µg/kg), midazolam (0.2–0.26 mg/kg), and butorphanol (0.2–0.4 mg/kg) IM was evaluated in California sea lions (Spelman, 2004). The combination is completely reversible using atipamezole (0.05–0.06 mg/kg IM), flumazenil (0.0002–0.002 mg/kg IM), and naltrexone (0.1 mg/kg IM), respectively. The combination produced safe, light anesthesia in animals that was supplemented with isoflurane for deeper planes of anesthesia. Atropine (0.02 mg/kg IM) was given after injection of the combination.

Inhalant Anesthetics

Inhalant anesthetics, including isoflurane (Heard and Beusse, 1993; Heath et al., 1996, 1997; Gales and Mattlin, 1998; Haulena et al., 2000), sevoflurane, and halothane (Work et al., 1993) have all been used in otariids. The safest anesthesia with the best recovery characteristics has been obtained with isoflurane and sevoflurane. Otariids uptake anesthetic gases very rapidly and efficiently and are readily induced with a mask. Controlled studies on the efficacy of mask induction and maintenance of anesthesia with an inhalant are few for marine mammals. However, California sea lions, New Zealand fur seal (*Arctocephalus forsteri*) bulls, and adult female New Zealand sea lions (*Phocarctos hookeri*) appear to be more rapidly masked to anesthetic depths in comparison with terrestrial species (Heath et al., 1997; Gales and Mattlin, 1998). The use of inhalant anesthetic agents alone appears to be a reliable and safe method of anesthesia in otariids if it is possible to accomplish restraint and masking (Figure 41.1). Premedication and induction with intramuscular drugs facilitates masking and maintenance of anesthesia with an inhalant agent if the animals cannot be masked voluntarily or with physical restraint (Heard and Beusse, 1993; Heath et al., 1996; Haulena et al., 2000; Haulena and Gulland, 2001). Once anesthesia has been attained reversible induction agents may be antagonized.

ANALGESIA

There are very few studies evaluating the use of analgesics in otariids. Use and dosage of analgesic agents has been based on extrapolation from other species and personal experience. Analgesics that have been used with good clinical response in otariids include opiates, non-steroidal anti-inflammatory drugs (NSAIDs), and α_2-agonists. The most commonly used opioid is butorphanol (0.05–0.2 mg/kg PO, IM, or IV q6 hours). NSAIDs used by the author include flunixin meglumine (1 mg/kg IM q24 hours) for up to 3 days, ketoprofen (1 mg/kg IM q 24 hours) for up to 5 days, buffered acetylsalicylic acid (5 mg/kg PO q 24 hours) for up to 5 days, and carprofen (2–4 mg/kg PO q 24 hours) for up to 14 days (Dold, Haulena, and Gulland, 2004). For additional analgesia, butorphanol has been combined with an NSAID. Medetomidine (10–40 µg/kg) has also been used to provide analgesia.

Figure 41.1. *Training a Steller sea lion (Eumetopias jubatus) for voluntary acceptance of a mask for induction with isoflurane at the Vancouver Aquarium. (Photo courtesy of the University of British Columbia Marine Mammal Research Unit.)*

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