ZOO ANIMAL AND WILDLIFE IMMOBILIZATION AND ANESTHESIA

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48 Rhinoceroses

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THE RHINOCEROTIDAE

Introduction

Like the fabricated creature in Albrecht Dürer's famous lithograph, the rhinoceros has long been a source of mystery, myth and intrigue (Figure 48.1). Part unicorn and part armored beast, the current knowledge of rhinoceros anesthesia likewise represents a melding of pure art and hard science. Today rhinoceros anesthesia is relatively commonplace, yet no less demanding in practice.

The Rhinocerotidae are truly living fossils—a remnant and archaic mammalian family represented by only five extant species in four genera restricted to Africa and Asia. The relic survivors belie a one-time place of dominance among organismic groups with over 150 fossil rhinoceros species discovered by paleontology across four continents (Prothero, 2005). Today, however, four of the five rhinoceroses are critically endangered from poaching and loss of habitat (Foose and van Strien, 1997; Emslie and Brooks, 1999).

Field anesthesia made possible the rhinoceros conservation success stories of the twentieth century (Harthoorn and Lock, 1960; Player, 1972; Meadows, 1996) and remains a critical tool for proactive rhinoceros management programs incorporating translocation, ear-notching, radiotelemetry, microchip implantation, and other techniques designed to secure the conservation of both African and Asian species (Dinerstein, 1990, 2003; Kock, 1990, 1995). Historical and current rhinoceros anesthesia protocols are based on highly effective reversible opioid combinations, yet new anesthesia techniques continue to improve efficacy and safety for both animals and human personnel (Bush, 2005; Bush and Citino, personal communication).

Taxonomy and Evolutionary History

The odd-toed ungulates of the order Perissodactyla include three living families: the rhinos, horses, and tapirs. As the order name denotes, all Perissodactylids bear weight on one (equids) or three (rhinos and tapirs) digits. Rhinos and tapirs are among the most primitive of the world's large mammals and are further grouped into the suborder Ceratomorpha based on a similar ancient body plan. The stout body of the rhinoceros is graviportal or designed for weight bearing with limb modifications to support large mass rather than the long angular limbs of equids in the suborder, Hippomorpha, specialized for speed.

The Perissodactyla enjoyed a period of extraordinary diversity in the Eocene epoch 34 to 55 million years ago before climate change presumably limited species radiation, culminating in extinction of 10 of the 14 perissodactyl families by the end of the Oligocene epoch (Radinsky, 1969). Prehistoric rhinoceroses in particular, as interpreted from fossil evidence, represented a far expanded group of organisms than exist today and included both horned and hornless forms. In fact, rhinoceroses were once the most common large herbivore in North America for most of the last 50 million years (Prothero and Schoch, 2002). An extinct hornless rhinoceros named Paraceratherium (also classified as Indricotherium) is known to science as the largest land mammal that ever lived, measuring over 6 meters at the shoulder and weighing an estimated 20 tons (Prothero and Schoch, 2002; Prothero, 2005).

Biology and Morphology

The Rhinocerotidae are large terrestrial herbivores that have evolved either a browsing (black, Sumatran, Javan) or grazing (white, greater one-horned) strategy to process large quantities of fibrous feeds or simple grasses, respectively. As such, they share bulky elongated skulls, dental patterns largely devoid of canines and incisors (retained to various degree in the Asian species), and prehensile or wide, flat lips in the browsers and grazers, respectively. Like the equids, fermentation takes place in the cecum and colon. The rhino gut is less efficient than that of ruminants since the microfloral protein formed in the hindgut is largely unavailable to the animal. As a result rhinoceroses must eat more, have

Figure 48.1. *The famous 1515 drawing by German artist Albretch Dürer, a rhinoceros portrayal that persisted in Europe for three centuries despite the anatomical errors suggesting a mythical unicorn heritage. (Image copyright The Trustees of the British Museum, London.)*

a relatively fast passage of gut contents, and possess limited time to reabsorb water from the feces. Therefore, rhinoceroses must drink every day or every second day, making it a water-dependent species rarely found more than 15 km from a water source.

Despite their often conspicuous absence in many fossil rhinoceroses, the single horn (*Rhinoceros* sp.) or pair of horns (*Ceratotherium, Diceros,* and *Dicerorhinus* sp.) is certainly the most distinguishing feature of the living Rhinocerotidae, giving name to the group literally as the *Nose-horned beasts* (Prothero and Schoch, 2002). Rhinoceros horns differ from true horns of the Artiodactyla by having no central core of bone. Instead the tubular hair-like keratin filaments are compressed in a linear fashion and set upon a bony protuberance of the skull. Underneath the horns, the skull incorporates extensive nasal bones and sinuses; structures inordinately prone to complications from trauma during capture and translocation.

Rhinoceros skin is thick (several centimeters of primarily collagenous dermis) (Cave and Allbrook, 1958; Shadwick, 1992), with the Asian species sporting subdermal plates and heavy skin folds, making skin anatomy an important consideration for remote drug delivery in the rhinoceros. The greater one-horned rhinoceros of Asia is perhaps best known for the exaggerated armor-like plates or folds first popularized in the famous Dürer woodcut of the Middle Ages (Figure 48.1). However, the epidermis is very thin (1 mm) and heavily keratinized, incorporating extensive vasculature, which may predispose the rhinoceros to pressure necrosis, particularly in calves (Cave and Allbrook, 1958; Gandolf, 2006). Significant body hair is an antiquated trait retained in only one living species, the Sumatran or "hairy rhinoceros," so-called for its shaggy coat of hair (Figure 48.2). Wild *Dicerorhinus* have shorter more bristly coats than their captive relatives, a trait providing protection for the skin from the numerous biting insects that share its environment. Hair, a primordial trait of many fossil rhinoceroses, including the woolly rhino *Coleodonta* and massive one-horned hairy *Elasmotherium,* perhaps no other feature so eloquently links the Sumatran rhino with its long and prosperous past.

RHINOCEROS IMMOBILIZATION AND CAPTURE

Rhinoceros Capture Beginnings

Before widespread application of chemical capture techniques, early African rhino capture operations used ropes and a chase vehicle. Although dangerous to the operator and stressful to the animal, some teams in East Africa became remarkably proficient at this form of capture (McCulloch and Achard, 1969). Chemical capture of rhinoceroses was first attempted with the dissociative anesthetic, phencyclidine, and the curariform muscle relaxant, gallamine triethiodide. In 1960 during Operation Noah, many black rhino (*Diceros bicornis*) were saved from the rising waters of the newly constructed Lake Kariba, bordering Zambia and Zimbabwe, using these novel techniques (Child and Fothergill, 1962; Harthoorn and Lock, 1960; King, 1965; Meadows, 1996). Phencyclidine and gallamine were succeeded by the easily reversible opioids (Condy, 1964), first morphine and dimethylthiambutene followed quickly by

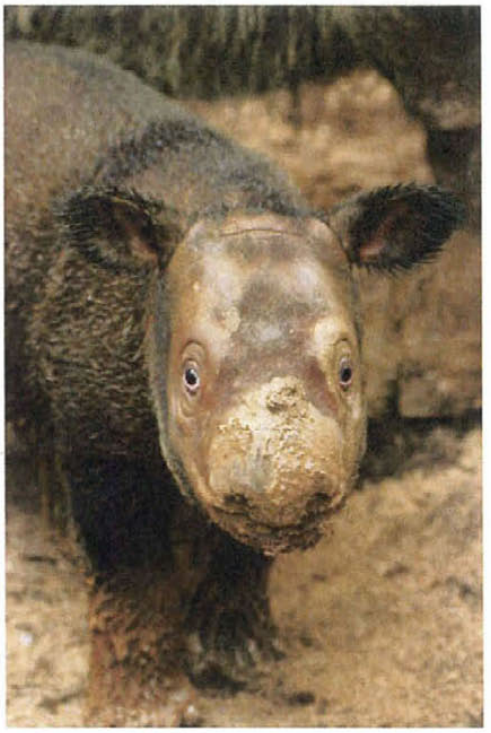


Figure 48.2. *The distinctive hair coat of a young Sumatran rhinoceros: a feature linking the primitive Dicerorhinus genus with its prehistoric past. (Image courtesy Dave Jenike, Cincinnati Zoo & Botanical Garden.)*

the more potent opioids. Over the past 40 years, etorphine HCl (M99) has become the standard opioid for capture of the African and Asian rhinoceroses (Flamand, 1984; Henwood, 1989; Hitchens, 1972; Keep, 1969, 1973; King, 1965, 1969). Fentanyl citrate (Sublimaze), carfentanil citrate (Wildnil), and thiafentanil oxalate (A3080) are useful alternatives (De Vos, 1978; Hofmeyr, 1975). Pioneering investigation by early practitioners such as Toni Harthoorn, Eddie Young, Ian Hofmeyr, Ian Player, and many others provided the foundation on which future rhino chemical capture methods, including the present work, are based (Player, 1972; Harthoorn, 1973; Young, 1973).

Remote Drug Delivery: Equipment and Darting Techniques

An assortment of remote drug delivery equipment is available for rhinoceros capture, including new developments, yet some of the early systems are still in common practice today attesting to their simple and durable design. In captive and boma situations all darting systems can be used, but nylon darts (Daninject or Telinject with 60- mm × 2-mm smooth needles) are preferred as they are quiet and relatively atraumatic. The authors prefer to hand-inject (using appropriate human protective safety measures) or pole-syringe captive rhinos, including animals held in bomas to eliminate the excitement phase associated with projectile darting.

For field capture of rhinoceroses on the ground or from a helicopter a robust and reliable darting method such as the Cap-Chur system is preferable. Dart barrels made of aluminum (Cap-Chur, Powder Springs, GA) or stainless steel (Deon Joubert, RSA) are the most reliable for field use, especially since power settings and impact energy are high, wind or downdrafts from the helicopter can be a problem, and the operator is often forced to shoot through vegetation. The dart needle should be 5 to 6 cm long for an adult rhino. Rhino skin can plug the lumen of a dart needle unless the needle has a relatively thick wall and narrow lumen (Cap-Chur NCL needles) or the tip is bent over (Fauncap dart needles) or the point is sealed and side ports are provided. The needle must have a bead, low barb or small collar about 25 mm from the base to hold the dart in the thick skin (Morkel, 1994).

Proper dart placement is essential to ensure good drug deposition. The dart should be placed perpendicular to the skin for deep intramuscular (IM) injection. (The thick skin of a rhino often makes an angled shot ineffective.) When darting from the helicopter, the muscles of the rump or the upper part of the hind leg offer the best target. In the boma or on foot, any large muscle mass can be used for dart placement although the neck and shoulder are preferable.

Recumbency and Positioning

Recumbency and positioning are critical considerations for safe anesthesia of rhinoceroses whether in a zoological setting or in the wild. Prior to induction in captivity, thick padding or heavy mats should be used to protect recumbent animals from the concrete floors common in these environments. Myositis and neuropathy are serious potential complications. Traditionally, rhinos immobilized in the field are maintained in or moved into sternal recumbency; however, irreversible muscle damage has developed in this position (especially if the rhino goes down on a slope facing upward with the full weight on its hind legs) as a result of occlusion of the blood supply to the limbs. Although uncommon, problems even occur with careful "placement" of the legs in an apparently natural position. With the rhino on its side, blood flow to the limbs is improved and circulation to the muscles allows delivery of oxygen and dissipation of carbon dioxide and heat generated while running. With the animal in lateral recumbency, the legs should be physically "pumped" up and down by hand every 20 minutes to aid circulation. We recommend that all black rhino that have undergone any degree of exertion be placed in lateral recumbency for at least a few minutes. The decision to move white rhinos onto their sides should be based on several factors including the degree of exertion, presence of muscle tremors, and duration of recumbency. White rhinos often experience significant muscle rigidity, paddling, and even convulsions under opioid anesthesia. These effects are exacerbated by lateral positioning but tend to resolve with time.

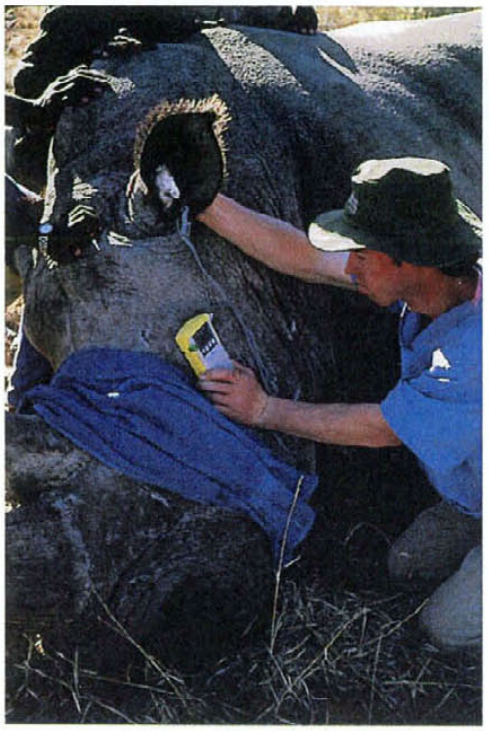


Figure 48.3. *Anesthetic monitoring of free-ranging white rhinoceros (Ceratotherium simum). Note the use of a blindfold, cotton wool plugging ear canals and pulse oximeter attached to the pinnae.*

Therefore, white rhinos should be positioned initially in sternal recumbency until complete relaxation is achieved (Kock, 1995).

Eyes and Ears The eyes of the recumbent rhino should be shielded with a large towel or appropriate-sized blindfold to prevent retinal damage from direct sunlight, dirt accumulation, and corneal abrasion from the environment (Figure 48.3). Foreign material should be washed from the eyes using physiological saline. The ear canals are plugged with cotton wool or a cloth while the rhino is anesthetized, leaving tabs for quick removal. Alternatively, when a large number of rhino are to be immobilized, connect two cloth-covered cotton wool plugs with cord so they remain together. If the rhino is being transported, its ears should remain blocked for the entire trip; however, the blindfold must be removed once the rhino is secure within the crate.

Anesthesia Monitoring

A thorough clinical examination with monitoring of vital functions (respiration, temperature, heart rate, capillary refill time) must be done regularly for the duration of anesthesia. Concentrate on respiration, temperature and heart rate, in that order. These functions are very much dependent on the degree of exertion and excitement before and during induction and must be kept in mind during your evaluation. Careful monitoring is especially important in old, debilitated, very young, and heavily pregnant animals. Check if the entire dart contents were injected, especially if more than one dart was used, as the success of drug delivery may dictate protocols for anesthetic monitoring and antidote administration (Morkel, 1994).

Pulse oximetry provides an indirect measure of oxygen saturation of hemoglobin (SaO_2) and is valuable to help monitor blood oxygenation and pulse in anesthetized rhinoceroses (Figure 48.3). However, it should not be a replacement for thorough patient monitoring. Without simultaneous correlation with arterial blood gases, pulse oximetry is a tool best used to monitor trends in oxygen saturation rather than actual values. Based on lower oxygen affinity of white rhinoceros hemoglobin, it has been suggested that SaO_2 levels (ranging from lows of 40% up to 98%) (Kock, 1995; Atkinson, 2002) in rhinoceroses underestimate true oxygen saturation of hemoglobin when calculations are made using human formulae (Bush, 2004). The sensor clip is attached to the pinnae of the ear after removal of the epidermis by careful scraping with a serrated kitchen knife or on mucosal folds of the penis, vulva, or rectum. Place a cloth over the sensor, as ambient light affects the reading. In animals with excessive muscle rigidity or tremors, as is common in immobilized white rhinos, the sensor may fail to obtain an accurate reading. A rectal probe held against the nasal mucosa works well (must be applied beyond the pigmented area) and has also been used with varying success on the inner surface of the lips, against the gums, and in the rectum or vagina.

Respiration and Oxygen Respiratory depression is perhaps the most significant life-threatening complication encountered during routine anesthesia of rhinoceroses (Heard, 1992; Kock, 1995; Atkinson, 2002; Bush, 2004, 2005; Fahlman, 2004). Large recumbent animals experience cardiopulmonary depression and perfusion-ventilation disparities because of large size and abdominal organs impinging on the diaphragm. Severe respiratory compromise with hypoxemia, hypercapnia, and acidosis is more common with long captive procedures or under field conditions where higher doses of opioids are used to shorten induction times (Heard, 1992; Kock, 1995). Among the African species, these physiological changes are more prevalent in the white than black rhinoceroses (Bush, 2004, 2005).

Respiration is the first and most critical function to be monitored in rhinoceroses under anesthesia. In the field situation it is valuable to have a reliable person who does nothing but watch the respirations, noting rate and depth. Be sure there is a free flow of air in and out of the nostrils and that the blindfold does not restrict airflow. Concentrate on respiratory rate and depth by observing chest movement. When monitoring breaths on a bouncing vehicle, as with immobilized rhino transported on a sledge where it is difficult to watch chest movement, hook a finger in the nostril or hold your

Figure 48.4. *Feeling for the warm expired air facilitates respiratory monitoring of an anesthetized juvenile white rhinoceros (Ceratotherium simum).*

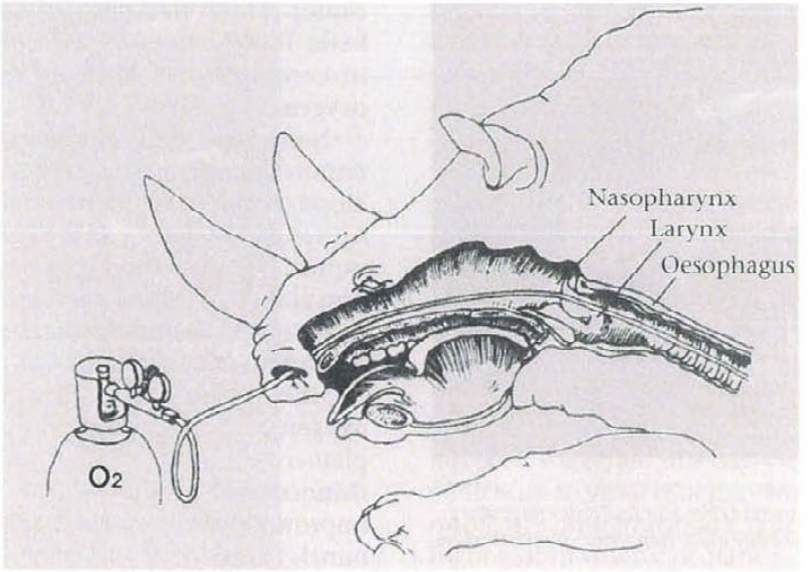
hand close to the nares to feel the warm exhaled air (Figure 48.4). Breathing must be deep and regular. Monitor respiration for at least 30 to 60 seconds to obtain an accurate picture of ventilatory pattern, as an immobilized rhino often gives two or three quick breaths followed by a period of apnea. Respiratory rate is approximately 10 to 15 breaths per minute on induction, going down to 4 to 8 bpm about 10 minutes post-induction when using potent opioids. Observation of venous blood color during venipuncture provides a reliable early indicator of blood oxygenation. Dark red, almost black blood indicates poor oxygenation, whereas a lighter red color is normal and correlates well with mucous membrane color (Morkel, 1994).

If a rhino stops breathing, give nalorphine HBr (Nalline), or consider complete reversal with naltrexone HCl (Trexonil). A painful stimulus often incites the apneic rhino to take a breath. Nalorphine given intravenously (IV) produces a marked and sustained improvement in the quality of respiration (Tables 48.2 and 48.3). Its use has been associated with an approximate 20% increase in the hemoglobin saturation of oxygen (SaO_2) based on pulse oximetry (Kock, 1995). Although widely reported to improve oxygenation (Rogers, 1993a,b), however, recent investigation suggests that nalorphine produces negligible change to oxygen partial pressures (PaO_2) in anesthetized rhinoceroses (Bush, 2004; Fahlman, personal communication). Since black rhino stand up readily with very small volumes of nalorphine, we recommend a dose of 5 mg nalorphine for adult black rhino and 25 to 30 mg for white rhino under field conditions. It is very safe to use nalorphine in white rhino, as this species rarely rises without stimulation, and even if arousal does occur adult animals are relatively harmless in a semi-narcotized state. IV doxapram HCl (Dopram; black rhino 200 mg, white rhino 400 mg) provides a smaller, transient improvement in respiratory rate and depth. Use doxapram with caution in white rhinos as it causes central nervous system excitation and exacerbates muscle tremors; effects are best noted if used in conjunction with nalorphine and supplemental oxygen.

Nasal or tracheal insufflation of oxygen (O_2; 15–30 L/minute) can produce a rapid and significant increase in blood oxygen saturation in immobilized rhinoceroses. Although it did not correct systemic acidosis or hypercapnia, O_2 insufflation substantially improved oxygenation and anesthetic safety (Bush, 2004; Fahlman, 2004). A variety of factors influence pulmonary blood gas exchange, including dose of anesthetic drug, position of the rhino during immobilization, body temperature, oxygen delivery, and size of the animal. Oxygen supplementation at the flow rates commonly used for rhinoceroses appears to produce a more profound improvement of patient oxygenation (PaO_2 108–194 mmHg) in subadult African rhinos compared to adults, perhaps indicating greater ventilation-perfusion mismatch with larger body size (Fahlman, 2004, unpublished data).

A control valve and flow meter are attached to the O_2 bottle and oxygen is administered via a flexible silicon or rubber nasogastric tube (smooth and round edges to prevent damage to nasal mucosa), measuring 2 m long and 9 to 14 mm inside diameter (Figure 48.5). Concurrent monitoring of the respiratory rate and depth, and blood oxygenation remains essential. A low dose of nalorphine increases the rate and depth of respiration and improves the efficacy of oxygen supplementation. Oxygen supplementation is one of the few practical solutions to enhance pulmonary gas exchange in immobilized rhinoceroses and if used wisely one bottle is sufficient for many animals. Therefore, we recommend immediate intranasal or tracheal insufflation of oxygen in all recumbent rhinoceroses. Within a few minutes vital statistics provide information about respiratory function and in most situations all physiological parameters are satisfactory. A small percentage of animals, however, develop a physiological crisis in which oxygen supplementation is critical. Aluminum oxygen bottles are now available that are small and lightweight, making them convenient for helicopter use.

Body Temperature Body temperature is an important parameter and the best indicator of the degree of exertion endured by the rhinoceros before induction. Keep in mind that for every 1° increase in body temperature above normal, there is a marked increase in oxygen consumption. A rhino's body temperature varies slightly during the day as the ambient temperature changes. Black rhino immobilized without excessive exertion have a rectal temperature of between 36°C and 39°C. Young rhino tend to have a higher body temperature than adult rhino after running a comparable distance. An animal with a body temperature of greater than 39°C must be liberally soaked with cool water. Although



Source: Bush, M.R., Raath, J.P., Grobler, D. and L. Klein. 2004. Severe hypoxaemia in field-anaesthetised white rhinoceros (*Ceratotherium simum*) and effects of using tracheal insufflation of oxygen. J. S. Afr. Vet. Assoc. 75(2): 79-84

Figure 48.5. *Illustration of the nasogastric tube tracheal insufflation technique for oxygen delivery in a recumbent white rhinoceros (Ceratotherium simum) under field conditions. (Adapted from Bush MR, Raath JP, Grobler D, et al. Severe hypoxaemia in field-anaesthetised white rhinoceros (Ceratotherium simum) and effects of using tracheal insufflation of oxygen. J South Afr Vet Assoc 2004;75:79–84. (Illustration courtesy the South African Veterinary Association.)*

drenching with water is important, it does not have a dramatic effect in lowering the body temperature, as there is considerable thermal inertia in such a large mammal. It helps to fan the rhino with branches or cloths after the animal has been wetted with water. Holding leafy branches over the rhino to provide shade can help lower the temperature, but it is important that people do not crowd around an immobilized rhino and prevent air movement. A rhino with a body temperature over 39°C must be processed quickly, whereas a temperature of greater than 41°C mandates immediate delivery of the antidote.

Pulse and Blood Pressure Heart rate is best obtained using a stethoscope, whereas the pulse is readily palpable under the base of the tail (caudal artery) or on the inside of the ear (medial auricular artery). Subjectively evaluate pulse quality and compare with the pulse oximeter reading. It is often quite easy to visualize heart compressions by watching the chest wall or by feel with a hand placed over the cardiac window. The heart rate is usually 55 to 80 beats per minute (bpm), although it is higher in rhino that have undergone marked exertion, especially young animals (as much as 140 bpm). Cardiovascular function and peripheral perfusion are assessed by capillary refill time (CRT) and is measured by blanching the rhino's gum for several seconds and then releasing. The observed delay or refill time should not exceed 2 seconds (Morkel, 1994).

Hypertension is prevalent under etorphine anesthesia in black and white rhinoceroses (LeBlanc, 1987; Heard, 1992; Hattingh, 1994). One report in white rhino anesthetized under field conditions noted an apparent reduction in blood pressures when azaperone tartrate (Stressnil) replaced fentanyl in etorphine-based combinations, an effect observed despite the higher dose of etorphine used in the cocktails containing azaperone (Hattingh, 1994). These conclusions, however, appear dubious as fentanyl is itself a potent opioid with prominent hypertensive effects; possessing an activity approximately 1/15 that of etorphine, fentanyl was likely a confounding factor in the study. Although no definitive mechanism has been identified, increased sympathetic nervous system action, peripheral vasoconstriction, and hypoxemia are proposed factors in etorphine-induced hypertension in both rhinos and equids (Daniel and Ling, 1972; Heard, 1992). Opioid related hypoxemia might induce sympathetic system stimulation and hypertension; once hypoxia resolves the sympathetic response and associated hypertension disappear.

RHINOCEROS ANESTHESIA IN CAPTIVITY

Guidelines for Anesthesia of Captive Rhinoceroses

The large size of the rhinoceros belies an unexpected sensitivity to the opioid class of pharmacological agents (Raath, 1999). Surprisingly, the same dose of carfentanil citrate used to immobilize a 20-kg blackbuck (*Antelope cervicapra*) would also fully immobilize a 2,200-kg white rhinoceros (*Ceratotherium simum*), making the rhino over 100 times more opioid sensitive per unit mass than

the average artiodactylid. This inordinate sensitivity of the rhinoceros family to the opioid class—although responsible for the undesirable changes observed in cardiopulmonary function—also makes it possible to adapt less potent mixed agonist-antagonist opioid agents into anesthetic protocols for both captive and wild rhinoceroses (Radcliffe, 2000a; Walzer, 2000; Bush, 2005).

Planning for anesthetic events should include preparation of the subject and environment in which these variables can be controlled. Depending on the purpose for anesthesia it is generally desirable to fast the animal for 12 to 48 hours prior to anesthesia (Radcliffe, 2000b). However, fasting is certainly not essential, as evidenced by the many successful field operations in which capture of wild rhinoceroses is conducted in the absence of pre-anesthetic fasting. Water access should be denied for at least 12 hours and all water sources removed from the environment prior to drug delivery, as regurgitation has been noted in white rhinoceroses (Raath, 1999). Both passive and active regurgitation of stomach contents are known. The latter are very rare but quite spectacular. Passive regurgitation is common in immobilized rhinos, presumably secondary to drug- or hypoxemia-induced relaxation of the cardiac sphincter. Because of the risks of regurgitation and inhalation pneumonia, great care must be taken with positioning of the head and nostrils, especially with animals in lateral recumbency.

Habitual patterns of behavior are important aspects of captive rhinoceros husbandry, facilitating close medical management. Anesthesia techniques should be adapted as part of these conditioning protocols. Regular visits by animal health staff to rhino barns or bomas for acclimatization to the sights, sounds, and smells of the veterinary profession help limit the stress of such procedures. In boma situations it is helpful to learn the nature of each animal, including its likes and dislikes while also listening carefully to the keeper in charge of caring for the animal.

African Rhinoceros Captive Anesthetic Regimens

White Rhinoceros (*Ceratotherium simum*) The adult white rhinoceros is large and generally placid in captivity. Anesthesia with potent opioids is often associated with marked hypermetria, muscle rigidity, trembling, head shaking, and limb paddling (Figure 48.6). These effects are undesirable and can be prevented by preanesthetic administration of the sedative or tranquilizer component of the cocktail. In captive animals, initial dosing with IM azaperone 20 to 30 minutes prior to induction with etorphine helps preclude muscle spasms and rigidity. With wild rhinos, positioning in sternal recumbency until complete relaxation is achieved was deemed important in field practice (Kock, 1995). New techniques offer alternatives for long or painful procedures by incorporation of the highly specific α_2-agonist medetomidine HCl (Domitor) into routine rhino immobilization protocols (Citino, personal communication).

Figure 48.6. *Typical induction posture in adult white rhinoceros (Ceratotherium simum) under the effects of etorphine, illustrating characteristic head elevation, raised hackney action of forelimbs, and muscle rigidity. (Image courtesy Rolfe Radcliffe, Living Fossil Productions.)*

Mixtures of etorphine or carfentanil combined with a sedative are standard agents for anesthesia of the captive white rhinoceros (Table 48.1). Doses ranging from 0.8 mg to 3 mg of etorphine and 1.2 mg carfentanil are common, with supplemental opioids given IM or IV to extend anesthesia (Heard, 1992; Walzer, 2000). Following immobilizing doses of etorphine or carfentanil, other agents provide additional muscle relaxation and a deeper plane of anesthesia, including IV propofol, guaifenesin, ketamine, midazolam, and α_2-agents (Klein, 1997; Zuba and Burns, 1998; Walzer, 2000; Kock, 2006). Muscle relaxation is critical for deep ventilation and to counteract the associated risk of oxygen depletion from muscle tremors and hyperthermia inherent with use of potent opioids. Lower opioid doses are indicated in zoo-conditioned animals, yet the potent opioids are still associated with significant cardiopulmonary changes, especially as procedure length increases (Heard, 1992). One captive white rhinoceros immobilized with etorphine remained hypoxemic despite maintenance of inhalation anesthesia using intermittent partial pressure ventilation (Cornick-Seahorn, 1995). Hypertension is common, whereas hypoventilation, pulmonary shunting, and atelectasis induce hypoxia and hypercapnia (Heard, 1992; Bush, 2004).

Butorphanol tartrate (Torbugesic) combinations are replacing use of more potent opioids for rhinoceros anesthesia in many zoological settings as safe and reliable anesthetic planes can be achieved for most procedures, including surgery (Radcliffe, 2000a,b,c). Although not appropriate for all applications (i.e., fractious, nonconditioned animals or those with access to large areas) butorphanol combinations are highly effective. The author has used a mixture of butorphanol and azaperone

Table 48.1. Suggested doses for chemical restraint of adult captive rhinoceros producing anesthetic planes from sedation to recumbency.

Rhino Species	Standing Sedation Protocol	Reversal	Reference Comments	Recumbency Protocol	Reversal	Reference Comments
White rhinoceros	50–70 mg Butorphanol (BT) + 100 mg Azaperone IM hand-injection plus constant rate infusion (CRI)	Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Radcliffe, 2000a, 2000b; use CRI in long procedures	70–120 mg Butorphanol + 100–160 mg Azaperone IM hand injection	Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Radcliffe, 2000a; supplemental IV dosing or CRI
	120–150 mg Butorphanol + 5–7 mg Medetomidine (MED) IM dart (Give 1–2 mg Nalorphine IV to keep standing)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino, unpubl. data	120–150 mg Butorphanol + 5–7 mg Medetomidine (IM dart; recumbency ~20 minutes)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino, unpubl. data; improved analgesia for surgery
	0.8–1.5 mg Etorphine (M99) IM dart	Naltrexone at 40 mg per mg M99	Portas, 2004	2–3 mg Etorphine + 20–40 mg Azaperone IM dart	Naltrexone at 40 mg per mg M99	Portas, 2004
				1.2 mg Carfentanil IM dart	Naltrexone at 100 mg per mg M99	Portas, 2004
Black rhinoceros	25–50 mg Butorphanol IV or IM hand injection	Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Radcliffe, 2000c and unpubl. data; use for sub-adults and crating	1–1.5 mg Etorphine + 100 mg Azaperone IM hand injection	Naltrexone at 50 mg per mg M99 1/2IV 1/2IM	Radcliffe, unpubl. data; lower M99 doses with hand injection
	1.5–2 mg Etorphine + 2–3 mg Medetomidine (Give 1–2 mg Nalorphine IV to keep standing) IM dart	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino, unpubl. data	1.5–2 mg Etorphine + 2–3 mg Medetomidine (IM dart; recumbency ~15 minutes)	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino, unpubl. data; enhanced analgesia for dental surgery
	2–2.5 mg Etorphine + 10 mg Detomidine (DET) + 15 mg Butorphanol IM dart	Naltrexone at 40 mg per mg M99 Atipamezole at 5 mg per mg DET	Portas, 2004	2.5–3 mg Etorphine + 60 mg Azaperone IM dart	Naltrexone at 20–40 mg per mg M99	Portas, 2004
Greater one-horned rhinoceros	100 mg Butorphanol + 100 mg Azaperone IM hand injection	Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Radcliffe and Lung, unpubl. data	3.5–3.8 mg Etorphine + 14 mg Detomidine + 400 mg Ketamine IM pole syringe	150–300 mg Naltrexone 1/2IV 1/2IM No reversal DET	Atkinson, 2002
Sumatran rhinoceros	25–40 mg Butorphanol IM hand injection	Same	Radcliffe, 2002; use Azaperone in longer procedures	30–50 mg Butorphanol + 50–60 mg Azaperone IM hand injection	Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Radcliffe, 2002; higher doses for recumbency
				1 mg Etorphine + 60 mg Azaperone IM hand injection	Naltrexone at 50 mg per mg M99 1/2IV 1/2IM	Radcliffe, unpubl. data; Azaperone 20 min. < M99 or suppl. Midazolam

From Citino, unpublished data; Radcliffe, unpublished data; Atkinson MW, Bruce H, Gandolf AR, et al. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. *J Zoo Wildl Med* 2002;33:157–162; Portas TJ. A review of drugs and techniques used for sedation and anaesthesia in captive rhinoceros species. *Aust Vet J* 2004;82:542–549.

for standing sedation and recumbent anesthesia in all four rhinoceros species maintained in captivity (white, black, greater one-horned, and Sumatran) with safe, predictable results (Radcliffe, 2000a,c, unpublished data). Butorphanol doses for white rhino range from 50 to 120 mg for an adult and 10 to 20 mg for a calf or juvenile animal, whereas azaperone doses range from 100 to 160 mg for an adult with supplemental doses given up to a maximum of 300 mg (Tables 48.1 and 48.4). IV butorphanol supplementation is highly effective at inducing recumbency in white rhinos after initial drug delivery, if needed and desirable. IV azaperone has been associated with adverse extrapyramidal reactions in the horse and white rhino and should be avoided (Radcliffe, 2000a).

Black Rhinoceros (*Diceros bicornis*) Black rhino appear predisposed to excitation during induction with etorphine, especially with remote drug delivery in zoological environments (Portas, 2004). Using appropriate human safety practices, the stress of darting can be avoided by hand injection, thereby alleviating much of the undesirable excitatory phase black rhinos experience while also significantly reducing the total dose of opioid agents required (Radcliffe, unpublished data; Table 48.1). In bomas, to limit the "undesirable excitatory phase" great care should be taken to minimize the number of people and unusual objects close to the boma. Noise and movement should be avoided and once recumbent the rhino's eyes should be covered and ears blocked as soon as possible. Significant induction risks include lacerations, limb and foot injuries, head trauma, damage to nasal sinuses, horn avulsion, and even death. With careful animal conditioning and procedure planning the risks of induction excitation are easily minimized. Likewise, antagonism of narcotic anesthesia in the black rhino is characterized by rapid and aggressive recoveries mandating extra care; never stand in front of a narcotized rhino, as arousal is often sudden and unpredictable (Kock, 2006).

As in the other rhinoceros species, potent opioids (primarily etorphine) have historically been used for anesthesia of captive black rhinoceroses with predictable results (Portas, 2004). Zoo conditioned animals require much lower doses of etorphine (1–1.5 mg) than their wild counterparts, especially when administered by hand injection or pole syringe (Table 48.1). Butorphanol alone or in combination with azaperone or detomidine HCl (Dormosedan) has also been used in the black rhino, although its use is primarily limited to subadult animals, crating and translocation procedures or well-conditioned animals, because black rhino are easily excitable and may override drug effects (Radcliffe, 2000c, unpublished data). If butorphanol is chosen as the primary opioid agent, expect light planes of anesthesia and the need for frequent redosing. A more thorough discussion of mixed agonist-antagonist opioid cocktails and newer α_2-agents for use in both captive and field immobilization protocols for the African rhinoceros can be found in the New Techniques section of this chapter and Tables 48.1 and 48.2.

Asian Rhinoceros Captive Anesthetic Regimens

Indian or Greater One-Horned Rhinoceros (*Rhinoceros unicornis*) Despite the common occurrence of Indian rhinos in zoological parks and a propensity for foot problems necessitating chronic care, few published accounts of anesthesia in captive greater one-horned rhinoceroses exist (Atkinson, 2002; Portas, 2004). One report combined injectable and inhalation anesthesia in a female *Rhinoceros unicornis* for ovariohysterectomy using etorphine and isoflurane in oxygen. The 7-hour-long anesthesia (much of it in dorsal recumbency) was considered effective despite the animal succumbing to post-surgical complications (Klein, 1997). The most complete summary of captive anesthesia in this species, however, describes serial opioid-based anesthesia to facilitate long-term medical foot care in one animal. A combination of etorphine-detomidine (3–3.6 mg and 10–14 mg IM, respectively) was given by projectile dart or etorphine-detomidine-ketamine (3.5–3.8 mg, 14 mg and 400 mg IM, respectively) administered by pole-syringe (Atkinson, 2002). Use of the pole-syringe for drug delivery was preferred, because darting was limited by a small target area among the peculiar anatomic neck folds and by drug selection for small dart volumes. Although both drug combinations proved efficacious, subjective assessment suggested that the etorphine-detomidine-ketamine protocol produced more rapid induction, lowered the need for supplemental ketamine, and shortened reversal times (Atkinson, 2002).

The author has used butorphanol and azaperone (100 mg of each drug mixed in a syringe and given by hand injection) to induce standing sedation in the Indian rhinoceros (Radcliffe and Lung, unpublished data). A combination of butorphanol and detomidine (120 and 80 mg, respectively) produced sternal recumbency for surgical repair of a rectal prolapse (Bertelsen, 2004). As in the white rhinoceros, these protocols provide adequate muscle relaxation, sedation, and analgesia while being completely reversible with the pure opioid antagonists naltrexone or naloxone hydrochloride (Narcan). Naltrexone is preferred unless short immobilization intervals are anticipated since renarcotization is common using naloxone alone; naloxone provides complete reversal for a short duration (approximately 30–60 minutes) and is only suggested if repeat procedures are planned for the same day (Gandolf, 2000; Radcliffe, 2000a; Bertelsen, 2004; Portas, 2004).

Javan or Lesser One-Horned Rhinoceros (*Rhinoceros sondaicus*) *Rhinoceros sondaicus* is the only rhino not presently represented by captive specimens and was only extraordinarily displayed in zoological gardens

during the seventeenth, eighteenth, and nineteenth centuries (Rookmaaker, 1998). Although historical records indicate that at least 22 Javan rhino were captured between 1647 and 1939, only four survived long enough to reach zoo exhibits in Adelaide, Calcutta, and London (Rookmaaker, 1998). The entire surviving wild population of Javan rhinoceroses can be found in Ujong Kulon National Park in West Java (*n* approx 50) and Cat Tein National Park in Vietnam (*n* approx five). No accounts of Javan rhinoceros anesthesia exist, but techniques presumably would be analogous to approaches used for the Sumatran rhinoceros (*Dicerorhinus sumatrensis*) or greater Asian one-horned rhinoceros (*Rhinoceros unicornis*), with size difference being a notable exception.

Sumatran Rhinoceros (*Dicerorhinus sumatrensis*) Few reports of Sumatran rhinoceros anesthesia exist because captive specimens are rare. Etorphine (0.98–1.23 mg or 1 mg) combined with acepromazine (PromAce; 4–5 mg) or azaperone (60 mg) has been used to anesthetize captive Sumatran rhinos (Portas, 2004; Radcliffe, unpublished data). As with the African species, muscle rigidity and cardiopulmonary depression are common with use of these potent opioid agents and pre-anesthetic administration of a tranquilizer is prudent to limit muscle tremors and improve respiratory function. Total azaperone doses should be kept to 100 mg or less, as ataxia has been noted upon recovery with higher doses in this species. Butorphanol has been combined with detomidine for standing sedation (Citino and Morris, personal communication), whereas the author routinely uses a mixture of butorphanol and azaperone for standing sedation and full recumbent procedures (Table 48.1) (Radcliffe, 2002).

As with the African species, butorphanol combinations are preferred in captive Sumatran rhinoceroses to preclude the adverse cardiopulmonary changes associated with use of more potent opioids. For adult animals, butorphanol at a dose of 60 to 80 µg/kg with azaperone at 80 to 100 µg/kg and a range of 30 to 50 mg and 50 to 60 mg butorphanol and azaperone, respectively, is recommended, with higher butorphanol doses being used on occasion to produce recumbency. Antagonism of the butorphanol effects is accomplished with naltrexone at a dose of 2.5 times the induction dose of butorphanol (Table 48.1) (Radcliffe, 2002). Other tranquilizers may be used in place of azaperone such as the α_2-agonists, but care should be exercised as hypoxemia has been reported with use of these sedatives. Local anesthetics may facilitate invasive procedures; however, use of more potent narcotics such as etorphine or other pharmacological agents such as ketamine may be indicated to induce surgical anesthesia.

New Captive Anesthesia Techniques

Although much has been learned about rhinoceros anesthesia, limitations still hinder safe and reliable procedures for these large mammals, especially where prolonged recumbency or surgery is required (Heard, 1992; Klein, 1997). Standing restraint where possible using mixed agonist-antagonists shows promise (Radcliffe, 2000a,b). For the black rhinoceros, in which potent opioids are still often preferred over mixed agonists, challenges include marked respiratory depression, inadequate muscle relaxation, need for frequent re-dosing, and incomplete analgesia in painful procedures. An exciting new development in captive anesthesia of African rhinoceroses incorporates the potent α_2-agonist medetomidine with etorphine or butorphanol (Citino, unpublished data). Because α_2-agonists exacerbate respiratory depression and hypotension, contribute to dehydration, and alter thermoregulatory mechanisms, they must be used with caution in rhinos of unknown health status, especially old and debilitated animals. However, under captive conditions in which the health of an animal is known and a specific type of anesthesia is desirable, α_2 agents are effective supplements.

For the black rhino, medetomidine (2–3 mg representing 2–2.9 µg/kg IM dose; 20 mg/ml solution) is combined with etorphine (1.5–2 mg representing 1.5–1.7 µg/kg IM dose) (Citino, unpublished data) and given by dart. The investigators were able to begin safe animal manipulations at approximately 9 minutes, with full recumbency achieved in 15 minutes. This combination facilitated very painful procedures, including molar extractions and foot surgery, with the additional supplement of an IV guaifenesin-ketamine drip (1 g ketamine in 1 L 5% GGE solution) to enhance peripheral analgesia. Relaxation was excellent with easy access to the oral cavity for dental surgery. Physiological parameters were considered normal with concomitant nasal oxygen insufflation. Recovery from anesthesia was smooth and rapid, with no evidence of resedation or renarcotization using naltrexone at 30 mg per mg etorphine and atipamezole HCl (Antisedan) at 5 mg per mg medetomidine.

For white rhino, in which butorphanol has proved so effective in captive settings, the same investigator is using medetomidine (5–7 mg IM) and butorphanol (120–150 mg IM) to provide enhanced muscle relaxation and analgesia properties (Citino, unpublished data). The animals can be manipulated within approximately 11 minutes of IM drug delivery, with full recumbency in 20 minutes. The addition of medetomidine into these protocols has significantly improved analgesia properties for such painful ophthalmic procedures as eye enucleation and conjunctival flap surgery. As with the black rhinoceros, a 5% guaifenesin-ketamine drip was deemed useful for long procedures and to enhance peripheral analgesia. Antagonism was complete using naltrexone at 1 mg per mg butorphanol and atipamezole at 5 mg per mg medetomidine (Citino, unpublished data).

RHINOCEROS ANESTHESIA IN THE WILD

Guidelines for Anesthesia of Wild Rhinoceroses Field anesthesia of Asian and African rhinoceroses is often undertaken to facilitate urgent conservation actions such as dehorning, ear-notching, microchip application, radio-collaring, and horn transmitter implantation or translocation to safe areas (Dinerstein, 1990; Kock, 1990, 1995). Ideally rhino capture operations should be conducted when temperatures are lower than 25°C, usually in the early morning or late afternoon. Darting free-ranging rhino when ambient temperatures are high increases the risk of elevated body temperatures and associated physiological stress. If working in the late afternoon, do not dart a rhino unless there is enough daylight remaining (leave an hour or more to process the animal and deal with potential problems) (Rogers, 1993a,b). If a rhino has run hard enough for its skin to become dark with sweat, the rhino's body temperature often exceeds 39°C. Such an animal should not be darted or if it has already been darted, it must be drenched with water and processed quickly. If the temperature of an immobilized rhino rises above 41°C, give the antidote and release the animal immediately.

With good dart placement, recumbency should follow within 3 to 6 minutes post-drug delivery (Morkel, 1989; Kock, 2006). Induction is usually quicker in young rhino and longer in large bulls and heavily pregnant cows. If there are no signs at about 6 minutes, the rhino should be darted again. Induction times of less than 3 minutes may indicate an overdose and it is important to get to such an animal quickly so that the respiration and other vital functions can be monitored and oxygen, nalorphine or doxapram given, if necessary. In protocols incorporating thiafentanil, rapid inductions are expected and less of a concern. IV opioid use should be avoided because of risks of apnea; however, if necessary give the opioid slowly while keeping a close eye on respiration. For the same reason caution must be exercised when giving midazolam or α_2-agonists by the IV route.

As a rhino becomes affected by etorphine, its pace shortens, the forelegs are lifted higher in a classic "Hackney gait," and the head is elevated (Figure 48.6). The rhino then starts to blunder through bushes and slows down before going into lateral or sternal recumbency. In rough terrain rhino have a tendency to run downhill once they are heavily narcotized and may easily injure themselves by running into a gully or water source. With a quick induction, rhino tend to go down in sternal recumbency. Occasionally the forelegs collapse first and the hindquarters remain elevated. In this situation the full weight of the abdominal organs press on the diaphragm and respiration can be severely compromised, especially in heavily pregnant females, in which the weight of the fetus adds additional pressure. Such animals must be immediately pushed onto their side. Usually a rhino is fully recumbent on arrival; however, if it is still on its feet the brake rope can be placed around one of its rear legs, the blindfold over its eyes, and cotton wool in its ears. On arrival at an immobilized rhino make a quick estimate of its age and body condition. Older or debilitated rhino need special care. Be sure that nothing impedes respiration or is pushing against the rhino's belly, chest, or nostrils. Also be sure the rhino is not facing downhill with pressure against the diaphragm. Field personnel must work quickly, whereas the rhino is recumbent and it helps to prepare a prioritized checklist before beginning each rhino capture (see Practical Strategies for Rhinoceros Field Anesthesia).

African Rhinoceros Wild Anesthetic Regimens

White Rhinoceros (*Ceratotherium simum*) With the high doses of opioids used to speed induction under field conditions, the safe anesthesia of wild white rhinos represents one of the most challenging branches of rhinoceros anesthesia (Table 48.2). Hypoxia, hypercapnia, hypertension, tachycardia, and acidosis are common physiological abnormalities reported in anesthetized white rhino (Heard, 1992; Bush, 2004). Numerous techniques have been developed to help alleviate the significant opioid-induced cardiopulmonary depression in African rhinos. These include use of partial agonist-antagonist agents such as nalorphine to reverse the μ-regulated opioid respiratory depression, respiratory stimulants such as doxapram, nasal or tracheal insufflation of oxygen, and incorporation of mixed agonist-antagonist agents into more potent opioid-based protocols to influence receptor effects (Kock, 1995; Radcliffe, 2000a; Bush, 2004, 2005; Fahlman, 2004).

Opioid doses for field anesthesia of adult white rhinoceroses range from 3 to 4.5 mg of etorphine plus 40 to 60 mg azaperone or 10 to 20 mg detomidine (Table 48.2) (Rogers, 1993a; Kock, 1995; Bush, 2004). Hyaluronidase (Hylase; 5,000 IU) is often incorporated into darting protocols for rhinoceroses to shorten induction time (Morkel, 1989). With hyaluronidase white rhinos stopped moving 2 to 3 minutes sooner but often remained standing (Kock, 1995). Fentanyl has been incorporated into drug cocktails for white rhinoceroses but is rarely used today. The rule of thumb for replacing the opioid component mixes etorphine and fentanyl at the following ratio: one-third etorphine with two-thirds equivalent dose of fentanyl (1 mg of etorphine is equipotent to 15 mg of fentanyl) (Rogers, 1993a). Historically, the parasympatholytic agent, hyoscine, was combined with opioids to induce pupillary dilation and "temporary blindness" to ease handling (Player, 1972; Rogers, 1993a); however, its use is no longer widely accepted because of undesirable side effects and is now considered obsolete (Kock, 1995; Raath, 1999).

An extensive study of white rhinoceros anesthesia incorporating several drug protocols and 141 immobilizations over a 2-year period was conducted in

Table 48.2. Suggested doses for chemical restraint of adult wild rhinoceros including supplemental agents used for respiratory support.

	Immobilization			Respiratory Support	
Rhino Species	Protocol	Reversal	Reference Comments	Protocol	Reference Comments
White rhinoceros	2–3.5 mg etorphine (M99) + 40–90 mg butorphanol (BT) + 25–50 mg midazolam (MDZ) + IM dart	Naltrexone at 40 mg per mg M99 IV (full reversal) OR 2–2.5 mg diprenorphine (M50:50) per mg M99 IV (reverses M99, but not BT)	Bush, 2005; NEW technique reduces respiratory depression, hypoxia, muscle rigidity and tremors, but with relatively long induction times	Produces immobile rhino in ~10 minutes and crating WITHOUT partial opioid reversal In case of inadvertent overdose or cardiopulmonary suppression give Diprenorphine to reverse the M99 while preserving the sedative effects of the BT	Bush, pers. com. and unpubl. data; reverse part or all of opioid effects based on desired outcome
	3–4.5 mg etorphine + 100–250 mg azaperone (40–60 mg if crating) (replace azaperone with 10–20 mg detomidine if no transport) IM dart	For crate reversal: 6–12 mg (2–3 × M99 dose IV) M50:50 plus 1–2 mg naltrexone IV if pushing	Kock, 1995, 2006; Rogers, 1993a; still considered standard translocation protocol	ALL WHITE RHINO: Mandatory 1 mg M50:50 plus 10 mg nalorphine IV OR 20–30 mg nalorphine IV OR	Kock, 1995, 2006; Morkel, unpubl. data
	Consider 5–20 mg midazolam slowly IV for muscle relaxation	For field/boma reversal: naltrexone at 40 mg per mg M99 IV (full reversal)	Morkel, unpubl. data Kock, 1995	20–40 mg nalbuphine IV	
Black rhinoceros	4 mg etorphine + 40–60 mg azaperone (replace azaperone with 100 mg xylazine or 10 mg detomidine)+ 5000 IU hyaluronidase IM dart	For crate reversal: 10–20 mg nalorphine per mg M99 plus 1–2 mg M50:50 IV For field/boma reversal: naltrexone at 40 mg per mg M99 IV (full reversal)	Morkel, 1989; Higher M99 doses for Diceros bicornis bicornis Kock, 1992, 2006; hyaluronidase is always recommended	NOTE: DO NOT use the white rhino respiratory protocol in black rhino as it will cause arousal INSTEAD: 5 mg nalorphine IV; titrate to effect IMPORTANT to have animal lateral and "pump" legs every 20 minutes	Morkel, unpubl. data Kock, 2006
	2–2.5 mg thiafentanil (A3080) + 2–2.5 mg etorphine IM dart	Same	Rogers, 1993b		
Greater one-horned rhinoceros	2–2.5 mg etorphine + 10 mg acepromazine IM dart OR 0.7 mg carfentanil (CF)	Diprenorphine at 2.5 mg per mg M99 IV Naltrexone at 100 mg per mg carfentanil IV	Dinerstein, 1990; One sudden arousal noted; Induction times longer for breeding males	Cardiopulmonary depression not reported; 6–10 breaths per min Surround target rhino with 10–15 trained elephants	Dinerstein, 1990
Sumatran rhinoceros	2 mg etorphine + 80 mg azaperone + 5000 IU hyaluronidase IM dart OR Use M99: BT: MDZ 80 mg butorphanol + 80 mg azaperone IM dart	Naltrexone at 50 mg per mg M99 1/2IV 1/2IM Naltrexone at 2.5 mg per mg BT 1/2IV 1/2IM	Author suggestion (extrapolated from captive animals) Use for compromised animal in snare	Treat like black rhino; muscle rigidity and tremors common Use 5 mg midazolam to relax Use 5 mg nalorphine for partial reversal of respiratory depression If rhino is approachable give 25–40 mg butorphanol IV rather than via dart	Radcliffe, 2002 and unpubl. data Radcliffe, 2002 and unpubl. data

From Bush, Citino, Grobler, unpublished data; Morkel, unpublished data; Radcliffe, unpublished data; Kock MD, Meltzer D, Burroughs R, eds. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Harare, Zimbabwe: Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services, 2006; Rogers PS. 1993a. Chemical capture of the white rhinoceros (*Ceratotherium simum*) OR 1993b. Chemical capture of the black rhinoceros (*Diceros bicornis*). In: McKenzie AA, ed. *The Capture and Care Manual*. Pretoria, South Africa: Wildlife Decision Support Service and South African Veterinary Foundation.

Zimbabwe to enable dehorning operations (Kock, 1995). Initial immobilization mortality was quite high at 7% and was primarily attributed to hypoxemia and cardiovascular collapse. Subsequent captures used lower opioid immobilizing doses and simultaneously incorporated routine use of nalorphine (10–20 mg) or nalbuphine HCl (Nubain; 20–40 mg) to help improve respiration, especially in longer procedures in which mortality was most prevalent. Of the various drug combinations tested (etorphine alone and in combination with fentanyl, xylazine, or detomidine), the etorphine-detomidine combination was considered superior because it was empirically judged as smoother and more rapid (no statistical significance). Pulse rates and creatinine phosphokinase (CPK) levels were significantly lower with the etorphine-detomidine combination, suggesting improved cardiac function and less muscle damage, respectively (Kock, 1995). Good muscle relaxation was observed without the rigidity and paddling common with use of potent opioids in the white rhino. The ratio of etorphine to tranquilizer was critical and dose dependent, likely reflecting differences in drug pharmacology and onset of action.

An effective alternative for mitigating muscle rigidity in wild white rhinos is use of midazolam (Morkel, unpublished data) (Table 48.2). Since immobilized white rhinoceroses are often first encountered in a standing position with a rigid body stance (Figure 48.6), IV midazolam at 5 to 20 mg is effective in inducing good muscle relaxation and recumbency. The Zimbabwe workers noted that even small incremental increases in etorphine in the initial immobilizing dose or re-dosing with etorphine resulted in poorer muscle relaxation and increased head shaking, jerking, and limb paddling (Kock, 1995). Midazolam has excellent muscle relaxation properties and is a useful adjunct in these situations.

Black Rhinoceros (*Diceros bicornis*) Capture related stress appears to be a significant factor in field immobilization of the black rhinoceros resulting in morbidity and mortality in the post-capture period. Rapid immobilization using high opioid doses in combination with hyaluronidase is the single most critical factor in reducing stress during black rhino capture (Morkel, 1989; Kock, 1992). Furthermore, higher etorphine doses and use of hyaluronidase were associated with significantly shorter induction times, lower body temperatures, shorter distances moved, and reduced muscle damage as evidenced by lower CPK and lactate dehydrogenase levels (Kock, 1992). Although two accounts list 3 mg etorphine as a standard opioid immobilizing dose for wild black rhinoceroses (Kock, 1990; Rogers, 1993b), subsequent study suggests that 3 mg of etorphine is inadequate because of prolonged induction periods and associated capture stress (Kock, 1992). Based on review of published material and considerable author experience, 4 mg of etorphine is recommended as a good standard dose for an adult black rhino bull or cow in good condition (Table 48.2) (Morkel, 1989, unpublished data). A scaled-down opioid dose should be used in young animals or those in poor body condition; however, in all other circumstances a low dose of etorphine is contraindicated for free-range capture of the black rhinoceros (Kock, 2006). Azaperone is incorporated into etorphine-based African rhino immobilization protocols at 100 to 250 mg total dose (Table 48.2) with a lower dose of 40–60 mg azaperone used for crating. Xylazine or Detomidine alone (100 or 10 mg per adult, respectively) can be substituted for azaperone based on individual preference.

There appears to be a slight disparity in the opioid dose required for immobilization of the various subspecies of black rhinoceroses. The desert subspecies (*Diceros bicornis bicornis*) needs a slightly higher dose than the other subspecies. Although 5 or even 6 mg etorphine may be necessary for an adult *D. b. bicornis* bull in good condition, 4 mg is usually more than adequate for a comparable response in animals of the *D. b. minor* or *D. b. michaeli* subspecies. Not only is there variation among subspecies, but there also appears to be some difference among individuals. Therefore, the capture veterinarian must be aware of these vagaries in dose response and be prepared to respond if an animal reacts unfavorably.

Asian Rhinoceros Wild Anesthetic Regimens

Indian or Greater One-Horned Rhinoceros (*Rhinoceros unicornis*) Techniques for field anesthesia of the greater one-horned rhinoceros were developed to meet research needs, including the elucidation of basic ecology, genetics, social organization, and dispersal biology (Dinerstein, 1990, 2003). Furthermore, translocation programs are proving essential for reaching long-term population management goals for *Rhinoceros unicornis* in India and Nepal. Capture of wild greater one-horned rhinoceroses is usually conducted from atop trained elephants to facilitate finding and darting of rhinos among the dense tall-grass habitats in the flood plain grasslands and riverine forests where these rhinos live. In addition to providing an elevated platform, elephants (10–15 animals) are used to surround the target rhino before and after darting to facilitate observation of the rhino during induction and prevent escape into open water (Dinerstein, 1990).

Adult greater one-horned rhinoceroses weigh an estimated 2,000 kg, with males slightly larger than females. Dinerstein and colleagues immobilized 39 animals (representing 51 events) using a combination of etorphine and acepromazine (2–2.5 mg and 10 mg, respectively) delivered via remote IM injection either in the shoulder or rump using Cap-Chur darts with 5-cm needles (Table 48.2) (Dinerstein, 1990). One adult female was immobilized with carfentanil (0.7 mg) and all animals were successfully reversed in the field using diprenorphine

HCl (M50:50). Induction times were found to be significantly longer in breeding versus non-breeding males with the former group rarely moving far from the site of darting. A large disparity in induction times was noted across all age and sex groups presumably related to variable drug delivery from dart placement among the thick skin folds characteristic of the species (Dinerstein, 1990).

Javan or Lesser One-Horned Rhinoceros (*Rhinoceros sondaicus*) There have been no published reports describing field capture or anesthesia of the Javan rhinoceros. As with the Sumatran rhino, pitfall trap methodologies rather than stockade-style traps are recommended for capture of lesser one-horned rhinoceroses in the rainforest environment, provided the risks of flooding can be controlled (Nardelli, 1987b; Sadmoko, 1990). Field anesthesia is also possible—especially where animals are pushed out of the forest by human activities—and would be based on extrapolation of the best available information from the other Asian species.

Sumatran Rhinoceros (*Dicerorhinus sumatrensis*) Several intensive operations have been conducted to capture wild Sumatran rhinos using corral or stockade traps with little or no success (Abdullah, 1987; Sadmoko, 1990). In one instance an adult female Sumatran rhino suffered severe head injuries and acute death following capture in a stockade trap from apparent panic-related self-trauma (Nardelli, 1987a). Planned capture of wild Sumatran rhinoceroses in the forests of Southeast Asia is most effectively accomplished by use of the pitfall trap. Effective pitfall traps measured 10' × 4' × 8' (length × width × depth) and incorporated strong plywood walls to preclude landslides and a breakaway false ceiling that drops the animal into the excavated pit beneath (Figure 48.7). Site selection favoring heavily used rhino trails

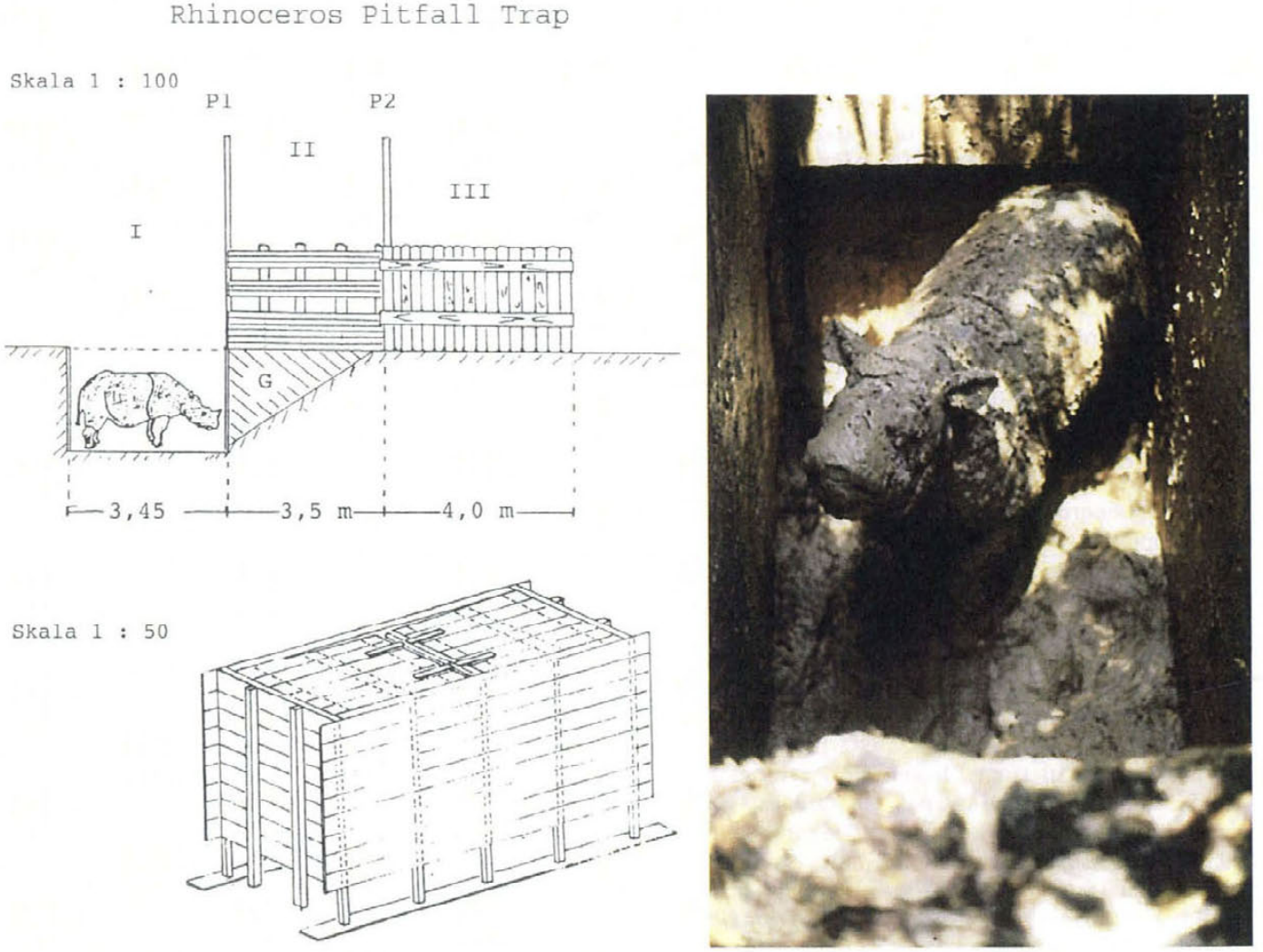


Figure 48.7. *Diagrammatic sketch of pitfall capture method used in Indonesia's Riau Province for capture of wild Sumatran rhinoceros (Dicerorhinus sumatrensis). (Sketches adapted from Sadmoko AS. Study on capture techniques of Sumatran rhinoceros (Dicerorhinus sumatrensis, Fischer, 1814) in Riau Province. Bogor, Indonesia: Department of Forest Resource Conservation, Faculty of Forestry; Bogor Agricultural University (IPB), 1990. (Image courtesy Mohd Khan bin Momin Khan, Malaysia Department of Wildlife and National Parks.)*

was considered the single most important criteria for success or failure of the pitfall trap (Abdullah, 1987). Nevertheless, pitfalls suffer from significant problems. In many Sumatran rhino areas poor drainage results in flooding of the pit despite careful preventive measures. Interference from non-target species is also a common hazard; tapir, elephants, cattle, and even human beings have fallen into pitfall traps despite sign boards erected for the benefit of man (Abdullah, 1987).

Because of the dense nature of the rainforest environment and rare sighting of individual rhinos therein, routine chemical capture techniques developed for Asian and African rhinoceroses are too dangerous as an animal may be lost in the darting process. Increasingly, however, animals are being pushed from the jungle by human encroachment and once beyond the protective boundary of the forest are immediately threatened. In these circumstances, pitfall capture methods are not feasible and chemical capture techniques are indicated. Therefore, the capture process for an at-risk Sumatran rhinoceros found wandering within a Southeast Asian village or otherwise outside a protected area should be approached with careful planning of some urgency. Once the appropriate National Park, Rhino Protection Unit (RPU) and Sanctuary staff have been contacted, the following stepwise approach to capture and translocation is suggested (Radcliffe, 2002).

Guidelines for Capture of Displaced Sumatran rhinoceros

Secure Immediate Area In the event a wild Sumatran rhinoceros is found wandering outside a protected area the first priority would be to secure the area from villagers and would-be poachers to prevent the animal from being shot or otherwise harmed before capture or relocation of the rhino is possible.

Determine Relocation Strategy If possible, a small core group of decision makers should be formed to make immediate assessment of the risks and benefits of rhino relocation. If the rhino were unharmed and close to a protected area (less than 10 km), then it would be desirable to move the rhino back into the forest. If the animal were injured or otherwise in need of medical attention or far (greater than 10 km) from the forest, a decision should be made to capture the animal.

Make a Plan for Rhino Capture Considering the high risks associated with capture by the "chase to exhaustion" method (i.e., rhino is captured following an extensive stressful chase without the use of routine chemical capture methods) (Figure 48.8), this approach should only be attempted as a last resort. The following are suggested guidelines and methodology for capture of at-risk Sumatran rhinos outside a protected area.

Capture Method One: Field Capture Using Chemical Restraint If a trained capture team is available (i.e., within 5 hours travel time) then it may be wise to have the RPU ranger staff carefully monitor and secure the rhino and surrounding area from a distance without pushing the animal to run as they await the capture team. A rapid induction and recumbency will be essential for safe capture of a tropical ungulate species such as the Sumatran rhino that may risk drowning or suffer from capture myopathy.

For field anesthesia of the Sumatran rhinoceros a combination of equal parts butorphanol and azaperone (80 mg each) is recommended for simplicity and its inherent safety for both rhino and people alike (Table 48.2) (Radcliffe, 2002). However, if a well-trained veterinary capture team is available, then use of more potent opioids such as etorphine combined with azaperone and hyaluronidase (2 mg, 80 mg, and 5,000 IU, respectively) or the newer etorphine-butorphanol-midazolam protocols may be considered depending on the situation. If the rhino is already compromised from a chase or is restrained by a snare, the use of the safer butorphanol protocol is preferable to the potent opioids (Table 48.2). The butorphanol-azaperone combination may require confinement within a temporary boma or some additional restraint via a body or head rope to facilitate crating in healthy animals.

Capture Method Two: Field Capture by Erecting Temporary Boma The Sumatran rhinoceros is perhaps the only species of rhino that can be captured by human physical restraint alone, albeit after much chasing and associated capture stress. Therefore, if a trained capture team is not available and the rhino is in immediate peril, physical capture can be a feasible option. To begin, follow the animal from a safe distance and without excessive chasing until the rhino is located within an area where it is resting and approachable (i.e., in water or other suitable location) (Figure 48.8). Large rolls of shade cloth or tarp are then carefully erected without disturbance to form a temporary boma surrounding the rhino that facilitates sedation, crating and transport. Once the animal is restricted within the confines of the "artificial boma," hand injection or pole syringe delivery of the butorphanol-azaperone combination would facilitate safe crating and transfer. The boma method is not likely to eliminate the long chase periods and accompanying stress, but it was effective in the recent capture and relocation of a young adult Sumatran rhinoceros in Indonesia (Figure 48.9).

RHINOCEROS CRATING AND TRANSPORT

Walking a Rhino

If a crate cannot be placed directly in front of the anesthetized animal, the rhino can be "walked" a distance and guided into the crate (Figure 48.10 and Table 48.3). When the rhino becomes recumbent, the blindfold, cotton wool, head rope, and brake rope are applied. Four to

Figure 48.8. *Like the other rhinoceros species, the Sumatran rhinoceros (Dicerorhinus sumatrensis) is prone to capture myopathy. Here a wild "hairy" rhino is restrained with a girth rope in hopes of moving the animal into a temporary boma. Hyperthermia is best avoided by limiting chase periods and liberal application of water. (Image courtesy Sugiyo, Wildlife Conservation Society, Indonesia Program.)*

Figure 48.9. *A wild Sumatran rhinoceros (Dicerorhinus sumatrensis) undergoing "hand translocation" without use of chemical restraint after displacement from a protected forest reserve in Indonesia. Although this animal survived significant capture-related morbidity, chemical capture techniques are preferred if trained staff are readily available. (Image courtesy Chandra Putra, Way Kambas National Park, Sumatra.)*

Figure 48.10. *"Walking" an etorphine-immobilized white rhinoceros (Ceratotherium simum) using ropes and trained personnel to guide and stabilize the narcotized animal.*

six people are stationed on each rope, two people on each shoulder, one person to the side leading the team, and two people walking in front of the rhino, clearing obstacles in its path. The rhino is given small incremental doses of IV nalorphine depending on species; doses vary, but as little as 20 to 25 mg may be needed in total (Table 48.3) (Kock, 2006). After each dose, wait a few minutes and check the rhino's response to the prodder or by squirting water in the ear. If there is no response, give another dose of nalorphine. Once the rhino stands, it should begin to stagger forward and can then be readily guided with the head rope and by the people on the sides. If the rhino moves too fast, go slowly with the head rope and pull the brake rope to slow the moving rhino. Particularly with young and female individuals it is important to slow the rhino as it approaches the crate so it does not traumatize itself upon entrance.

When walking a black rhino into a crate, very encouraging results have also been achieved using diazepam just prior to arousal of the recumbent rhino (Table 48.3). First administer 10 to 15 mg IV diazepam and then wake the rhino with nalorphine at 10 to 20 mg per mg etorphine plus 1 to 2 mg diprenorphine IV. This crating protocol results in a rhino that is well tranquilized for about 8 hours but does not push or traumatize itself in the crate. In our experience, animals off-loaded into a boma were calm for up to 18 hours. Crating captive rhinos has historically involved low doses of etorphine (<1mg). As with anesthesia, however, mixed agonist-antagonist are effective alternatives. Butorphanol and azaperone are mixed with hyaluronidase in a syringe and given by hand injection. The rhino is lured into the crate with food or by waving a white flag. If the rhino does not enter, staff can place a blindfold and head rope and pull the rhino into the crate (Table 48.3).

Table 48.3. Suggested opioid reversal protocols for walking, crate loading and transport of adult African rhinoceros.

	Opioid Use for Crating and Translocation of African Rhinoceros	
Method	Reversal Drug or Opioid	Technique for Crating or Translocation
Nalorphine walking and crating Method	**White Rhino** *Walking*: 1 mg diprenorphine (M50:50) plus 40 mg IV nalorphine. Give further incremental 10–20 mg nalorphine IV up to 75 mg (Kock 2006) *Crating*: 2–3 × etorphine dose of mg diprenorphine (M50:50) IV (add 1–2 naltrexone if pushing in crate) **Black Rhino** *Walking*: Start with 10 mg nalorphine IV. Give incremental 5 mg doses every 5–10 min up to 20–40 mg (Kock, 2006) *Crating*: 10–20 mg nalorphine per mg etorphine plus 1–2 mg M50:50 IV (Morkel) AVOID myopathy in crating process by ensuring rhino does not squat in crate; use prodder plus repeat 1–2 mg M50:50 doses	Blindfold rhino; cover eyes completely and position rhino's head close to or inside crate door: black rhino very important to have head in door; white rhino not critical; keep ears plugged until crated or leave in for transport Place *head rope*; use 20-m soft nylon behind posterior horn with knot on side of head passing rope end through hole in crate Place *break rope* on rear leg just below hock; use 8 meter nylon rope Position 6–8 people on head rope and 3–4 people on break rope Reverse; WAIT 50 seconds (M50:50) or 90–120 seconds (nalorphine); use prodder or water in ear to stimulate rhino *Walk* rhino into crate by pulling on head rope, slow rhino with break rope or go slowly with head rope; guide rhino by ground personnel; slide and secure pipes in crate (most crates have horizontal pipes, some only have the doors) If black rhino pushes in crate give 1 mg naloxone or 0.6–1.2 mg diprenorphine IV; if white rhino pushes give 1 to 2 mg naltrexone IV; use prodder on forehead (not on backside) as needed to stop pushing
Diazepam: nalorphine crating method	**White and Black Rhino** 10–15 mg Diazepam IV ten minutes before "waking" rhino with reversal protocol Use standard crating methodologies above for white and black rhinos after giving diazepam	Give diazepam to recumbent rhino and wait 10 minutes Use same crating procedure as above using diprenorphine alone (white rhino) or nalorphine combined with diprenorphine (black rhino) This protocol eliminates much of the pushing often observed in the crate following diprenorphine or nalorphine reversal procedures Diazepam provides good sedation for ~8 hours especially in white rhino
Butorphanol crating method	*White Rhino*: 50 mg Butorphanol IV (Radcliffe, 2000a, unpubl. data). *Crating Nervous White Rhino:* Male, 120 mg Butorphanol plus 160 mg Azaperone IM Female, 100 mg Butorphanol plus 160 mg Azaperone IM Combine with 2000 IU Hyaluronidase and hand inject (Radcliffe and Lopez, unpubl, data). *Black Rhino*: 25–50 mg Butorphanol IV at time of crating for conditioned animals	Butorphanol is a useful agent for crating and transport of crate-conditioned rhinos in zoological settings: combine with Azaperone as needed Butorphanol provides excellent sedation without concerns of excessive head pressing in crate and occlusion of nostrils in corner. Upon first signs of sedation, keepers entice rhino into crate using food or by waving a white flag; if rhino does not enter crate, apply blindfold and head rope; "walk" rhino into crate by pulling on head rope through front of crate; use prodder on backside. No reversal required for butorphanol once rhino is in crate Use low doses etorphine to crate rhinos from boma; combine with azaperone in black rhino; wave white flag on pole to lure rhino into crate
Etorphine: azaperone boma crating	**White and Black Rhino:** *Boma-Crating*: 0.7–1.2 mg etorphine IM (Kock, 2006) OR 0.3 mg and 0.5 mg etorphine for black and white rhino, respectively without need to reverse *Crate Sedation*: 0.05–0.15 mg Etorphine IM plus 100–200 mg azaperone IM *Transport*: 25–150 mg Zuclopenthixol acetate IM or 5–10 mg diazepam IV	Etorphine is the ONLY AGENT to calm an excitable rhino inside a crate For crate sedation: Nalorphine sedation wears off ~5 hrs post-crating; thereafter give etorphine every 2 hours for duration of trip If rhino is not excitable, give Azaperone up to 200 mg azaperone per 6 hours NOTE: Etorphine is not effective within 3–4 hours of diprenorphine use and 12–24 hours following reversal with naltrexone Avoid perphenazine in white rhino (if going to boma) as it causes anorexia; low dose OK if going straight to field

From Morkel, unpublished data; Radcliffe, unpublished data; Kock MD, Meltzer D, Burroughs R, eds. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Harare, Zimbabwe: Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services, 2006; Rogers PS. Chemical capture of the white rhinoceros (*Ceratotherium simum*), 1993a, OR Chemical capture of the black rhinoceros (*Diceros bicornis*), 1993b. In: McKenzie AA, ed. *The Capture and Care Manual*. Pretoria, South Africa: Wildlife Decision Support Service and South African Veterinary Foundation.

Tranquilization during Transport

All black rhinoceroses require tranquilization during transport (even most crate-conditioned animals) to preclude excessive struggles and associated trauma (Table 48.3). Other rhino species tolerate transport better than black rhinos, but still often benefit from some sedation. The veterinarian must always travel with the rhino and be prepared to give additional sedatives or even narcotics if needed. It is imperative that the veterinarian anticipates the animal's tranquilization needs as waiting until the rhino is alert and bouncing around, will risk unnecessary trauma. Additionally, a cool animal is generally more relaxed than an overheated one.

Rhinos settle into the rhythm of transport quite well after just a few hours. However, as the short-acting tranquilizers begin to wear off, the animal may become very excited if suddenly startled (i.e., from stopping, offloading, etc.). It helps to re-dose the rhino with tranquilizers while the vehicle is in motion or alternatively, stop, inject, and start moving again immediately. In most instances, hand injection is the best method to deliver additional tranquilizer. Insert a 20-gauge 1.5-in. needle into the lateral muscles of the neck while avoiding the nuchal region. Once the rhino has settled, attach the syringe and inject. A pole syringe can also be used, but beware of coring, as the rhino's skin may block the needle. Keep in mind that an IM injection takes 5 to 10 minutes for first effect. For a faster response, an IV injection into the ear vein is sometimes possible, although care must be taken to avoid the dangerous area around the animal's head and horn. Resting by the rhinoceros during transport can be beneficial or a potential problem, depending on the rhino's position and duration of recumbency. If the rhino lies down while the vehicle is moving, the rocking and bouncing action of the truck helps to facilitate limb circulation. Beware, however, if the rhino lies down for a long period (greater than 30 minutes) in a stationary vehicle unless you are very comfortable with its position. Rhinos heavily sedated with opioids often struggle to work out a way to lie down; however, if they manage to do it once, they will lie down more easily thereafter.

Short-acting tranquilizers such as azaperone, xylazine, and detomidine are useful agents to produce a calming effect in rhinos during transport. Azaperone is the tranquilizing agent of choice at 100 to 250 mg per adult and can be repeated every 6 hours as needed (Rogers, 1993a,b; Kock, 2006). A 40-mg per ml azaperone solution is a convenient preparation and mixes well with etorphine for IM administration to a fractious crated rhino. Examine azaperone solutions carefully before use, as they often crystallize under field conditions. The administration of opioids, either alone or in combination with IM azaperone or diazepam, is the only effective way to preclude an excited black rhino from traumatizing itself inside a crate (Table 48.3). Etorphine and azaperone (0.05–0.15 mg and 100–200 mg, respectively) are delivered by pole syringe with sedation achieved in 5 to 10 minutes for durations of 2 hours or more. Long-acting tranquilizers can help to calm an animal, however, are inadequate by themselves to sedate an excited animal during transport. Zuclopenthixol acetate (Clopixol Acuphase; 25–150 mg per adult rhino up to 400 mg) takes about an hour to provide sedative effects after administration, whereas perphenazine enanthate (Trilafon; 200–400 mg per adult) takes about 12 hours for first noticeable effects (Swan, 1993; Kock, 2006) (Table 48.3). Perphenazine works well for the translocation of black rhinoceroses, whereas caution should be exercised in white rhino, as its use has been implicated in anorexia (Portas, 2004; Kock, 2006).

ALTERNATIVE RHINOCEROS ANESTHESIA TECHNIQUES

Antidote Choice

Antidotes are best given IV in rhino, as response after IM injection is often slow and incomplete. Arousal of rhinoceros cow-calf combinations is the one instance in which IM injection of the antidote is preferred so the pair awakens slowly and has time to join together without dashing off in opposite directions. Following IV antidote administration, the rhino will stand up within 45 to 80 seconds. Rapid recoveries occur using the pure antagonists (naltrexone and naloxone) while longer recoveries are observed with the partial agonist-antagonists (diprenorphine and nalorphine). White rhino reversed with diprenorphine or nalorphine often require prodding. Response to the antidote is first noted at 40 seconds as an increase in the depth and rate of respiration and movement of the ears and eyes. Rhinoceroses get to their feet quickly and are immediately strong and aggressive. A rhino should always be moved into sternal recumbency before giving the antidote or it will bash its head on the ground as it attempts to rise from the lateral position. Re-narcotization has been reported in the white rhino, but it is a rare occurrence in the black rhino (Kock, 1990; Portas, 2004).

Out of tradition, opioid antagonists are dosed using empirically derived ratios rather than on a mg-per-kg basis; for the pure opioid antagonist, naltrexone, dosage ratios of 20 to 50 times the etorphine mg dose and 90 to 100 times the carfentanil mg dose are considered standard for captive rhinoceroses (Swan, 1993; Allen, 1996; Kock, 2006). Field workers frequently use lower naltrexone doses (12.5:1 naltrexone to etorphine ratio) without a problem (Kock, 1995); however, sedative signs at these doses have been reported in white rhino and a minimum of 40:1 is recommended to preclude re-narcotization (Rogers, 1993a; Kock, 1995; Portas, 2004). Although naltrexone is considered the agent of choice for complete reversal of narcotic anesthesia, a number of scenarios arise under both captive and field conditions in which a full reversal of an opioid-based procedure is undesirable.

The choice of antagonist and its desired action is dependent on two factors: species and location. Black

rhino are reversed into a crate with nalorphine alone or perhaps with 1 or 2 mg of diprenorphine (Kock, 2006; Morkel, unpublished data) (Tables 48.2 and 48.3). In the boma, *Diceros bicornis* are reversed with naltrexone, although very nervous or aggressive individuals may benefit from reversal with diprenorphine. *Diceros bicornis* are completely reversed in the field using naltrexone; however, because it is expensive and difficult to obtain a combination of naltrexone and diprenorphine is often used for field reversal. In this case, give the standard diprenorphine dose (2–2.5 times the etorphine dose) (Swan 1993) by IM injection together with 10–20 mg naltrexone. In marked contrast to black rhino, white rhino are reversed into a crate using diprenorphine with perhaps 1 or 2 mg of naltrexone. In the boma and field, *Ceratotherium simum* are always reversed with naltrexone. Diprenorphine is often used for transport of *Ceratotherium simum* since its partial agonist-antagonist actions provide significant narcosis during travel. However, diprenorphine has minimal agonist effects in *Diceros bicornis*; therefore, they should be used judiciously for transport in this species. For any partial antagonism in a crate situation it is critical that the rhino be monitored very carefully to prevent excessive head pressing and occlusion of the airway or damage to the neck and limbs. A cattle prodder is a vital piece of equipment in managing sedated rhinos during travel.

Other Drugs and Immobilization Doses

Rhinoceroses can also be immobilized with the other potent opioids carfentanil, fentanyl, and thiafentanil. The following drug dosages are indicated for adult free-ranging rhino in good condition:

- Carfentanil at 2.5–3.0 mg for adult African rhino Hofmeyr, 1975; De Vos, 1978). Carfentanil produces a quick induction and it is not necessary to combine with azaperone or xylazine.
- Etorphine at 1.8 mg plus 30 mg fentanyl (black rhino) (Rogers, 1993b; Kock, 2006).
- Fentanyl alone at 60 mg (black rhino) (Rogers, 1993b).
- Thiafentanil is mixed equally with etorphine. The adult rhino dose is 2 to 2.5 mg thiafentanil plus 2 to 2.5 mg etorphine. This mixture gives a faster induction time than etorphine alone. The usual antidotes for etorphine work well.

Rhinoceros Anesthesia Complications

With opioid-induced cardiopulmonary depression common in anesthetized rhinoceroses, the need may arise to deliver artificial ventilation. For emergency respiratory support in a rhinoceros, push the animal onto its side. A large person uses the knee of one leg (with foot placed firmly on the ground) to vigorously force the abdomen diagonally upward and forward against the diaphragm. This moves the diaphragm, forcing air into and out of the lungs and keeps the animal alive while the IV opioid antagonist takes effect. When one leg is tired use the other leg and recruit additional people to assist. Jumping on the ribs or back of the rhino is ineffective and does nothing but fracture ribs and inflict unnecessary trauma.

Myopathies are common in rhinoceroses that experience excessive chase periods, impaired limb circulation with sternal recumbency or hyperthermia during capture (Figure 48.11). An especially critical period occurs at the time of crate loading and initial transport during field translocation of rhinoceroses. If stimulated to rise too early after partial reversal or with excessive tranquilization, animals may enter the crate and assume a rigid, semi-squatting position with their hind legs. This is undesirable and must be resolved quickly before the muscles are irreversibly damaged (Figure 48.11). Use of the electric prodder on the head never on the hindquarters can often stimulate the animal to rise and stand. If this does not work consider prompt IV administration of diprenorphine or nalorphine. A sling can also be placed under the belly of the animal, just in front of the rear legs to lift the hindquarters (using the crane on the recovery truck) until the strength has returned to the hind limbs.

A very small percentage of black rhinoceroses develop an adverse reaction that the author refers to as the "fat nose syndrome" (Morkel, unpublished data). Essentially the nostrils close up and appear edematous with a much-reduced opening to the nares. The anesthetist is often forced to hold open or pull the nostrils apart. It may indicate a hypersensitivity reaction; morphine is known to cause histamine release in humans and perhaps etorphine—derived from the same group of opium alkaloids—can produce the same uncommon effect in susceptible rhinoceroses.

New Field Anesthesia Techniques

Today's understanding of Rhinocerotidae anesthesia is truly the embodiment of many courageous pioneers

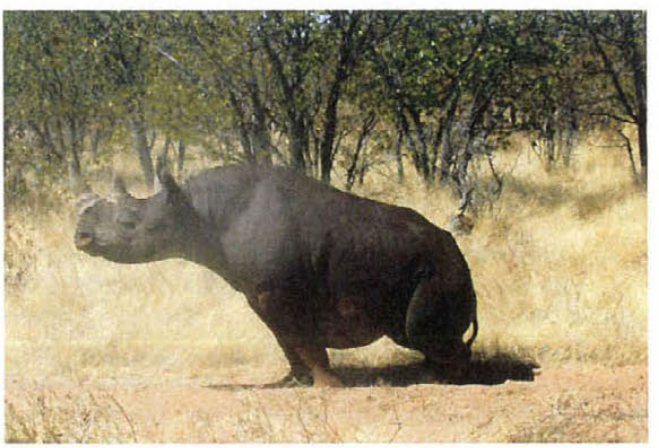


Figure 48.11. *Post-translocation myopathy in a black rhinoceros; capture complications are more prevalent in animals that experience excessive chase periods, hyperthermia, or struggle to stand upon crating. (Image courtesy Birgit Kötting, Etosha Ecological Institute, Namibia.)*

who led by exciting experimentation and hard-won experience (Harthoorn and Lock, 1960; Player, 1972; Young, 1973; Kock, 2006). Yet with the immense challenges inherent in practical anesthesia of these complex mammals, innovative procedures are welcome. The newest ideas for rhinoceros anesthesia are arising from a combination of practical experience and a desire to explore the depths of pharmacology. Nowhere are such explorations more exciting than the emerging science of mixed opioid receptor action on central nervous system activity (Chindalore, 2005). Various opioid receptor affinities and their pharmacological action are well described in humans but remain little understood in animals, including the rhinoceros, which is certain to be unique in many respects. Indeed the most exciting of these novel investigations is, perhaps, the incorporation of mixed agonist-antagonist opioid cocktails as part of routine field capture methodologies for the African rhinoceros (Bush, 2005).

Recent work by Bush and colleagues combines a mixture of concentrated butorphanol (40–90 mg; 30 mg/ml solution) with etorphine and midazolam (2–3.5 mg and 25–50 mg, respectively) (Table 48.2) (Bush, 2005). The addition of butorphanol to the anesthetic combination of etorphine and midazolam produces enhanced muscle relaxation and oxygenation with improved physiological parameters compared with the standard protocol for white rhinoceros albeit with substantially longer induction periods (~ 10min.). Butorphanol is a mixed opioid agonist-antagonist; its agonist κ receptor produces analgesia and marked sedation, whereas the weak μ receptor antagonism reduces respiratory depression and rigidity. The weak σ receptor agonist stimulates respiratory drive. Etorphine is a μ agonist, causing respiratory depression and muscle rigidity. These adverse μ agonist actions are reversed by butorphanol and significantly reduce the cardiopulmonary depression typical of the pure opioids alone (Bush, personal communication).

Besides the marked improvement in oxygen saturation there is a decrease in heart rate closer to normal making the heart a more effective pump. Blood gas values reveal a more normal pH and PCO_2, whereas blood pressures remain lower than with the standard pure opioid agonist protocols. Administering diprenorphine, a μ antagonist, IV 12 minutes into the anesthetic episode reverses etorphine but not butorphanol further counteracting adverse μ effects of etorphine while preserving butorphanol sedation effects. Therefore, if inadvertent opioid overdosage should occur, compromised physiological parameters can be rapidly corrected without losing control of the animal (Bush, personal communication). These discoveries may help to bring field rhinoceros capture into the realm of safety realized with captive animals in which butorphanol-based protocols are now standard replacements for more potent opioids (Radcliffe, 2000a; Portas, 2004).

RHINOCEROS CALF ANESTHESIA

Captive Calf Protocols

Anesthesia of captive white and black rhinoceros calves is safely accomplished with butorphanol alone or in combination with detomidine (Radcliffe, 2000c; Langan, 2001; Gandolf, 2006). Because of high sensitivity to opioid agents, rhinoceros calves respond very well to sedation and anesthesia with mixed agonist-antagonists, precluding many of the adverse cardiopulmonary depressant effects observed with more potent pure agonists of this class. Furthermore, a rapid onset of action is attained by IV delivery or a slower induction by IM administration with both methods proving safe and effective for serial anesthesia (Gandolf, 2006) (Table 48.4). The combination of the α_2-agonist, detomidine, along with the butorphanol was thought to enhance muscle relaxation and depth of anesthesia with IM use in white rhino calves. Complete reversal is achieved using naltrexone at 4 to 5 times the butorphanol mg dose and yohimbine HCl (Yobine) or atipamezole at 0.125 mg/kg for antagonism of the α_2-agent.

Cow and Calf Field Capture

Field immobilization of juvenile rhinoceroses is not without inherent risk, as calves may separate from their dams after darting or become recumbent at different times despite concurrent drug delivery (Figure 48.12). Additionally, calves are more susceptible to capture stress, hyperthermia, and post-capture morbidity and mortality in boma situations (Kock, 1995). Translocation of cows with calves less than 18 months of age can be traumatic and is best avoided, whereas movement of very young calves 2 to 3 months old is particularly high risk. Even with successful translocation, it can be difficult to reunite the cow and calf, as the stress of capture and confinement often results in adult aggression directed toward the calf or the cow drying up. Methods for opioid sedation (0.2 and 0.05 mg etorphine for a cow and calf, respectively) have been used to facilitate boma reintroduction of cow-calf combinations (Kock, 2006). The wild black rhino cow is solitary by nature and usually retreats to a quiet spot to calve and stays there for the first month afterward. Therefore, if a black rhino gives birth in a boma, she rarely manages to raise the calf.

Opioid doses lower than those reported for adult animals are used for juvenile rhinoceroses with subadults receiving approximately one-half the adult dose. For example, when combined with a tranquilizer subadult African rhino (age approx 2.5 years) should receive 1.75 to 2 mg etorphine, whereas very young calves (age 2–3 months) can be immobilized with as little as 0.5 mg etorphine (Rogers, 1993a,b) (Table 48.4). A marked difference is observed in the escape behavior of African rhinoceros cow-calf pairs and should be anticipated during the chase and capture. White rhino calves run ahead of their mothers, whereas black rhino calves

run close at their mothers' heels (Kock, 2006). Additionally, the bond between white rhino cow-calf pairs is much stronger, making separation or splitting a more likely sequela in the black rhino.

When darting a cow with a calf from a helicopter, a fixed-wing aircraft is desirable to circle the capture site to assist with spotting. As a general rule, dart the cow first and about a minute later dart the calf (Kock, 2006). If the timing and darting are good, the pair will often go down together. Should the pair split up, the fixed-wing aircraft can stay with one animal. In open country where visibility is good, the calf can be darted once the cow shows early signs of narcosis. In more thickly vegetated country where it is difficult to observe two separated animals, it is better to wait until the cow shows marked effects or is even recumbent before darting the calf. If the calf splits from its mother, the position of the immobilized mother can be taken by GPS or marked with a smoke grenade or toilet paper and the calf followed. Losing sight of a darted rhino must be avoided; therefore, it is mandatory to have experienced trackers as part of the ground team. When darting a cow-calf pair

Table 48.4. Suggested doses for immobilization and anesthesia of rhinoceros calves under both captive and wild conditions.

	Captive Calves			Wild Calves		
Rhino Species	Protocol	Reversal	Reference Comments	Protocol	Reversal	Reference Comments
White rhinoceros	10–20 mg Butorphanol (BT) IV for 66–159 kg calf (dose 0.13–0.15 mg/kg IV)	Naltrexone at 5 mg per mg BT	Gandolf, 2006 Heavy sedation Light anesthesia Mild resedation noted 8 hours post-reversal in one calf	*Calf*: 0.5–1 mg Etorphine (M99) *Juvenile*: 1.5–2.5 mg Etorphine *Subadult*: 3–3.5 mg Etorphine	Diprenorphine µ50:50 at 2.5 mg per mg Etorphine for transport Naltrexone at 40 mg per mg Etorphine	Kock, 2006 from SANP NOTE: Always dart mother rhino 30–60 seconds BEFORE calf
				NOTE: All white rhinos including calves get 1 mg Diprenorphine µ50:50 plus 10 mg nalorphine ASAP upon recumbency to provide respiratory support		
	2.5–5 mg Butorphanol + 1.5–1.8 mg Detomidine (DET) IM for 69–122 kg calf (dose 0.03 mg/kg BT plus 0.07 mg/kg DET)	Naltrexone at 4 mg per mg BT Yohimbine at 0.125 mg/kg	Gandolf, 2006 Surgical anesthesia	*Calf*: 1 mg Etorphine + 15–20 mg azaperone *Subadult*: 2 mg Etorphine + 30–40 mg azaperone	Diprenorphine at 3 mg per mg Etorphine	Rogers, 1993a
Black rhinoceros	25 mg Butorphanol IV for ~500 kg subadult calf	Naltrexone at 5 mg per mg BT	Radcliffe, 2000c Heavy standing sedation	*Calf*: 1 mg Etorphine *Subadult*: 2 mg Etorphine	Naltrexone at 40 mg per mg M99	Kock, 2006 from SANP
				Calf: 0.5 mg Etorphine + 50 mg Azaperone *Subadult*: 1.75 mg Etorphine + 100 mg Azaperone	Diprenorphine at 3 mg per mg Etorphine	Rogers, 1993b Kock, 2006
				NOTE: Do not use the Diprenorphine µ50:50 plus nalorphine protocol in black rhinos as it will cause arousal INSTEAD: 5 mg Nalorphine IV; titrate to effect		NOTE: Always dart mother rhino 30–60 seconds BEFORE calf
Greater one-horned rhinoceros	Butorphanol IV or IM Use white rhino as model	Naltrexone at 5 mg per mg BT	Author suggestion based on use in African rhino calves	*Calf*: 0.5–1 mg Etorphine + 5 mg Acepromazine *Subadult*: 2–2.5 mg Etorphine + 10 mg acepromazine	Diprenorphine at 2.5 mg per mg Etorphine	Dinerstein, 1990 Same dose used for adult/ sub-adult

From Dinerstein E, Shrestha S, Mishra H. Capture, chemical immobilization, and radio-collar life for greater one-horned rhinoceros. *Wildl Soc Bull* 1990;18:36–41; Atkinson MW, Bruce H, Gandolf AR, et al. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. *J Zoo Wildl Med* 2002;33:157–162; Gandolf AR, Wolf TM, Radcliffe RW. Serial chemical restraint for treatment of decubitus ulcers in two neonatal white rhinoceroses (Ceratotherium simum). *J Zoo Wildl Med* 2006;37:387–392; Kock MD, Meltzer D, Burroughs R, eds. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Harare, Zimbabwe: Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services, 2006.

Figure 48.12. *Anesthesia of rhinoceros calves is challenging, particularly under field conditions in which darting of the cow-calf pair must be well coordinated in order to limit stress on both parent and offspring.*

on foot, the calf usually stays close to its immobilized mother. If approached carefully, the calf can be darted and generally becomes recumbent close to its mother. Note that black rhino calves are skittish and run off more easily than white rhino calves.

Wild subadult greater one-horned rhinoceroses have been immobilized using the same dosage as adult animals (2–2.5 mg etorphine plus 10 mg acepromazine) (Dinerstein, 1990). However, sub-adult animals proved more difficult to capture and often evaded dart attempts by outrunning the trained elephants that are commonly used for field immobilization of greater one-horned rhinos in the tall grassland habitats of India and Nepal. Indian rhinoceros calves were immobilized with 0.5 to 1 mg etorphine and 5 mg acepromazine using shorter 2.5 cm Cap-Chur needles. As with capture of African rhinoceros cow-calf pairs, it is recommended that greater one-horned cows be immobilized before their calves. Calves did not run away and were easier to capture if the mother was immobilized first to avoid trampling risk to calves or aggression toward the ground crew (Dinerstein, 1990).

Conclusion

During the Indian Mutiny a British soldier fired a bullet into the regiment's cherished mascot, a rhinoceros. In a spirit of scientific inquiry the soldier was testing the long held-belief—a conviction still strongly held by many since Dürer's famous rhinoceros—that its skin was held together with rivets like a knight's armor and impenetrable to any volley a person could throw its way. To the surprise of royalty and commoners alike, the rhino quickly expired.

The future of the world's rhinoceroses will remain tenuous, as human conflicts over shared resources escalate and rhino horn continues to be cherished by traditional Asian societies for supposed unicorn-like mythical properties. Nevertheless, it is comforting to know that humans, although solely responsible for the current crisis, are also simultaneously making strides to save the relic rhinocerotoids from their greatest enemy, ourselves. Safe anesthesia of wild and captive rhinoceroses alike will help scientists realize these conservation goals. Let us not make the same mistake as the British soldier and believe, naïvely, that the *armored* rhinoceros is invincible to the actions of our kind.

PRACTICAL STRATEGIES FOR RHINOCEROS FIELD ANESTHESIA

- Darts should be tested and prepared ahead of time, leaving only the drug-loading process to complete immediately prior to capture. Load the dart once you have visualized the rhino—tailoring the dose for size, age, and condition of the animal. The rhino should not be chased while the dart is being loaded. Dart quickly and back off.
- Dart sites must be given special care in rhinoceroses because of the propensity for abscess formation. Rhinoceros skin is thick and tough, making drainage of SC infections unlikely without appropriate wound care. Intramammary antibiotic preparations are common; however, the authors prefer infusion of 500 mg oxytetracycline directly into the dart wound. Oxytetracycline is a broad-spectrum antibiotic in high concentration, stable at room temperatures, viscous (it does not easily run back out of wound), and readily available.
- Tranquilizers are often combined with potent opioids to improve muscle relaxation in recumbent animals and help sedate and calm the rhino during transport.
- The addition of hyaluronidase, a hydrolytic enzyme that increases tissue permeability, greatly improves drug absorption and can markedly shorten the induction time. Xylazine or detomidine in mixture results in a slightly quicker induction and more salivation than azaperone.
- A lower opioid dose must be used for rhino that are in bomas, debilitated, old, or where you cannot get to the immobilized animal quickly (e.g., when darting on foot). Be very careful with animals in poor body condition. In most other situations underdosing of opioids is contraindicated for free-range capture of rhino.
- In general, any need for repeat darting of animals following partial or incomplete injection of immobilizing agents should redeliver the original full immobilizing dose. This is a useful rule for captive animals as well, since repeat darting is often associated with excitation and prolonged drug effects if titration is attempted.
- A rapid induction shortens the period the rhino is moving in a semi-narcotized state and thereby lessens the chance that the rhino will injure itself by encountering a hazard. This is especially true when immobilizing rhino in rough terrain. A quick induc-

tion also limits the exertion and physiological stress associated with increased body temperature, heart rate, oxygen consumption, and related physiological changes. However, caution must be used, as very rapid induction times are often associated with marked respiratory depression, especially in the more susceptible white rhinoceros.

- Nalorphine is useful in African rhinoceroses (Tables 47.2 and 47.3):
  - -To improve respiration, give 5 mg IV for black rhino and 20 to 30 mg for white rhino. Black rhino are very sensitive to nalorphine, so administer small incremental 5-mg doses given IV to effect. Nalbuphine (Nubain) may be used at approximately twice the nalorphine dose (20–40 mg) in a similar fashion for improving respiration in white rhino.
  - -To walk a rhino, start with 10 mg IV in black rhino up to a total dosage of 20 to 40 mg in 5-mg increments. For white rhino give 40 mg nalorphine IV followed by small incremental doses of 10 to 20 mg nalorphine up to 75 mg. Addition of 1 mg diprenorphine may help with arousal.
  - -For transport, wake the black rhino up into the crate with 10 to 20 mg IV nalorphine per 1 mg etorphine. You may also need to give diprenorphine IV at 2–3 × etorphine dose if the animal is pushing or collapsing in the crate. Wake the white rhino with 1 mg diprenorphine plus 1 to 2 mg naltrexone if the animal is pushing in the crate.

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