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Capture and Immobilization of Free-Ranging Edentates (9-Dec-2002)

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Introduction

The order Edentata, or Xenarthra, is a group of highly specialized mammals that diverged from ancestral species prior to the differentiation of most modern mammalian orders. It is therefore a very ancient group. The term "edentate" means lacking teeth, but only the anteaters are actually toothless. The name "xenarthran" is preferred by some authors, as it refers to a unique type of vertebral articulation that is common to all members of the order [1]. The three living families of the order, which is considered monophyletic, include the *Myrmecophagidae* (anteaters), *Dasypodidae* (armadillos), and *Bradypodidae* (sloths). Twenty-nine species are currently extant, all of which inhabit the New World (Table 1).

Table 1. Names of the twenty-nine extant species in the order Edentata

Group	Common Name	Latin Name
Anteaters	Giant	*Myrmecophaga tridactyla*
	Lesser	*Tamandua mexicana* *T. tetradactyla*
	Silky	*Cyclopes didactylus*
Armadillos	Hairy	*Chaetophractus vellerosus* *C. nationi* *C. villosus*
	Naked-tailed	*Cabassous unicinctus* *C. centralis* *C. chacoensis* *C. tatouay*
	Three-banded	*Tolypeutes tricinctus* *T. matacus*
	Long-nosed	*Dasypus novemcinctus* *D. septemcinctus* *D. hybridus* *D. sabanicola* *D. kappleri* *D. pilosus*
	Fairy	*Chlamyphorus truncatus* *C. retusus*
	Six-banded	*Euphractus sexcinctus*
	Pichi	*Zaedyus pichiy*
	Giant	*Priodontes maximus*
Sloths	Two-toed	*Choloepus didactylus* *C. hoffmanni*
	Three-toed	*Bradypus variegatus* *B. triadactylus* *B. torquatus*

Eight of these species are listed as endangered or vulnerable by the International Union for the Conservation of Nature (IUCN), three are listed under Appendix II of the Convention on International Trade in Endangered Species (CITES), and one is listed under Appendix I of CITES.

Because this clade is so old, its members do have some anatomic peculiarities; some of these are shared by all edentates, while others are specific to certain genera [1]. All edentates have double caudal vena cavae, instead of the single vessel possessed by other mammals. Females have a common urogenital and urinary tract. Males have intra-abdominal testes, which are found between the rectum and urinary bladder. Another trait common to all of these animals is the lack of incisor and canine teeth, although members of the genus *Choloepus* (two-toed sloths) have caniniform premolars. All edentates have low body temperatures (30º to 35ºC) with *Choloepus* spp. having the most variable, from 24º to 33ºC. *Choloepus* spp. are also distinguished by having six, seven, or eight cervical vertebrae, unlike nearly all other mammals, which have seven. Three-toed sloths (*Bradypus* spp.) generally have either eight or nine cervical vertebrae. A unique characteristic of sloths is the tendency for the pelage to harbor algae, which gives a greenish hue to the coat and may provide some camouflage. Baseline blood values for a variety of edentate species are available as reviewed in Wallach and Boever, 1983 [2].

Anteaters

Anteaters currently range from southern Mexico to central South America, although at one time they did occur in Europe. Anteaters are characterized morphologically by an elongated, narrow skull, a long tongue coated with papillae and sticky saliva, and powerful forelimbs with long, curved claws (Fig. 1). The lesser and silky anteaters have prehensile tails. The giant anteater is wholly terrestrial, the lesser anteater species are both terrestrial and arboreal, and the silky anteater is completely arboreal. All anteaters forage on ants, termites, and other insects, although the giant anteater may occasionally take fruit. While the giant and lesser anteaters are active both day and night, the small silky anteater is strictly nocturnal.

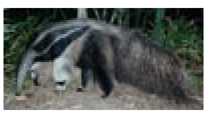

Figure 1. Giant anteater (*Myrmecophaga tridactyla*) in the Kaa-Iya Gran Chaco National Park, Bolivia. Photo courtesy of L Maffei. - To view this image in full size go to the IVIS website at www.ivis.org . -

The giant anteater is considered vulnerable by the IUCN, and is listed on Appendix II of CITES. They are hunted both for food and for trophy purposes, and their range in Central America has been greatly diminished due to habitat loss. Although populations of the other anteater species are not thought to be at high risk of extinction at this time, rapid deforestation and habitat loss in Latin America may soon threaten these animals.

Armadillos

There are twenty species of armadillos, which occur from the southern United States throughout Central and South America. Armadillos are easily recognized by their unique body armor. Like the anteaters, this group varies greatly in body size. The giant armadillo may be as much as 60 kg, while the fairy armadillos are less than 100 grams. The different species vary in their ability to roll up for protection; the three-banded species are the most adept at this. All armadillos are terrestrial, and most are strong diggers, using burrows for protection. They live in a variety of habitats, including semiarid grasslands, brush land, forests, moist lowlands, savannahs, and riparian regions. Insects form the largest part of the diet, although other invertebrates, small rodents, lizards, snakes, carrion, fruits, and plant material are also taken.

Two armadillos are endangered: The giant armadillo and the pink fairy armadillo. The giant armadillo is hunted by people for food; this and habitat loss are the main factors in its decline and continued endangered status. The tiny pink fairy armadillo is threatened by the plowing of fields for agriculture, and predation by domestic dogs. The Bolivian hairy armadillo, *C. nationi*, is considered vulnerable by the IUCN, and is listed on Appendix II of CITES. This species is threatened by human overexploitation for food, and by persecution. These animals are hunted in farm areas, where their burrowing is thought to disturb the soil. Populations of the three-banded armadillo, *T. tricinctus*, are declining, and this species is listed as vulnerable. It inhabits montane tropical rainforest in Brazil, where mining, logging, and hunting are the principle threats to its continued survival.

One species of long-nosed armadillos, the nine-banded (*D. novemcinctus*), is increasing in numbers. This speices has recently expanded its range throughout much of the southern United States. On the other hand, the hairy long-nosed armadillo, *D. pilosus*, is considered vulnerable. Little is known about this animal, and it is restricted geographically to the southwestern mountains of Peru. Habitat loss, and possibly overhunting, are the most likely threats to all armadillo species that are declining.

Sloths
Although sloths were once a highly morphologically diverse and specious group, they are currently limited to two genera and five species, all of which are strictly arboreal and geographically restricted to Central and South America. All sloths are largely herbivorous, eating mainly young leaves, twigs, and buds. The two-toed sloths are thought to be more active and have a more varied diet than the three-toed sloths. Sloths are slow-moving, relatively sedentary animals who spend most of their lives hanging in trees (Fig. 2). They are very awkward on the ground, but they are surprisingly good swimmers and take to the water readily. Sloths typically descend from the trees once or twice a week to defecate. All sloths are mostly nocturnal, probably to avoid predation; felids and raptors are their major predators.

Figure 2. A female *Bradypus variegatus* with infant. Photo by CV Fiorello. - To view this image in full size go to the IVIS website at www.ivis.org . -

Two-toed sloths are considered common in many of the areas where they are found, but both species are listed as "Data Deficient" on the IUCN Red List. The maned sloth, *B. torquatus*, is considered endangered by the IUCN, and *B. variegatus* is listed on Appendix II of CITES. The main threat to these species is habitat destruction.

Immobilization Procedure

General Principles
There are a number of anatomical and physiological characteristics unique to this order that should be considered when immobilizing members of this order (see Introduction). Edentates are heterothermic and thus body temperature fluctuates depending on the environmental temperature, especially during periods when the animal is inactive [3,4]. They are also easily adversely affected by sudden climatic changes [1,3], so clinicians should avoid the capture and immobilization of edentates during cold, inclement weather or during periods of rapid ambient temperature changes. Consequently, body temperature may not be a useful indicator of anesthetic depth. Although these animals do have a more variable body temperature than most mammals, sudden and/or severe changes in temperature should be assessed to determine whether the temperature change is related to anesthesia or needs to be appropriately addressed. Often when immobilizing these animals in a field setting, one does not have the actual body weight. Clinicians frequently underestimate body weights and the animals are often heavier and larger than estimated.

Standard equipment for monitoring the anesthetized edentate should include, at a minimum, a stethescope, pulse oximeter, and thermometer. All handling equipment (i.e., towels, non-disposable gloves, veterinary supplies) should be disinfected prior to use on another animal to prevent the spread of disease.

Dosages in the literature that have been safe and effective in all edentates include ketamine (10 - 20 mg/kg; IM) in combination with diazepam (0.1 mg/kg; IM or IV) or acepromazine (0.1 mg/kg; IM) [4]; ketamine (10 mg/kg; IM) and xylazine (2 mg/kg; IM) [2]; ketamine (1 - 20 mg/kg; IM), which provides a range from sedation to surgical anesthesia [2]; and tiletamine and zolazepam (1.9 - 6.0 mg/kg; IM) [2]. Isoflurane anesthesia using chamber inductions have been used in a variety of captive edentates and might be practical in some field settings. However, it should be noted that armadillos and sloths have the ability to hold their breath for prolonged periods. Supplemental drugs may include atropine (0.04 mg/kg; IM) to control salivation if necessary [4].

Anteaters
<u>Capture Methods</u> - The use of mesh nets to capture anteaters should be avoided because these nets may cause wounds to their mouths, preventing animals from feeding properly following capture [2]. In one study of free-ranging giant anteaters, animals were initially captured using a remote delivery system and 3 cc darts, but the capture method was later switched to chasing animals down and then pinning them with a forked stick and hand injecting [5].

<u>Pre-anesthetic Management</u> - It must be remembered for human safety that anteaters are unpredictable and often aggressive, and can inflict injury using their claws.

Anesthetic Administration - Anesthesia can be administered using a remote drug delivery system [5] or hand syringe injection once the animal is adequately restrained [5,6].

Anesthesia - Anesthetic doses used in the immobilization of captive and free-ranging anteaters are presented in Table 2. Most protocols use a dissociative agent in combination with an alpha-2 agonist, benzodiazepine, or phenothiazine (M Miller, personal communication; JA Teare, personal communication) [5,7-9]. However, some clinicians report successful anesthesia of anteaters using ketamine alone, although catatonia and movement have been associated with this protocol [2,3,8,9]. Etorphine with or without supplemental IV diazepam has also been used in *M. tridactyla* [4]. Although, not practical in most field situations, isoflurane chamber induction with maintenance anesthetic delivered by face mask is an alternative method of anesthesia in this group of animals (JA Teare, personal communication).

In the earlier publication by Gillepsie and Adams [4], it was recommended that xylazine NOT be used due to the potential for abortion during pregnancy and vomition. However, in a recent study [7] there was no vomiting in 17 collared anteaters immobilized with a drug combination that included xylazine. Some clinicians have found that multiple doses of ketamine are necessary even for short procedures, although presumably this could be avoided if a higher initial dose is used.

Table 2 Anesthetic doses used in the immobilization of captive and free-ranging anteaters.

Species / Status	Drug	Dosage (mg/kg)* /Route**	Sample Size***/Comments	Ref.
Myrmecophaga tridactyla free-ranging	Ketamine	9.7	n = 29	[5]
	Xylazine	1.6		
Tamandua tetradactyla free-ranging	Ketamine	20	Especially for manipulations longer than 30 to 40 mins and for minor surgeries.	[7]
	Xylazine	1		
Myrmecophaga tridactyla Captive	Ketamine	1.8 - 2.5	Mild sedation to light anesthesia	Miller[#]
	Medetomidine	0.025 - 0.04		
	Atipamezole	5 X Medetomidine dose		
Myrmecophaga tridactyla Captive	Tiletamine/Zolezapam	3.4	Heavy sedation and prolonged recoveries.	Miller[#]
Myrmecophaga tridactyla Captive	Tiletamine/Zolezapam	2 - 4		Teare[#]
	Ketamine	2		
	Medetomidine	10 - 15 mcg/kg		
	Atipamezole	75 mcg/kg		
Myrmecophaga tridactyla Captive	Tiletamine/Zolezapam	3 - 4		Teare[#]
	Ketamine	1.5 - 2.5		
Myrmecophaga tridactyla Captive	Isoflurane	5% to 2.5%	Induction chamber to facemask	Teare[#]
Myrmecophaga tridactyla n/a ****	Ketamine	5		[9]
	Xylazine	3.5		
	Yohimbine	0.125		
Myrmecophaga tridactyla n/a	Ketamine	11	If use alone beware of catatonia and movement	[9]
Myrmecophaga tridactyla n/a	Ketamine	10 - 20	If use alone beware of catatonia and movement	[2,3]

Species / Status	Drug	Dosage (mg/kg)* /Route**	Sample Size***/Comments	Ref.
Myrmecophaga tridactyla n/a	Ketamine	500 mg/adult female	If use alone beware of catatonia and movement	[8]
Myrmecophaga tridactyla n/a	Ketamine	750 mg/adult male	If use alone beware of catatonia and movement	[8]
Myrmecophaga tridactyla n/a	Ketamine	200 - 400 mg/sick adult	If use alone beware of catatonia and movement	[8]
Myrmecophaga tridactyla n/a	Etorphine	0.8 - 1.2 mg total IM	Immobilization	[4]
	± Diazepam	2.5 - 5.0 mg IV	More relaxation or extend anesthesia	
	± Isoflurane	1 - 2%	More relaxation or extend anesthesia	
	Diprenorphine	1.6 - 2.4 mg total (IV or IM)	Reversal	
Tamandua tetradactyla n/a	Ketamine	10 - 20	If use alone beware of catatonia and movement	[2,3]

* Dosage (mg/kg) - unless otherwise stated
** Route - IM unless otherwise indicated
*** Sample sizes - given if available
**** n/a - Not available
[#]Personal Communication, 2002

Animal Handling and Monitoring - Anteaters should be placed in lateral recumbency and temperature, pulse, and respiration monitored as for other animals. In giant anteaters, the normal range for heart rate is 110 - 160 bpm and respiratory rate is 10 - 30 bpm. Some clinicians use bandaging of front claws to avoid injury to both the animal and humans, especially if using ketamine, although bandaging may be less practical in the field setting.

Post-anesthetic Recovery - Proper handling and monitoring during the recovery period are just as important as during the induction and maintenance periods. It is not uncommon for anesthetic related morbidity and mortality to occur during this period; in fact, most anesthetic complications occur during induction and recovery. Although there are reversal drugs for the zolazepam component of telazol (flumazenil), xylazine (yohimbine), and medetomidine (atipamezole), anteater anesthesia usually cannot be completely reversed with one specific antidote.
It is important to minimize wounds due to the high risk of screwworm (*Cochliomyia hominivorax*) infestation in many regions where anteaters live. Topical betadine and fly strike ointment should be applied to the anesthetic injection site and to any abrasions that occur during the procedure to prevent screwworm infection.

Armadillos

Capture Methods - There are a number of methods for the safe capture of free-ranging armadillos. These include hand capture, hand net, food-baited live traps [10], drift fences attached to live traps [10], leghold traps, dog (*Canis familiaris*) - live trap combination, dog-den excavations, and net pouches attached at den exits [10-14]. The 3-banded armadillo can roll into a complete ball, making the delivery of injectable anesthetics difficult (Fig. 3). Nine-banded and giant armadillos cannot roll into complete balls and thus can be grasped on the side plates (leather gloves may be beneficial) for manipulation and the delivery of injectable antesthetics. Nine-banded armadillos can also be grasped at the tail base and then held around the dorsum of the carpace [15].

Figure 3. A three-banded armadillo (*Tolypeutes matacus*) rolled up in a defensive posture. Photo by SL Deem. - To view this image in full size go to the IVIS website at www.ivis.org . -

Pre-anesthetic Management - Armadillos have a somewhat variable body temperature. For this reason, it is preferable to avoid immobilizing armadillos in the morning or during cold, dark days when they are inactive. Armadillos are unlikely to inflict serious injuries to humans. The 9-banded armadillo is a known carrier of leprosy in some regions so gloves should be worn during handling.

Anesthetic Administration - Armadillos are most commonly injected using a hand syringe and manual restraint. In those species in which injectable agents are difficult to deliver (ie, 3-banded armadillos), the placement of isoflurane soaked cotton balls on the nostrils for a few minutes will relax the animal so that it will unroll (please see below for caution).

Anesthesia - Injectable anesthetic agents are the mainstay for immobilizing free-ranging armadillos. Table 3 includes a review of the literature of both free-ranging and captive armadillos. Similar to anteaters, the most commonly used drug combinations for the anesthesia of armadillos is a dissociative agent with either a benzodiazepine, alpha-2 agonist or phenothiazine [9,13,16,17]. There are two published reports of anesthesia in armadillos using droperidol and fentanyl [17,18] and one with ketamine as the single anesthetic agent [15]. The authors have used isoflurane soaked cotton balls placed near the nares of 3-banded armadillos to provide sufficient relaxation for administering injectable agents. It should be noted however, that in our study of thirteen 3-banded armadillos that received isoflurane with or without some combination of injectable antesthetics, two died with possible liver damage associated with isoflurane use (Deem, unpublished data). Induction chambers are less practical in the field situation, but are routinely used in captivity.

Table 3. Anesthetic agents used in the immobilization of captive and free-ranging armadillos.

Species/Status	Drug	Dosage (mg/kg)* /Route**	Sample Size*** Comments	Ref.
Dasypus novemcinctus Free-ranging	Ketamine	25	n = 40	[17]
	Acepromazine	0.3		
Dasypus novemcinctus Free-ranging	Tiletamine/Zolazepam	3	n = 10	[16]
	Medetomidine	60 mcg/kg		
	Atipamezole	300 mcg/kg		
Dasypus novemcinctus Free ranging	Tiletamine/Zolazepam	4	n = 6	[16]
	Medetomidine	75 mcg/kg		
	Atipamezole	375 mcg/kg		
Dasypus novemcinctus Free ranging	Tiletamine/Zolezapam	8.5	Prolonged recovery (2-3 h); n = 12	[13]
Dasypus novemcinctus Free ranging	Ketamine	40	Prolonged recovery (2-3 h); n = 18	[13]
	Xylazine	1		
Dasypus novemcinctus Free ranging	Ketamine	7.5	Atipamezole antagonism beneficial to decrease recovery; n = 17	[13]
	Medetomidine	75 mcg/kg		
	Atipamezole	380 mcg/kg		
Dasypus novemcinctus Captive	Innovar Vet (Fentanyl Citrate and Droperidol)	0.11 ml/kg	n = 12	[17]
Dasypus novemcinctus Captive	Ketamine	25	If use alone beware of catatonia and movement	[15]

Species/Status	Drug	Dosage (mg/kg)* /Route**	Sample Size*** Comments	Ref.
Dasypus novemcinctus n/a***	Tiletamine/Zolezapam	10		[9]
	Ketamine (supplemental)	10		
Dasypus novemcinctus n/a	Droperidol and Fentanyl	0.20 - 0.25 ml/kg		[18]
Dasypus kappleri Free-ranging	Tiletamine/Zolezapam	8.5	Prolonged recovery (2-3 h); n = 10	[13]
Dasypus kappleri Free-ranging	Ketamine	40	Prolonged recovery (2-3 h); n = 9	[13]
	Xylazine	1		
Dasypus kappleri Free-ranging	Ketamine	7.7	Atipamezole antagonism beneficial to decrease recovery; n = 12	[13]
	Medetomidine	77 mcg/kg		
	Atipamezole	385 mcg/kg		
Tolypeutes matacus Free ranging	Isoflurane	0.25 ml	isoflurane was placed on cotton ball and over nose of animal for induction. Possible mortalities associated with Isoflurane (see text); n = 8	Deem, unpublished data
	Tiletamine/Zolezapam	3		
	Medetomidine	60 mcg/kg		
	Atipamezole	300 mcg/kg		

* Dosage (mg/kg) - unless otherwise stated
** Route - IM unless otherwise indicated
*** Sample sizes - given if available
**** n/a - not available

Animal Handling and Monitoring - Armadillos should be placed in lateral recumbency and temperature, pulse, and respiration monitored as for other animals (Fig. 4). Body temperature in these species is normally low (30 - 35ºC) and should be monitored for significant changes during the immobilization period. Pulse oximeter sensors may be placed on the loose skin of the axillary or inguinal areas (Fig. 5). In our field studies of nine-banded and three-banded armadillos, heart rates varied between 50 - 210 and 104 - 214 bpm, respectively. Intravenous access can be difficult, but sites that have been used successfully include the ventral and lateral tail, medial and lateral saphenous, cephalic, subclavicle, and jugular veins (A Manharth, personal communication, 2002) [15,19].

Figure 4. Lateral recumbency of an anesthetized nine-banded armadillo (*Dasypus novemcinctus*). Photo by CV Fiorello. - To view this image in full size go to the IVIS website at www.ivis.org . -

Figure 5. Pulse oximetry monitoring of an anesthetized three-banded armadillo (*Tolypeutes matacus*). Photo by SL Deem. - To view this image in full size go to the IVIS website at www.ivis.org . -

Post-anesthetic Recovery - Proper handling and monitoring during the recovery period are just as important as during the induction and maintenance periods. It is not uncommon for anesthetic related morbidity and mortality to occur duing this period; in fact, most anesthetic complications occur during induction and recovery. Although there are reversal drugs for the benzodiazepine component of telazol, zolazepam (flumazenil), xylazine (yohimbine), and medetomidine (atipamezole), armadillo anesthesia usually cannot be completely reversed with one specific antidote.

If an armadillo is injured during capture or restraint, it is best to keep the animal separate from others as cannibalism is common within captive groups.

It is important to minimize wounds due to the high risk of screwworm (*Cochliomyia hominivorax*) infestation in many regions where armadillos live. Topical betadine and fly strike ointment can be applied to the anesthetic injection site and to any abrasions that occur during the procedure to prevent screwworm infection.

Sloths

Capture Methods - There are no published reports detailing free-ranging capture methods for this group. In captivity, sloths are usually hand-captured by a keeper, and manually restrained for either an anesthetic injection or mask induction. In some cases, no capture is required; at the Roger Williams Park Zoo, for example, sloths sleep in a bucket, which doubles as an induction chamber. When immobilization is necessary, a cover is placed over the bucket before the sloth exits in the morning and isoflurane is delivered into the bucket (J Martin, personal communication, 2002). Remote drug delivery systems are sometimes used in the field, but more commonly, sloths are captured by hand. Researchers or collectors simply walk through the forest until they encounter an animal. If the sloth is not very high up in the tree, it is often possible to climb up a short distance and grab the animal [20]. Other methods include cutting down the branch, or entire tree, from which the sloth is suspended (D Meritt, personal communication, 2002) [20]. The sloth is then grasped by the scruff of the neck and the hindlimbs and placed face down on the ground (D Meritt, personal communication, 2002). However, when threatened, sloths often curl into a tight ball which can make grasping difficult. Once restrained, they can be put into a nylon mesh bag or net; in which they tend to relax.

The potential danger of sloths, especially two-toed sloths, should not be underestimated. Although sloths are not exactly fierce predators, they are extremely strong and can be aggressive. They can move very quickly and slash accurately with their front claws or draw someone in and deliver a painful bite [4,20]. Three-toed sloths are rarely aggressive and in many cases will allow simple procedures such as physical examinations, injections, wound cleansing, and bandaging with only gentle manual restraint.

Pre-anesthetic Management - Like all edentates, sloths have a somewhat variable body temperature. Because they use sunlight to some extent to increase their temperature, early in the morning they tend to be quite cold and sluggish (D Meritt, personal communication, 2002). This can lead to problems in the field determining the onset of anesthesia after an animal is injected with the anesthetic. For this reason, it is preferable to avoid immobilizing sloths in the morning or during cold, dark days when they are inactive.

Fasting before anesthetic administration is impossible in the field, and only sometimes done in captive situations (A Manharth, personal communication, 2002; S Murray, personal communication, 2002). Fortunately, regurgitation and vomiting are not common (D Meritt, personal communication, 2002; A Manharth, personal communication, 2002) [20].

Anesthetic Administration - In captive situations, sloths are frequently manually restrained and masked down with isoflurane or similar anesthetic gas. Manual restraint and hand injection of an anesthetic agent is also performed in captivity. In the field, sloths are either manually restrained for hand injection, or darted using a remote drug delivery system.

Anesthesia - Anesthetic agents used in the immobilization of captive and free-ranging sloths are presented in Table 4. Isoflurane or other inhalant anesthetics are commonly used with a mask to induce anesthesia in captive sloths. This is not practical for field situations, but fortunately many injectable agents are suitable for these species. Ketamine and tiletamine-zolazepam are the mainstays of injectable anesthetics for sloths now, but before these were available, phencyclidine together with atropine was used with success in the field (D Meritt, personal communication, 2002). Because two-toed sloths (*Choloepus* spp.) are much more common than three-toed sloths (*Bradypus* spp.) in zoos, most of the information available in the literature about anesthetic management concerns the former group.

Ketamine has been used successfully as a single agent for sedation or induction prior to maintenance on inhalant anesthesia [4,21,22]. Tiletamine-zolazepam is used commonly to immobilize captive sloths [20,23], and an intramuscular combination of medetomidine, midazolam, and butorphanol is preferred among some veterinarians who work with sloths in zoo settings (S Murray, personal communication, 2002; R Yates, personal communication, 2002). Atipamezole is sometimes used to reverse the medetomidine, at a dose of 0.05 mg/kg.

Recently, Vogel et al., [20] conducted a large (n = 202) anesthesia study comparing four protocols on free-ranging two-toed sloths. They recommended the use of ketamine with an alpha-2 agonist. This combination provided a smooth induction, good muscle relaxation, and a duration of immobilization adequate for most field procedures (40 to 50 minutes) [20]. While bradycardia typical of alpha-2 agonists did occur, this study did not report any resulting clinical problems. Those animals receiving medetomidine were given atipamezole (0.2 mg/kg) as a reversal agent.

Table 4. Anesthetic agents used in the immobilization of captive and free-ranging sloths.

Species /Status	**Drug**	**Dosage (mg/kg)* /Route****	**Sample Size*** Comments**	**Ref.**
Choloepus didactylus Free-ranging	Ketamine	10	n = 30	[20]
	Acepromazine	0.1		
Choloepus didactylus Free-ranging	Ketamine	10	n = 89	[20]
	Xylazine	1		
Choloepus didactylus Free-ranging	Ketamine	3	n = 46	[20]
	Medetomidine	0.04		
	Atipamezole	0.2		
Choloepus didactylus Captive	Tiletamine/Zolezapam	1.9 - 6.0	n = 14	[23] (Martin#; Murray#)
Choloepus didactylus Captive	Ketamine	8 - 13	n = 3	Murray#
	Midazolam ± Isoflurane	0.22 - 0.42		
Choloepus didactylus Captive	Medetomidine	0.01	n = 5	Murray#
	Midazolam	0.21 - 0.25		
	Butorphanol ± Atipamezole	0.21 - 0.25 0.05		
Choloepus hoffmanni Captive	Tiletamine/Zolazepam	4.4	n/a	[9]
Choloepus hoffmanni Captive	Ketamine	2 - 20	n = 29	[4,22]
Choloepus sp. Captive	Ketamine	4.7 - 5.8	n = 2	[21]

* Dosage (mg/kg) - unless otherwise stated
** Route - IM unless otherwise indicated
*** Sample sizes - given if available
**** n/a - not available
#Personal Communication, 2002

<u>Animal Handling and Monitoring</u> - Respiratory rates are quite variable in sloths, and the normal range is from 10 - 78 bpm [20]. Although data from the Smithsonian National Zoological Park indicate that respiratory rates in the maintenance phases of anesthesia are quite low, from 5 to 12 bpm [20], recorded somewhat higher rates for sloths in field conditions. They found mean initial respiratory rates of 14 bpm, which decreased or increased during anesthesia depending on the protocol used. Ketamine/medetomidine anesthesia was correlated with slower respiratory rates and a decrease during the immobilization. Apnea and irregular respiratory rates were occasionally noted to occur with the ketamine/xylazine combination.

Bradycardia is common when alpha-2 agonists are part of the anesthetic protocol. In general, the initial heart rates recorded by Vogel et al., [20] were just over 60 bpm, and gradually decreased to about 45 bpm. These are roughly similar to heart rates of captive sloths immobilized at the Smithsonian National Zoological Park (S. Murray, personal communication).

Sloths do not regulate their body temperature as closely as most other mammals [4], so body temperature is not a good indicator of anesthetic depth or anesthetic complications. In the study by Vogel et al., [20], rectal temperatures of anesthetized sloths in the field were highly variable, with initial temperatures ranging from 31 to 38.2 ºC.

The measurement of oxygen saturation is a valuable tool in field immobilization. In the field study by Vogel et al., [20], initial oxygen saturation was generally in the upper 80's, and it gradually increased to the mid-90's during anesthesia. Hypoxia was noted in a few animals, but no ill effects were observed clinically. Data from captive sloths at the Smithsonian National Zoological Park show relatively stable levels of oxygen saturation, usually in the mid to upper 90's throughout anesthesia.

Blood may be collected from the cephalic or femoral veins of immobilized or well-restrained sloths [4]. The medial vein in the antecubital region [22] and the dorsal vertebral vein have also been used. It has been reported in the literature to use cardiocentesis or a nail clip [4], although the authors do NOT recommend cardiocentesis. Bush and Gilroy [23] describe a technique that utilizes a Doppler probe to collect blood from the femoral artery; this is unlikely to be practical in a field situation, however.

Post-anesthetic Recovery - Proper handling and monitoring during the recovery period are just as important as during the induction and maintenance periods. It is not uncommon for anesthetic related morbidity and mortality to occur during this period; in fact, most anesthetic complications occur during induction and recovery. Although there are reversal drugs for the zolazepam component of telazol (flumazenil), xylazine (yohimbine), and medetomidine (atipamezole), sloth anesthesia usually cannot be completely reversed with one specific antidote. Sloths recovering from dissociative anesthetics should be monitored for self trauma of the face and mouth using the claws on their forelimbs. Claws can be held against the wrist to help minimize this problem.

It is important to minimize wounds due to the high risk of screwworm (*Cochliomyia hominivorax*) infestation in many regions where sloths live. Topical betadine and fly strike ointment can be applied to the anesthetic injection site, and to any abrasions that occur during the procedure, to prevent screwworm infection.

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