54 Rhinoceroses

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THE RHINOCEROTIDAE

Introduction

Like the fabricated creature in Albretch Durer's famous lithograph, the rhinoceros has long been a source of mystery, myth, and intrigue. 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 vertebrate organisms, 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 (Emslie & Brooks 1999; Foose & van Strien 1997).

Field anesthesia made possible the rhinoceros conservation success stories of the twentieth century (Harthoorn & Lock 1960; Meadows 1996; Player 1972) 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 2003; Dinerstein et al. 1990; Kock et al. 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 et al. 2005, 2011).

Taxonomy and Evolutionary History

The odd-toed ungulates of the order Perissodactyla include three living families: the rhinoceroses, horses, and tapirs. As the order name denotes, all Perissodactylids bear weight on one (equids) or three (rhinoceroses and tapirs) digits. Rhinoceroses and tapirs are among the most primitive of the world's large mammals and are further grouped into the suborder, Ceratomorpha, based upon 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 specie radiation, culminating in extinction of 10 of the 14 perissodactyl families by the end of the Oligocene (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 & Schoch 2002). An extinct hornless rhinoceros, named Paraceratherium (also called 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 2005; Prothero & Schoch 2002).

Biology and Morphology

The Rhinocerotidae are large terrestrial herbivores that have evolved either a browsing (black, Sumatran, and Javan) or grazing (white and greater one-horned) strategy in order 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

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the browsers and grazers, respectively. Like the equids, fermentation takes place in the cecum and colon. The rhinoceros 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, rhinoceros must eat more, have a relatively fast passage of gut contents, and possess limited time to reabsorb water from the feces. Therefore, rhinoceros 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 rhinoceros, 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 & 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 & Allbrook 1958; Shadwick et al. 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 Durer woodcutting of the Middle Ages. The epidermis, however, is very thin (1 mm) and heavily keratinized, incorporating extensive vasculature, which may predispose the rhinoceros to pressure necrosis, particularly in calves (Cave & Allbrook 1958; Gandolf et al. 2006). Significant body hair is an antiquated trait retained in but one living species, the Sumatran or "hairy rhinoceros," so-called for its shaggy coat of hair (Fig. 54.1). 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 rhinoceros including the wooly rhinoceros *Coleodonta* and massive one-horned hairy *Elasmotherium*, eloquently links the Sumatran rhinoceros with its long and prosperous past.

Figure 54.1. The distinctive hair coat of a Sumatran rhinoceros—a feature linking the primitive Dicerorhinus genus with its prehistoric past (image courtesy of Mohd Khan bin Momin Khan, Malaysia Department of Wildlife and National Parks).

RHINOCEROS IMMOBILIZATION AND CAPTURE

Rhinoceros Capture Beginnings

Before widespread application of chemical capture techniques, early African rhinoceros capture operations utilized ropes and a chase vehicle (made famous in the film *Hatari* staring John Wayne and Hardy Kruger). Although dangerous to the operator and stressful to the animal, some teams in East Africa became remarkably proficient at this form of capture. Chemical capture of rhinoceros was first attempted with the dissociative anesthetic, phencyclidine, and the curariform muscle relaxant, gallamine triethiodide. In 1960, during Operation Noah, many black rhinoceros (*Diceros bicornis*) were saved from the rising waters of the newly constructed Lake Kariba, bordering Zambia and Zimbabwe, using these novel techniques (Child & Fothergill 1962; Condy 1964; Harthoorn & Lock 1960; Meadows 1996). Phencyclidine and gallamine were succeeded by the easily reversible opioids, first morphine and diethylthiambutene, followed quickly by the more potent opioids, including etorphine HCl (M99) (Keep et al. 1969; King 1969; King & Carter 1965). Over the past 50 years, etorphine has become the standard opioid for capture of the African and Asian rhinoceros, with fentanyl citrate (Sublimaze), carfentanil citrate (Wildnil) and thiafentanil oxalate (A3080) proving useful alternatives. Pioneering investigation by early practitioners, such as Toni Harthoorn, Eddie Young, Ian Hofmeyr, Ian Player, and many others, provided the foundation upon which future rhinoceros chemical capture methods including the present work are based (Harthoorn 1976; Player 1972; 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 utilized, but nylon darts (Daninject or Telinject with 60 × 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 rhinoceroses including animals held in bomas to eliminate the excitement phase associated with projectile darting.

For field capture of rhinoceros 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 or stainless steel are the most reliable for field use, especially since power settings and impact energy are high, wind or down drafts from the helicopter can be significant, and the operator is often forced to shoot through vegetation. The dart needle should be 5- to 6-cm long for adult rhinoceros. Rhinoceros 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. A novel spiral-threaded needle was developed by Deon Joubert specifically for use on thick-skinned pachyderms; the dart needle can be easily removed by screwing it out of the animal, thereby reducing skin and tissue trauma (Fig. 54.2; Joubert Capture Equipment, Hadison Park, Kimberley, South Africa).

Figure 54.2. Robust dart needles for the capture of free-ranging rhinoceroses demonstrating the various design configurations of needle tip and barb. The spiral-threaded needles on the right are less traumatic and can be screwed back out of thick rhinoceros skin (photo courtesy of Joubert Capture Equipment, South Africa).

Proper dart placement is essential to ensure good drug deposition. The dart should be placed perpendicular to the skin for deep intramuscular injection (the thick skin of a rhinoceros 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 rhinoceros whether in a zoological setting or in the wild. Prior to induction in captivity, thick padding or heavy mats should be utilized to protect recumbent animals from the concrete floors common in these environments. Myositis and neuropathy are serious potential complications. Traditionally, rhinoceroses immobilized in the field are maintained in or moved into sternal recumbency; however, irreversible muscle damage has developed in this position (especially if the rhinoceros goes down on a slope facing upwards 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 rhinoceros 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. The weight must be taken off the lower legs to "pump" them effectively; this is accomplished by two or more people lifting the upper legs and rolling the animal partially onto its back about 45 degrees from horizontal. We recommend that all black rhinoceros that have undergone any degree of exertion be placed in lateral recumbency for at least a few minutes.

The decision to move white rhinoceroses onto their sides should be based on several factors, including the degree of exertion, presence of muscle tremors, and duration of recumbency. White rhinoceroses 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. Therefore, white rhinoceroses should be positioned initially in sternal recumbency until complete relaxation is achieved (Kock et al. 1995).

The position in which a rhinoceros is placed during recumbency is also an important consideration for respiratory function. It has been observed that oxygen saturation of the blood is higher in sternal than lateral posture. Consistent with observations in domestic

animals and humans, the black rhinoceros has greater dead space in lateral compared with sternal posture as indicated by a lower end-tidal carbon dioxide and higher dead space ratios and volumes in lateral (Morkel et al. 2010). The most appropriate posture for rhinoceroses during anesthesia must be based on the circumstances of each capture and should strike a balance between maintaining respiratory function while providing optimal circulation to the limbs.

Eyes and Ears

The eyes of the recumbent rhinoceros must 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. For black rhinoceroses undergoing translocation, the use of a muslin cloth wrapped tightly around the face several times and secured to the horn with cable ties is preferred to a simple blindfold (Fig. 54.3). This technique serves to fix the eyes in a closed position and quiets the rhinoceros during crate transport. Before blindfolding, foreign material should be washed from the eyes using physiologic saline. The ear canals are plugged with cotton wool or a cloth while the rhinoceros is anesthetized, leaving tabs for quick removal. Alternatively, when a large number of rhinoceroses are to be immobilized, two cloth-covered cotton wool plugs can be joined with cord so they remain together. If the rhinoceros is being transported, its ears should remain blocked for the entire trip. Caution should be used upon removal of the earplugs as the sudden auditory stimulation can result in excitement or aggression.

Figure 54.3. Attachment of the "mutton cloth" blindfold to aid in crate transport of the black rhinoceros (*Diceros bicornis*). Note the use of cable ties in front of the caudal horn to secure the blindfold over both eyes.

Anesthesia Monitoring

A thorough clinical examination with monitoring of vital functions (respiration, temperature, heart rate, capillary refill time) must be conducted regularly for the duration of anesthesia. The focus should be on respiration and blood oxygenation while temperature and heart rate are usually of lesser importance. 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. Any residual dart contents should be checked to see if they 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.

Pulse oximetry (SpO_2) provides an indirect measure of oxygen saturation of hemoglobin (SaO_2) and is a valuable aid to help monitor blood oxygenation and pulse in anesthetized rhinoceros. 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. SpO_2 values often fluctuate so the pulse oximeter should be observed for at least 1 minute; if the SpO_2 does not consistently read above 80% or is declining, an intervention is warranted. 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%; Atkinson et al. 2002; Kock et al. 1995) in rhinoceros underestimate true oxygen saturation of hemoglobin when calculations are made using human formulae (Bush et al. 2004). The sensor clip is attached to the pinnae of the ear after careful scraping with a sharp blade to remove the epidermis or on mucosal folds of the penis, vulva, or rectum. A cloth is placed over the sensor as ambient light can affect the reading. In animals with excessive muscle rigidity or tremors, as is common in immobilized white rhinoceroses, the sensor may fail to obtain an accurate reading. A reflectance probe held against the nasal mucosa works well (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.

Respiratory Gases: Oxygen and Carbon Dioxide Respiratory depression is perhaps the most significant life-threatening complication encountered during routine anesthesia of the rhinoceros (Atkinson et al. 2002; Bush et al. 2004, 2005; Fahlman et al. 2004; Heard et al. 1992; Kock et al. 1995). Large recumbent animals experience cardiopulmonary depression and perfusion-ventilation inequalities because of large size and abdominal organs pressing upon 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 et al. 1992; Kock et al. 1995). Among the African species, these physiologic changes are more prevalent in the white than black rhinoceros (Bush et al. 2004, 2005).

Respiration is the first and most critical function to be monitored in rhinoceros 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 both nostrils and that the blindfold does not restrict airflow. The lower nostril, in particular, should remain clear of obstruction so passive regurgitation of stomach contents is free to drain. A cloth sack can be placed under the head to limit inhalation of dust or other debris. Respiratory rate and depth are noted by observing chest movement. When monitoring breaths on a bouncing vehicle as with immobilized rhinoceros transported on a sledge where it is difficult to watch chest movement, a finger can be hooked in the nostril or a hand held close to the nares to feel for the warm exhaled air. Breathing should be deep and regular. Respiratory rate is approximately 10–15 breaths per minute on induction, going down to 4–8 bpm about 10 minutes post induction when using potent opioids. Respiration must be monitored for at least 30–60 seconds to obtain an accurate picture of ventilatory pattern as immobilized rhinoceroses often give two or three quick breaths followed by a period of apnea. Rhinoceroses often develop apnea when moved into a different position. Animals should be rolled slowly and watched for breathing. A painful stimulus often incites the apneic rhinoceros to take a breath. Observation of venous blood color during venipuncture provides a reliable early indicator of blood oxygenation. Dark red, almost black blood indicates poor oxygenation, while a lighter red color is normal and correlates well with mucous membrane color.

If breathing is slow, the rhino develops apnea or blood oxygenation is poor (SpO_2 consistently less than 80%); partial reversal with nalorphine HBr (Nalline), nalbuphine (Nubain), or butorphanol (Torbugesic) or complete reversal with naltrexone HCl [Trexonil] or diprenorphine are indicated. Nalorphine given intravenously produces a marked and sustained improvement in the quality of respiration (Table 54.2 and Table 54.3). Its use has been associated with an approximate 20% increase in the hemoglobin saturation of oxygen (SaO_2) based on pulse oximetry (Kock et al. 1995). Although widely reported to improve oxygenation (Rogers 1993a, 1993b), recent investigation suggests nalorphine produces negligible change to oxygen partial pressures (PaO_2) in the anesthetized rhinoceros (Bush et al. 2004; Fahlman et al. 2004). Since black rhinoceros stand up readily with very small volumes of nalorphine, we recommend dosing at 5-mg nalorphine for adult black rhinoceros and 25–30 mg for white rhinoceros under field conditions. It is very safe to use nalorphine in white rhinoceros, as this species rarely rises without stimulation and even if arousal does occur adult animals are relatively harmless in a semi-narcotized state. Intravenous doxapram HCl (Dopram; black rhinoceros 200 mg, white rhinoceros 400 mg) provides a smaller, transient improvement in respiratory rate and depth. Doxapram must be used with caution in white rhinoceroses, 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/min) can produce a rapid and significant increase in blood oxygen saturation in immobilized rhinoceros. Although it did not correct systemic acidosis or hypercapnia, O_2 insufflation did substantially improve oxygenation and anesthetic safety (Bush et al. 2004; Fahlman et al. 2004). A variety of factors influence pulmonary blood gas exchange, including dose of anesthetic drug, position of the rhinoceros during immobilization, body temperature, oxygen delivery, and size of the animal. Oxygen supplementation at the flow rates commonly used for rhinoceros appears to produce a more profound improvement of patient oxygenation (PaO_2 108–194 mm Hg) in subadult African rhinoceroses compared with adults, perhaps indicating greater ventilation-perfusion mismatch with larger body size (Fahlman et al. 2004).

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 (edges rounded to prevent damage to nasal mucosa), measuring 2-m long and 9- to 14-mm inside diameter (Fig. 54.4). Concurrent monitoring of the respiratory rate and depth, and

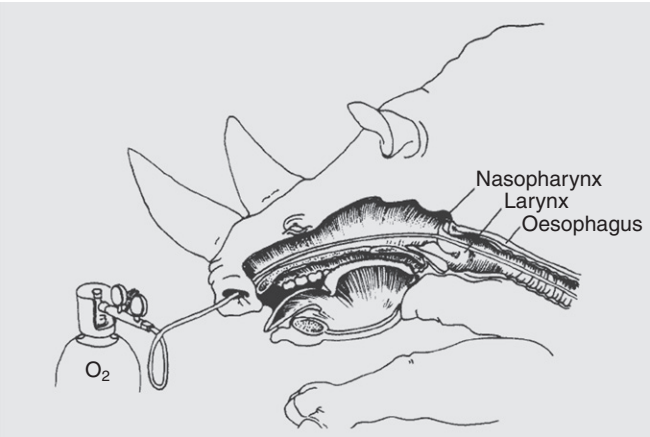

Figure 54.4. Illustration of the nasogastric tube tracheal insufflation technique for oxygen delivery in a recumbent white rhinoceros (*Ceratotherium simum*) under field conditions (adapted from Bush et al. 2004; illustration courtesy of the South African Veterinary Association).

blood oxygenation remains essential. Administration of a partial antagonist (nalorphine, nalbuphine, or butorphanol) will increase the rate and depth of respiration and improve the efficacy of oxygen delivery. Oxygen supplementation is a practical solution to enhance pulmonary gas exchange in immobilized rhinoceros and if used wisely one bottle is sufficient for many animals. Therefore, we recommend immediate intranasal or tracheal insufflation of oxygen at a high flow rate in all recumbent rhinoceroses. Within a few minutes, vital statistics will provide information about respiratory function, and in most situations, all physiologic parameters are satisfactory and the flow can be reduced or discontinued. A small percentage of animals, however, will develop a physiologic crisis where continued oxygen supplementation is critical. Aluminum oxygen bottles are now available that are small and lightweight, making them convenient for helicopter use.

Capnography offers a useful adjunct to monitoring with pulse oximetry, and many portable devices combine measurement of end-tidal carbon dioxide with oxygen saturation of hemoglobin. Capnography is common practice in human anesthesia as an aid to confirm placement of endotracheal tubes and for early warning of hypoventilation. In expired respiratory gases, capnography provides a direct measure of CO_2 eliminated from the lungs. It also gives an indirect measure of CO_2 production in the tissues and circulatory transport of CO_2 to the lungs. Capnography is a rapid noninvasive technique that offers reliable information about tissue CO_2 production, circulation, pulmonary perfusion, and alveolar ventilation and respiratory patterns. For rhinoceros capture, capnography is gaining acceptance by practitioners as a tool to help enhance anesthetic safety by offering a quick method to detect ventilatory or circulatory failure (Morkel et al. 2010).

Body Temperature Body temperature is an important parameter and the best indicator of the degree of exertion endured by the rhinoceros before induction. For every 1° increase in body temperature above normal, there is approximately a 10% increase in oxygen consumption. A rhinoceros's body temperature varies slightly during the day as the ambient temperature changes. Black rhinoceros immobilized without excessive exertion have a rectal temperature of between 36.5 and 38.5°C, and anything over 38.5°C should be liberally doused with cool water. Young rhinoceros tend to have a higher body temperature than adult rhinoceros after running a comparable distance. Although drenching with water is important, it will not have a dramatic effect in lowering the core body temperature, as there is considerable thermal inertia in such a large mammal. It helps to fan the rhinoceros with branches or a portable leaf blower after the animal has been wetted with water. Holding leafy branches over the rhinoceros to provide shade can help lower the temperature, but it is important that people do not crowd around an immobilized rhinoceros and prevent air movement. A beach umbrella is the most efficient way to make shade—it is cheap, light, and easy to transport and can be folded to fit in the helicopter. A rhinoceros with a body temperature over 39°C must be processed quickly, while a temperature greater than 41°C mandates immediate delivery of the antidote.

The rhinoceros, like other large mammals, is prone to hyperthermia during capture and translocation. The black rhinoceros appears to suffer a greater level of hyperthermia-related morbidity and mortality in the peri-capture period than the white rhinoceros. The goal of the capture team should be to minimize exertion and speed induction. The rise in body temperature can be documented while the animal is in recumbency following excessive exertion; however, a second phase apparently unrelated to the level of exertion appears to occur upon crating. The mechanism of this hyperthermic response observed at variable periods after the rhinoceros enters the crate is not well understood, but could result from postreversal agitation and muscle activity, a physiologic stress response, inadequate airflow inside the crate, or a combination of factors or mechanisms. Simultaneous comparison of rectal and muscle temperatures in recumbent rhinoceroses demonstrates a 1°F (0.5°C) higher temperature in the muscle (Morkel et al. 2012). Since equilibration between rectal and muscle temperatures occurs slowly with time, a deep muscle thermistor can be a useful aid to measuring core body temperature. The probe is readily inserted into the dart site; however, caution must be taken to avoid exposure to dangerous immobilizing drugs.

Additional options for temperature measurement in the rhinoceros include use of a handheld infrared thermometer (Fluke Hart Scientific Inc.) in the ear canal. In a recent pilot study, deep infrared ear temperatures were comparable with deep muscle temperatures, giving a reliable and rapid assessment of core body temperature. Surprisingly, the use of cotton or wool earplugs resulted in ear canal temperature above deep muscle temperature and suggests that the inner ear in the rhinoceros may be an important site for cooling. In animals predisposed to hyperthermia (prolonged capture, high environmental temperature, etc.), rhinoceroses may benefit from removal of the earplugs and perhaps even application of some cooling mechanism to this site, such as a cold pack (Morkel et al. 2010, 2012). Another tool that has been adopted to enhance temperature monitoring in rhinoceroses is the temperature microchip (LifeChip Inc.). After reaching equilibrium when implanted under the skin or into a muscle, chip temperatures were slightly lower than rectal temperatures.

Pulse and Blood Pressure Heart rate is best obtained using a stethoscope while the pulse is readily palpable on the inside of the ear (medial auricular artery) or under the base of the tail (caudal artery). Pulse quality should be evaluated subjectively and compared 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 55–80 beats per minute, although it will be higher in rhinoceros that have undergone marked exertion, especially in young animals (up to 140 bpm). Cardiovascular function and peripheral perfusion are assessed by capillary refill time (CRT) and is measured by blanching the rhinoceros's gum for several seconds and then releasing. The observed delay or refill time should not exceed 2 seconds.

Hypertension is prevalent under etorphine anesthesia in black and white rhinoceros (Hattingh et al. 1994; Heard et al. 1992; LeBlanc et al. 1987). One report in white rhinoceros anesthetized under field conditions noted an apparent reduction in blood pressures when azaperone tartrate (Stresnil) replaced fentanyl in etorphine-based combinations, an effect observed despite the higher dose of etorphine used in the cocktails containing azaperone (Hattingh et al. 1994). These conclusions are questionable since fentanyl itself is 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 purported factors in etorphine-induced hypertension in both rhinoceroses and equids (Daniel & Ling 1972; Heard et al. 1992). Opioid-related hypoxemia may induce sympathetic system stimulation and hypertension. Elevated heart rate is a good indicator of hypoxia; 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 pharmacologic agents (Raath 1999). Surprisingly, the same dose of carfentanil citrate used to immobilize a 20-kg blackbuck (*Antelope cervicapra*) would also fully immobilize a 2200-kg white rhinoceros (*Ceratotherium simum*) making the rhinoceros over 100 times more opioid sensitive per unit mass than the average artiodactylid. This inordinate sensitivity of the rhinoceros family to the opioid class—while 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 (Bush et al. 2005; Radcliffe et al. 2000a; Walzer et al. 2000).

Planning for anesthetic events should include preparation of the subject and environment where these variables can be controlled. Depending on the purpose for anesthesia, it is generally desirable to fast the animal for 12–48 hours prior to anesthesia (Radcliffe et al. 2000b). However, fasting is certainly not essential as evidenced by the many successful field operations where capture of wild rhinoceroses is conducted in the absence of preanesthetic 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 rhinoceros (Raath 1999). Both passive and active regurgitation of stomach contents are known, with the latter being very rare but quite spectacular. Passive regurgitation is common in immobilized rhinoceroses, presumably secondary to drug or hypoxemia-induced relaxation of the cardiac sphincter (Fig. 54.5). 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 rhinoceros barns or bomas for acclimatization to the sights, sounds, and smells of the veterinary profession will 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.

Figure 54.5. Passive regurgitation from the nares of a black rhinoceros (*Diceros bicornis*) forcefully expelled at expiration. The rhino is positioned in sternal recumbency to help the reflux drain away from the airway.

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 (Fig. 54.6). 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 intramuscular azaperone or detomidine 20–30 minutes prior to induction with etorphine helps preclude muscle spasms and rigidity. With wild rhinoceroses, positioning in sternal recumbency until complete relaxation is achieved was deemed important in field practice (Kock et al. 1995).

Mixtures of etorphine or carfentanil combined with a sedative are standard agents for anesthesia of the captive white rhinoceros (Table 54.1). Doses ranging from 0.8 mg up to 3 mg of etorphine and 1.2 mg carfentanil are common, with supplemental opioids given IM or IV to extend anesthesia (Heard et al. 1992; Walzer et al. 2000). Following immobilizing doses of etorphine or carfentanil, other agents provide additional muscle relaxation and a deeper plane of anesthesia, including intravenous alpha-2 agents, propofol, guaifenesin, ketamine, and midazolam (Klein et al. 1997; Kock et al. 2006; Walzer et al. 2000; Zuba & Burns 1998). 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 et al. 1992). One captive white rhinoceros immobilized with etorphine remained hypoxemic despite maintenance of inhalation anesthesia using intermittent partial positive pressure ventilation (Cornick-Seahorn et al. 1995). Hypertension is common, while hypoventilation, pulmonary shunting, and atelectasis induce hypoxia and hypercapnia (Bush et al. 2004; Heard et al. 1992).

Butorphanol 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 et al. 2000a, 2000b, 2000c). While 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 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 & Morkel 2007; Radcliffe et al. 2000a, 2000c). Butorphanol doses for white rhinoceros range from 50 to 120 mg for an adult and 10 to 20 mg for a calf or juvenile animal, while azaperone doses range from 100 to 160 mg for an adult with supplemental doses given up to a maximum of 300 mg (Table 54.1 and Table 54.4). Intravenous butorphanol supplementation is effective at inducing recumbency in white rhinoceroses after initial drug delivery, if needed and desirable. Intravenous dosing of azaperone without prior sedation has been associated with adverse extrapyramidal reactions in the horse and white rhinoceros and should be avoided (Radcliffe et al. 2000a).

Inhalation anesthesia is possible in captive rhinoceroses where more invasive procedures requiring surgery or longer anesthesia times are indicated. Intubation in the rhinoceros is accomplished by hand or with an endoscope to guide placement of the endotracheal tube or a guide catheter into the airway. Unlike the horse, rhinoceroses have a unique, blind diverticulum in the dorsocaudal pharynx above the glottis. The anesthetist must be careful not to pass the tube into the diverticulum during intubation (Radcliffe et al. 1998).

A white rhinoceros was safely anesthetized to treat a surgical colic by sedation with butorphanol (80 mg) and detomidine (50 mg), followed by IV glyceryl guaiacolate (50 g) and three boluses of ketamine (200 mg per bolus) for induction (Valverde et al. 2010). Anesthesia was maintained for an additional 6 hours using isoflurane in oxygen delivered at 1–2% using a circle breathing system. Positioning for surgery and recovery were challenging, but made possible through use of inflatable mats and expertise of the local fire department. Although the rhino eviscerated 3 days post surgery, the anesthesia and recovery were considered a success. Inhalation anesthesia together with a sling system facilitated laparoscopic-assisted transvaginal oocyte recovery in a black rhinoceros (Portas et al. 2006). Abdominal laparoscopy with insufflation of the abdomen is possible in rhinoceroses without general anesthesia by using a standing approach (Radcliffe et al. 2000b).

Black Rhinoceros (*Diceros bicornis*) Black rhinoceros 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 rhinoceroses experience while also significantly reducing the total dose of opioid agents required (Radcliffe & Morkel 2007; Table 54.1). 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 rhinoceros's eyes should be covered and ears blocked as soon as possible. Significant induction risks include lacerations, limb and foot

Table 54.1. Suggested doses for chemical restraint of adult *captive* rhinoceroses producing anesthetic planes from sedation to recumbency

	Standing Sedation			Recumbency		
Rhino Species	Protocol	Reversal	Reference and Comments	Protocol	Reversal	Reference and Comments
White rhinoceros	50- to 70-mg butorphanol (BT) + 100-mg azaperone IM hand-injection plus constant rate infusion (CRI)	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000a, 2000b); use CRI in long procedures	70- to 120-mg butorphanol + 100- to 160-mg azaperone IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000a); supplemental IV dosing or CRI
	120- to 150-mg butorphanol + 5- to 7-mg medetomidine (MED) IM dart (Give 1- to 2-mg nalorphine IV to keep standing)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (2008)	120- to 150-mg butorphanol + 5- to 7-mg medetomidine (IM dart; recumbency ~20 minutes)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	S. Citino, unpubl. data; improved analgesia for surgery
	0.8- to1.5-mg etorphine (M99) IM dart	Naltrexone at 40 mg per mg M99	Portas (2004)	2- to 3-mg etorphine + 20- to 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- to 50-mg butorphanol IV or IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000c) and unpubl. data; use for subadults and crating	1- to 1.5-mg etorphine + 100-mg azaperone IM hand-injection	Naltrexone at 50 mg per mg M99 1/2IV 1/2IM	R. Radcliffe, unpubl. data; lower M99 doses with hand-injection
	1.5- to 2-mg etorphine + 2- to 3-mg medetomidine (Give 1- to 2-mg nalorphine IV to keep standing) IM dart	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino (2008)	1.5- to 2-mg etorphine + 2- to 3-mg medetomidine (IM dart; recumbency ~15 minutes)	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	S. Citino, unpubl. data; enhanced analgesia for dental surgery
	2- to 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- to 3-mg etorphine + 60 mg azaperone IM dart	Naltrexone at 20–40 mg per mg M99	Portas (2004)

(*Continued*)

Table 54.1. (*Continued*)

	Standing Sedation			Recumbency		
Rhino Species	Protocol	Reversal	Reference and Comments	Protocol	Reversal	Reference and Comments
Greater one-horned rhinoceros	100-mg butorphanol + 100-mg azaperone IM hand injection	Naltrexone at 2.5 mg per mg BT IM or IV	R. Radcliffe and N. Lung, unpubl. data	3.5- to 3.8-mg etorphine + 14-mg detomidine + 400-mg ketamine IM pole-syringe	150- to 300-mg Naltrexone 1/2IV 1/2IM No reversal DET	Atkinson et al. (2002)
Sumatran rhinoceros	25- to 40-mg butorphanol IM hand-injection *Note*: Sumatran rhinoceros are easily conditioned to chute restraint and can be hand-injected. Even wild rhinos can be tamed quickly with feed and walked into a crate without use of drugs!	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2002); use azaperone in longer procedures	30- to 50-mg butorphanol + 50- to 60-mg azaperone IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2004); higher doses for recumbency
				1-mg etorphine + 60-mg azaperone IM hand-injection	Naltrexone at 50 mg per mg M99 1/2IV 1/2IM	R. Radcliffe, unpubl. data; azaperone 20 minutes < M99 or suppl. midazolam
				10-mg butorphanol + 10-mg detomidine IM dart Wait 20 minutes 1.2-mg etorphine 5-mg acepromazine IM dart	150-mg naltrexone IV + 20-mg atipamezole IV	Walzer et al. (2010); 530-kg adult male; ketamine boluses (50 mg each) to extend anesthesia

Sources: Citino SB. 2008. Use of medetomidine in chemical restraint protocols for captive African rhinoceroses. Proceedings of the American Association of Zoo Veterinarians and Association of Reptilian and Amphibian Veterinarians, pp. 108–109; R. Radcliffe unpubl. data; Atkinson MW, Bruce H, Gandolf AR, Blumer ES. 2002. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. *Journal of Zoo and Wildlife Medicine* 33(2):157–162; Portas TJ. 2004. A review of drugs and techniques used for sedation and anaesthesia in captive rhinoceros species. *Australian Veterinary Journal* 82(9):542–549; Walzer C, Goritz F, Hermes R, Nathan S, Kretzschmar P, Hildebrandt T. 2010. Immobilization and intravenous anesthesia in a Sumatran rhinoceros (*Dicerorhinus sumatrensis*). *Journal of Zoo and Wildlife Medicine* 41:115–120.

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 rhinoceros is characterized by rapid and powerful recoveries mandating extra care; never stand in front of a narcotized rhinoceros as arousal is often sudden and unpredictable (Kock et al. 2006).

As in the other rhinoceros species, potent opioids (primarily etorphine) have historically been used for anesthesia of captive black rhinoceros 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 54.1). Butorphanol alone or in combination with azaperone or detomidine HCl (Dormosedan) has also been used in the black rhinoceros, although its use is primarily limited to subadult animals, crating and translocation procedures, or well-conditioned animals since black rhinoceros are easily excitable and may override drug effects (Radcliffe et al. 2000c). In addition to reversing the respiratory depressant effects at the μ receptors, butorphanol also antagonizes the powerful μ sedative effects, thereby greatly lightening anesthesia. For butorphanol use in the black rhinoceros, expect light planes of anesthesia and the need for frequent redosing. A more thorough discussion of mixed agonist-antagonist opioid cocktails and newer alpha-2 agents for use in both captive and field immobilization protocols for the African rhinoceroses can be found in the New Techniques section of this chapter and Table 54.1 and Table 54.2.

Asian Rhinoceros Captive Anesthetic Regimens

Indian or Greater One-Horned Rhinoceros (*Rhinoceros unicornis*) Despite the common occurrence of Indian rhinoceroses in zoological parks and a propensity for foot problems necessitating chronic care, few published accounts of anesthesia in captive greater one-horned rhinoceros exist (Atkinson et al. 2002; Portas 2004). One report combined injectable and inhalation anesthesia in a female *Rhinoceros unicornis* for ovariohisterectomy 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 postsurgical complications (Klein et al. 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 and 10–14 mg IM, respectively) was given by projectile dart or etorphine-detomidine-ketamine (3.5–3.8, 14, and 400 mg IM, respectively) administered by pole-syringe (Atkinson et al. 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. While 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 et al. 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 (R. Radcliffe and N. Lung, unpubl. data., 2004). A combination of butorphanol and detomidine (120 and 80 mg, respectively) produced sternal recumbency for surgical repair of a rectal prolapse (Bertelsen et al. 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 hyrdochloride (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 (Bertelsen et al. 2004; Gandolf et al. 2000; Portas 2004; Radcliffe et al. 2000a).

Javan or Lesser One-Horned Rhinoceros (*Rhinoceros sondaicus*) *Rhinoceros sondaicus* is the only rhinoceros 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 rhinoceros 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 rhinoceros can be found in Ujung Kulon National Park in West Java ($n \sim 40$) and Cat Tien National Park in Vietnam ($n \sim 3$?). 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 since 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 rhinoceroses (Portas 2004; Radcliffe & Morkel 2007). One adult male was immobilized on two occasions using a two-stage darting protocol. The

Table 54.2. Suggested doses for chemical restraint of adult *wild* rhinoceroses including supplemental agents used for respiratory support

	Immobilization			Respiratory Support	
Rhino Species	Protocol	Reversal	Reference and Comments	Protocol	Reference and Comments
White rhinoceros	2- to 3.5-mg etorphine (M99) + 40- to 90-mg butorphanol (BT) + 25- to 50-mg midazolam (MDZ) IM dart. Detomidine, medetomidine, midazolam, or azaperone can be added to this mix: (a) for better muscle relaxation and often better blood oxygenation—DMM; (b) to speed induction—DMMA.	Naltrexone at 40 mg per mg M99 IV (full reversal) OR 2- to 2.5-mg diprenorphine (M50:50) per mg M99 IV (reverses M99, but not BT)	Bush et al. (2005, 2011); reduces respiratory depression, hypoxia, tachycardia, muscle rigidity, and tremors, but with slower induction and an animal that may stay on its feet. Avoid butorphanol in combination with etorphine in rough terrain where a quick induction is safer.	• 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 et al. (2005, 2011) and unpubl. data; reverse part or all of opioid effects based on desired outcome
	3- to 4.5-mg etorphine + 40- to 60-mg azaperone (replace azaperone with 10- to 20-mg detomidine if no transport) IM dart Consider 5- to 20-mg midazolam slowly IV for muscle relaxation	For crate reversal: 2.4-mg M50:50 per 1-mg M99 plus 1- to 2-mg naltrexone IV if pushing. NTX will be a relatively "lively" wake up and one must be adequately prepared (i.e., animal close to crate, rope properly attached to head, and well-organized team) For field/boma reversal: naltrexone at 40 mg per mg M99 IV (full reversal)	Kock et al. (1995, 2006) Rogers (1993a); still considered standard translocation protocol P. Morkel, unpubl. data Kock et al. (1995)	All white rhino: • 20-mg butorphanol per mg M99 (20X M99 considered minimum dose for white rhinoceros, reduce to 10X or 15X if light) OR • 20- to 30-mg nalorphine IV OR • 20- to 40-mg nalbuphine IV AND/OR • 1-mg M50:50	Kock et al. (1995, 2006) M. Hofmeyr and P. Morkel, unpubl. data; Butorphanol is being used with greater frequency for its partial agonist properties in white rhinoceros across Africa

Black rhinoceros	4-mg etorphine + 40- to 60-mg azaperone (Replace azaperone with 100-mg xylazine or 10-mg detomidine) + 5000 IU hyaluronidase IM dart. Azaperone can be increased to 200 mg for a quicker induction if no transport. Can also combine azaperone with alpha-2 agonists.	For crate reversal: 20-mg butorphanol per mg M99 OR 5- to 20-mg nalorphine per mg M99 or 1–1.8 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); Kock et al. (2006) Hyaluronidase is always recommended to speed induction, especially as black rhino (unlike white rhino) often run themselves into trouble	• Give 5-mg butorphanol IV to increase respiration and heart rate and lighten the anesthesia. A 10-mg dose will considerably lighten anesthesia and rhino may stand. Animal less likely to stand if kept in lateral position and rhino can be easily pushed down, if necessary • *Note*: Do *not* use the white rhino respiratory protocol in black rhino as it will cause arousal	M. Hofmeyr, unpubl. data; P. Morkel unpubl. data; Kock et al. (2006)
	2- to 2.5-mg thiafentanil (A3080) + 2- to2.5-mg etorphine IM dart. Can also use thiafentanil alone at etorphine doses (i.e., up to 5 mg) but watch the respirations	Same	Rogers (1993b)	• *Instead*: 5 mg nalorphine IV; titrate to effect • *Important* to have animal lateral and "pump" legs every 20 minutes	
Greater one-horned rhinoceros	2- to 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 et al. (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 et al. (1990)
Sumatran rhinoceros	2-mg etorphine + 80-mg azaperone + 5000 IU hyaluronidase IM dart OR Use M99 : BT : MDZ	Naltrexone at 50 mg per mg M99 IM or IV	Author suggestion (extrapolated from captive animals)	• 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	Radcliffe et al. (2004) and unpubl. data
	80-mg butorphanol + 80-mg azaperone IM dart	Naltrexone at 2.5 mg per mg BT IM or IV	Use for compromised animal in snare	• If rhino is approachable give 25- to 40-mg butorphanol IV rather than via dart • Sumatran rhinoceros are easily tamed and can even be fed into a crate; a temporary boma can be erected to facilitate capture and crating	Radcliffe et al. (2004) and unpubl. data

Sources: P. Morkel, unpubl. data; R. Radcliffe, unpubl. data; Bush M, Citino SB, Grobler D. 2005. Improving cardio-pulmonary function for a safer anesthesia of white rhinoceros (*Ceratotherium simum*): use of opioid cocktails to influence receptor effects. Proceedings of the American Association of Zoo Veterinarians, American Association of Wildlife Veterinarians and American Zoo and Aquarium Association Nutrition Advisory Group, pp. 259–260; Bush M, Citino SB, Lance WR. 2011. The use of butorphanol in anesthesia protocols for zoo and wild mammals. In: *Fowler's Zoo and Wild Animal Medicine Current Therapy 7* (RE Miller, ME Fowler, eds.), Chapter 77, pp. 596–603; Kock MD, Meltzer D, Burroughs R, eds. 2006. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services; Rogers PS. 1993a. Chemical capture of the white rhinoceros (*Ceratotherium simum*) OR 1993b. Chemical capture of the black rhinoceros (*Diceros bicornis*). In: *The Capture and Care Manual* (AA McKenzie, ed.). Pretoria: Wildlife Decision Support Service and South African Veterinary Foundation.

first doses were considered inadequate and the authors subsequently recommended 10-mg butorphanol plus 10-mg detomidine IM followed 20 minutes later with 1.2-mg etorphine and 5-mg acepromazine IM, plus 50-mg supplemental doses of ketamine IV to extend the anesthesia period (Walzer et al. 2010). Darting of this animal likely contributed to long induction times (up to 40 minutes)—Sumatran rhinoceros are easily conditioned for hand injection in a chute. As with the African species, muscle rigidity and cardiopulmonary depression are common with use of the potent opioids, and preanesthetic 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 while the author routinely uses a mixture of butorphanol and azaperone for standing sedation and full recumbent procedures (Table 54.1; Radcliffe et al. 2004).

As with the African species, butorphanol combinations are preferred in captive Sumatran rhinoceros to preclude the adverse cardiopulmonary changes associated with use of more potent opioids. For adult animals, butorphanol at a dose of 60–80 μg/kg with azaperone at 80–100 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 induce recumbency. Antagonism of the butorphanol effects is accomplished with naltrexone at a dose of 2.5 times the dose of butorphanol (Table 54.1; Radcliffe et al. 2004). Other tranquilizers may be used in place of azaperone, such as the alpha-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 pharmacologic agents, such as ketamine and medetomidine, 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 et al. 1992; Klein et al. 1997). Standing restraint where possible using mixed agonist-antagonists show promise (Radcliffe et al. 2000a, 2000b). For the black rhinoceros, where potent opioids are still often preferred over mixed agonists, challenges include marked respiratory depression, inadequate muscle relaxation, need for frequent redosing, and incomplete analgesia in painful procedures. The incorporation of the potent alpha-2 agonist medetomidine with etorphine or butorphanol enhances sedation and analgesia in captive rhinoceroses (Citino 2008). Because alpha-2 agonists exacerbate respiratory depression and hypotension, contribute to dehydration, and alter thermoregulatory mechanisms they must be used with caution in rhinoceroses of unknown health status, especially old and debilitated animals. However, under captive conditions, where the health of an animal is known and a specific type of anesthesia is desirable, alpha-2 agents are effective supplements.

For the black rhinoceros, medetomidine (2–3 mg representing 2–2.9 μg/kg IM; 20 mg/mL solution) is combined with etorphine (1.5–2 mg representing 1.5–1.7 μg/kg IM; Citino 2008) 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 extraction and foot surgery with the additional supplement of an intravenous guaifenesin-ketamine drip (1 g of ketamine in one liter 5% GGE solution) to enhance peripheral analgesia. Relaxation was excellent with easy access to the oral cavity for dental surgery. Physiologic 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 rhinoceros, where butorphanol has proven so effective in captive settings, the same investigator is using medetomidine (5–7 mg representing 2.47–2.81 μg/kg IM) and butorphanol (120–150 mg IM representing 62.5–64.9 μg/kg IM) to provide enhanced muscle relaxation and analgesia properties (Citino 2008). The animals can be manipulated within approximately 11 minutes of intramuscular drug delivery with full recumbency in 20 minutes. The addition of medetomidine into these protocols has significantly improved muscle relaxation and 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 (204–262 μg/kg naltrexone) and atipamezole at 5 mg per mg medetomidine (25.4–31.2 μg/kg atipamezole).

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 et al. 1990; Flamand et al. 1984). Ideally, rhinoceros capture operations should be conducted when temperatures are lower than 25°C, usually in the early

morning or late afternoon. Darting free-ranging rhinoceros when ambient temperatures are high increases the risk of elevated body temperatures and associated physiological stress. If working in the late afternoon, a rhinoceros should not be darted unless there is sufficient daylight remaining (an hour is a minimum time to process the animal and deal with potential problems; Rogers 1993a, 1993b). If a rhinoceros has run hard enough for its skin to become dark with sweat, the rhinoceros's body temperature will often exceed 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 rhinoceros rises above 41°C, the antidote should be administered immediately and the animal released.

With good dart placement, recumbency should follow within 3–6 minutes post drug delivery (Kock et al. 2006; Morkel 1989, 1994). Induction is usually quicker in young rhinoceroses and longer in large bulls and heavily pregnant cows. If there are no signs at about 6 minutes, the rhinoceros 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; oxygen combined with a partial antagonist should be given, if necessary. In protocols incorporating thiafentanil, rapid inductions are expected and less of a concern. Intravenous opioid use should be avoided due to 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 alpha-2 agonists by the intravenous route.

As a rhinoceros becomes affected by etorphine, its pace shortens, the forelegs are lifted higher in a classic "Hackney gait," and the head is elevated (Fig. 54.6).

Figure 54.6. 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 of Rolfe Radcliffe, Living Fossil Productions).

The rhinoceros then starts to blunder through bushes and slows down before going into lateral or sternal recumbency. In rough terrain, rhinoceros 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, rhinoceroses 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 may be compromised, especially in heavily pregnant females, who have the weight of the fetus, adding additional pressure. Such animals must be immediately pushed onto their sides. Usually, a rhinoceros will become fully recumbent; 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 rhinoceros, a quick estimate of its age and body condition should be made. Older or debilitated rhinoceros need special care. Nothing should impede respiration or push against the rhinoceros's belly, chest, or nostrils. On a slope, the rhinoceros should face uphill to alleviate pressure against the diaphragm. Field personnel must work quickly while the rhinoceros is recumbent—it helps to prepare a prioritized checklist before beginning each field capture exercise (Flamand et al. 1984; see also *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 rhinoceroses represents one of the most challenging branches of rhinoceros anesthesia (Table 54.2). Hypoxia, hypercapnia, hypertension, tachycardia, and acidosis are common physiologic abnormalities reported in anesthetized white rhinoceros (Bush et al. 2004; Heard et al. 1992). A variety of techniques have been adopted to help alleviate the significant opioid-induced cardiopulmonary depression in African rhinoceroses. These include use of partial agonist-antagonist agents 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 (Bush et al. 2004, 2005; Fahlman et al. 2004; Kock et al. 1995; Radcliffe et al. 2000a).

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 54.2; Bush et al. 2004; Kock et al. 1995; Rogers 1993a). Hyaluronidase (Hylase; 5000 IU) is often incorporated into darting protocols for rhinoceros to shorten induction times (Morkel 1989). White rhinoceroses

stopped moving 2–3 minutes sooner with hyaluronidase, but often remained standing (Kock et al. 1995). Fentanyl was once incorporated into drug cocktails for white rhinoceros but is rarely used today (1 mg of etorphine being equipotent to 15 mg of fentanyl; Rogers 1993a). The parasympatholytic agent, hyoscine, was historically combined with opioids for its sedative and amnesic properties, as well as to induce "temporary blindness" by pupillary dilation (Player 1972; Rogers 1993a). However, its use is no longer widely accepted because of undesirable side effects and is now considered obsolete (Kock et al. 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 Zimbabwe to enable dehorning operations (Kock et al. 1995). Initial immobilization mortality was quite high at 7% and was primarily attributed to hypoxemia and cardiovascular collapse. Subsequent captures utilized lower opioid immobilizing doses and simultaneously incorporated routine use of nalorphine (10–20 mg) or nalbuphine HCl (20–40 mg) to help improve respiration—especially in longer procedures, where 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 et al. 1995). Good muscle relaxation was observed without the rigidity and paddling common with use of potent opioids in the white rhinoceros. 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 rhinoceroses is the use of midazolam (Radcliffe & Morkel 2007; Table 54.2). Since immobilized white rhinoceros are often first encountered in a standing position with a rigid body posture, intravenous midazolam at 5–20 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 redosing with etorphine resulted in poorer muscle relaxation and increased head shaking, jerking, and limb paddling (Kock et al. 1995). Midazolam has excellent muscle relaxation properties and is a useful adjunct in these situations.

Black Rhinoceros (*Diceros bicornis*) Capture-related stress is a significant factor in field immobilization of the black rhinoceros, resulting in morbidity and mortality in the postcapture period (Keep 1973; McCulloch & Achard 1969). Rapid immobilization using high opioid doses in combination with hyaluronidase is the single most critical factor in reducing stress during black rhinoceros capture operations (Kock 1992; Morkel 1989, 1994). 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 rhinoceros (Kock et al. 1990; Rogers 1993b), subsequent study suggests that 3 mg of etorphine is inadequate due to 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 rhinoceros bull or cow in good body condition (Table 54.2; Morkel 1989, 1994).

A scaled-down opioid dose should be utilized 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 et al. 2006). Azaperone is incorporated into etorphine-based African rhinoceros immobilization protocols at 40–60 mg total dose (Table 54.2). Concentrated (100 mg/mL) azaperone solutions should be carefully examined before use as they often crystallize under field conditions. Xylazine or Detomidine (100 or 10 mg per adult, respectively) can be substituted for azaperone based on individual preference.

A disparity is evident in the opioid dose required for immobilization of the various subspecies of black rhinoceros. The desert subspecies (*Diceros bicornis bicornis*) needs a slightly higher dose than the other subspecies. While 5 or even 6 mg etorphine may be necessary for an adult *D. b. bicornis* in good condition, 4 mg is usually more than adequate for a comparable response in animals of the *Diceros bicornis minor* or *Diceros bicornis michaeli* subspecies. Not only is there variation between subspecies, but there also appears to be some difference among individuals. The capture veterinarian must therefore 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 2003; Dinerstein et al. 1990). Furthermore, translocation programs are proving essential for reaching long-term population management goals

for *R. unicornis* in India and Nepal. Capture of wild greater one-horned rhinoceros is usually conducted from atop trained elephants to facilitate finding and darting of rhinoceroses among the dense tall-grass habitats in the floodplain grasslands and riverine forests, where these rhinoceroses flourish. In addition to providing an elevated platform, elephants (10–15 animals) are used to surround the target rhinoceros before and after darting to facilitate observation of the animal during induction and to prevent escape into open water (Dinerstein et al. 1990).

Adult greater one-horned rhinoceros weigh an estimated 2000 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 and 10 mg, respectively) delivered via remote intramuscular injection either in the shoulder or rump using Cap-Chur darts with 5-cm needles (Table 54.2; Dinerstein et al. 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 nonbreeding 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 et al. 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 rhinoceros, pitfall trap methodologies rather than stockade style traps are recommended for capture of lesser one-horned rhinoceros 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 should 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 rhinoceroses using corral or stockade traps with little or no success (Abdullah 1987; Sadmoko 1990). In one instance, an adult female Sumatran rhinoceros 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 rhinoceros in the forests of Southeast Asia has been most effective by use of the pitfall trap. 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 (Fig. 54.7). Site selection favoring heavily used rhinoceros trails 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 rhinoceros areas, poor drainage results in flooding of the pit despite careful preventive measures. Interference from nontarget 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)!

Due to the dense nature of the rainforest environment and rare sighting of individual rhinoceroses therein, routine chemical capture techniques developed for Asian and African rhinoceros 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, Rhinoceros Protection Unit (RPU) and Sanctuary staff have been contacted, the following stepwise approach to capture and translocation is suggested (Radcliffe et al. 2004).

Guidelines for Capture of Displaced Sumatran Rhinoceroses

Secure Immediate Area In the event a wild Sumatran rhinoceros is found wandering outside a protected area, the first priority should 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 rhinoceros 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 rhinoceros relocation. If the rhinoceros were unharmed and close to a protected area (<10 km), then it may be desirable to move the rhinoceros without capture by pushing it back toward the forest. If the animal was injured or otherwise in need of medical attention or far (>10 km) from the forest, a decision should be made to capture the animal.

Make a Plan for Rhinoceros Capture Considering the high risks associated with capture by the "chase to exhaustion" method (i.e., rhinoceros is captured following an extensive stressful chase without the use of routine chemical capture methods), this approach

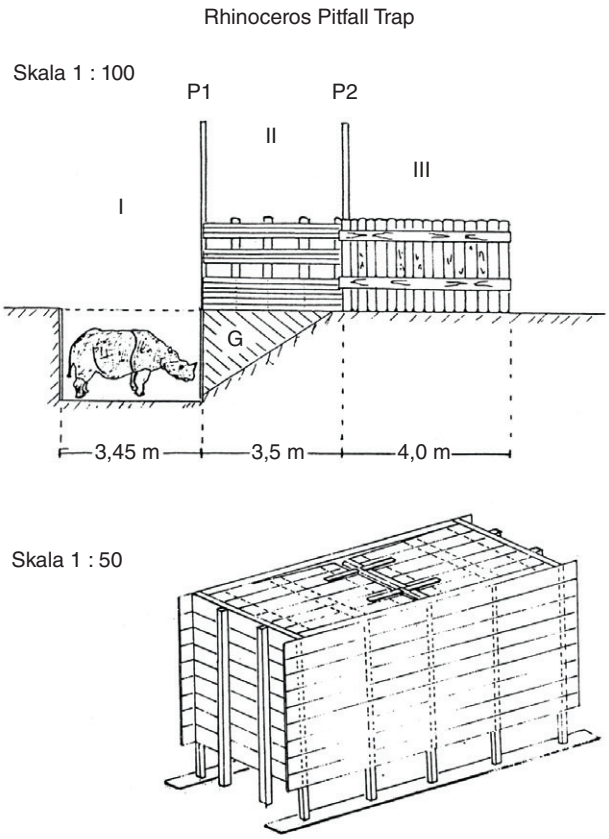

Figure 54.7. Diagrammatic sketch of pitfall capture method used in Indonesia's Riau Province for capture of wild Sumatran rhinoceros (*Dicerorhinus sumatrensis*) (sketches adapted from Sadmoko 1990; image courtesy of Mohd Khan bin Momin Khan, Malaysia Department of Wildlife and National Parks).

should only be attempted as a last resort (Fig. 54.8). The following are suggested guidelines and methodology for capture of at-risk Sumatran rhinoceroses outside a protected area.

Capture Method 1: 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 rhinoceros and surrounding area from a distance without pushing the animal to run as the rangers await the capture team. A rapid induction and recumbency will be essential for safe capture of a tropical ungulate species, such as the Sumatran rhinoceros, 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 rhinoceros and people alike (Table 54.2; Radcliffe et al. 2004). 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

Figure 54.8. Like the other rhinoceros species, the Sumatran rhinoceros (*Dicerorhinus sumatrensis*) is prone to capture myopathy. Here, a wild "hairy" rhinoceros 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 of Sugiyo, Wildlife Conservation Society, Indonesia Program).

5000 IU, respectively) or the newer etorphine-butorphanol-midazolam protocols, may be considered depending on the situation (Bush et al. 2011). If the rhinoceros 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 54.2). 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 2: Field Capture by Erecting Temporary Boma The Sumatran rhinoceros is perhaps the only species of rhinoceros 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 rhinoceros is in immediate peril, physical capture can be a feasible option. To begin, the animal can be followed from a safe distance and without excessive chasing until the rhinoceros is located within an area where it is resting and approachable (i.e., in water or other suitable location; Fig. 54.9). Large rolls of shade cloth or tarpaulin are then carefully erected without disturbance to form a temporary boma surrounding the rhinoceros that will facilitate 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 will 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 (Fig. 54.7).

Figure 54.9. 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 of Chandra Putra, Way Kambas National Park, Sumatra).

RHINOCEROS CRATING AND TRANSPORT

The moving or translocation of rhinoceroses is a specialized branch of rhinoceros anesthesia that has been practiced since the first African rhinoceroses were saved from the rising waters of Lake Kariba. The crating and relocation of rhinoceroses is now standard practice as urgent conservation measures, including enhanced animal monitoring and protected area management, have become effective tools in the fight against poaching (Flamand et al. 1984; Henwood 1989; Hitchens et al. 1972).

Walking a Rhinoceros

If a crate cannot be placed directly in front of the anesthetized animal, the rhinoceros can be "walked" a distance and guided into the crate (Fig. 54.10 and Table 54.3). When the rhinoceros becomes recumbent, the blindfold, cotton wool, head rope, and brake rope are applied. Four to 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 rhinoceros, clearing obstacles in its path. The rhinoceros is given small incremental doses of intravenous nalorphine or diprenorphine depending on species; doses vary, but as little as 20- to 25-mg nalorphine may be needed in total (Table 54.3; Kock et al. 2006). Alternatively, butorphanol can be given IV at 10 times the etorphine dose to walk a white rhinoceros into a crate. Regardless of antidote used, a prodder is

Figure 54.10. "Walking" an etorphine-immobilized white rhinoceros (*Ceratotherium simum*) using ropes and trained personnel to guide and stabilize the narcotized animal.

Table 54.3. Suggested opioid reversal protocols for walking, crate loading and transport of adult African rhinoceroses

Opioid Use for Crating and Translocation of African Rhinoceros

Method	Use	Reversal Drug or Opioid	Technique for Crating or Translocation
Butorphanol or nalorphine walking and crating method	For opiate (M99) immobilized rhinoceros	*White rhino* *Walking:* 10- to 20-mg BT per mg M99 Add 1- to 2-mg M50:50 for adult bulls for walking or crating (Hofmeyr) OR 1-mg M50:50 plus 20-mg nalorphine. Give further incremental 10–20 mg Nalorphine IV up to 75 mg (Kock et al. 2006) *Crating:* 10- to 20-mg BT per mg M99 OR Diprenorphine (M50:50) at 2.4× etorphine dose IV. Add 1- to 2-mg naltrexone to prevent pushing in crate. Can combine the naltrexone with diprenorphine—expect a relatively lively wake up so be properly prepared with head rope, position of crate, and so on! *Black rhino* *Walking:* Start with 5-mg nalorphine IV. Give incremental 5-mg doses every 5–10 minutes up to 20- to 40-mg (Kock et al. 2006) *Crating:* 15-mg butorphanol IV per mg M99 with rhino's head in crate door (Hofmeyr) Add 1- to 2-mg M50:50 to preclude pushing OR 10- to 20-mg nalorphine per mg etorphine. 1- to 2-mg M50:50 IV may be necessary to prevent pushing (Morkel) OR Blindfold with tight muslin cloth and wake up with 1- to 2-mg IV M50:50 *Avoid* myopathy in crating process by ensuring rhino does not squat in crate; use prodder plus repeat 1- to 2-mg M50:50 or 5-mg butorphanol doses	• Blindfold rhino; after cleaning eyes of mud or other debris, wrap a 4-m piece of muslin (mutton) cloth to cover eyes completely; attach three zip ties to secure muslin cloth forward of posterior horn. (*Note*: The blind fold option is really for black rhino and although it can be used on white rhino as well there is much less need for it.) • 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 8 people on head rope and 4 people on break rope. Can use a 4 × 4 pick-up truck for back-up on rope if not enough people for head rope • *Reverse*; *wait* 90–120 seconds. Use electric prodder or water in ear to stimulate rhino to stand if does not do so by itself • *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- to 1.2-mg diprenorphine iv; if white rhino pushes, give 1- to 2-mg naltrexone iv; use prodder on head or shoulders and not on hindquarters since rhino will tend to push/squat more with stimulation of hindquarters.

Butorphanol alone Crating method	For awake rhinoceros in zoo or boma environment	*White rhino:* 50-mg butorphanol IV (Radcliffe) *Black rhino:* 25- to 500-mg butorphanol IV at time of crating for conditioned animals (Radcliffe) Boma black rhinos: Start with 10-mg Butorphanol IV and increase in 5 mg increments (Hofmeyr)	• 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 • No reversal required for butorphanol once rhino is in crate
Diazepam: nalorphine Crating method	For field immobilized rhinoceros	*White and black rhino* 10- to 15-mg Diazepam IV 10 minutes before "waking" rhino with reversal protocol (Morkel) Use standard crating methodologies earlier for white and black rhinos after giving diazepam	• Give diazepam to recumbent rhino and wait 10 minutes • Use same crating procedure as earlier 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
Etorphine: azaperone Boma crating	For loading and transport of boma rhinos	*White rhino* *Boma crating:* 1- to 2.5-mg etorphine IM (higher dose for adult bull) plus butorphanol at 10X M99 dose plus 20-mg (subadult) or 40-mg (adult) azaperone *Note*: Butorphanol can also be given at time of recumbency rather than in the original dart, particularly if rhino requires medical care or other procedures under recumbency (Buss) *White or black rhino* 0.7- to 1.2-mg etorphine IM (Kock et al. 2006) OR 0.3- and 0.5-mg M99 for black and white rhino, respectively, without need to reverse *Crate sedation:* 0.05- to 0.15-mg etorphine IM plus 100- to 200-mg azaperone (can put in same syringe) IM or 10 to 30 mg diazepam IM (not in same syringe) *Transport:* 50- to 150-mg zuclopenthixol acetate IM	• 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 is the *only agent* to calm an excitable rhino inside a crate • Butorphanol is replacing nalorphine and diprenorphine for both respiratory support and arousal for crate loading in both white and black rhinoceros—use small incremental doses (5 mg or less) in black rhinos as reversal is more dramatic and produces a lively rhino • For crate sedation: Nalorphine sedation wears off ~5 hours post crating; thereafter give etorphine every 2 hours for duration of trip • If rhino is not excitable, give azaperone up to 200 mg per 6 hours • *Note*: More etorphine is not effective within 3–4 hours of diprenorphine use and 8–24 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

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

judiciously applied to the feet just above the nail, or to the muzzle or perineal area to get the rhinoceros to stand and keep it moving. After each dose, wait a few minutes (up to 10 minutes) and check the rhinoceros's response to the prodder or by squirting water in the ear. If there is no response, give another dose of antagonist. Once the rhinoceros 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 rhinoceros moves too fast, go slowly with the head rope and pull the brake rope to slow the moving rhinoceros. Particularly with young and fractious individuals, it is important to slow the rhinoceros as it approaches the crate so it does not traumatize itself upon entrance. A rhinoceros that charges too quickly into the crate can strike the far wall with such force to avulse the horn or crush the nasal bones. To preclude problems with loading, 10- to 15-mg intravenous diazepam 10 minutes before waking the rhinoceros helps to keep animals calm when aroused in the crate.

Black Rhinoceros Crating

The recent decline in availability and manufacture of the partial agonist-antagonist nalorphine in southern Africa has necessitated use of alternative techniques for partial reversal and crating of black rhinoceros in the field. Initial trials using similar agents, such as nalbuphine and butorphanol, have worked (sometimes quite well), although they have also been associated with irregular outcomes and responses in the black rhinoceros. Nalbuphine provides a satisfactory partial reversal that facilitates crating of black rhinoceros, but appears to predispose crated animals to dog-sitting and squatting that can lead to serious myopathy and inability to stand (Fig. 54.11). Butorphanol given in higher doses (25–30 mg for an adult animal) also provides a reliable partial reversal for crating of black rhinoceros. However, such animals seem prone to head pressing in the crate and require constant supervision.

Figure 54.11. Posttranslocation 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 of Birgit Kötting, Etosha Ecological Institute, Namibia).

A novel approach to crating the black rhinoceros currently practiced in Namibia combines diprenorphine together with methods to limit noise and visual stimulus during transport. A muslin (mutton) cloth works well for both white and black rhinoceros (Fig. 54.3). Animals remain remarkably tranquil, although some degree of chemical tranquilization is still necessary. The beauty of blindfolding is that a physical rather than a chemical means is used to calm the animal. The cloth must be placed with great care to avoid damage to the eyes or loss of the blindfold during crate transport. One must brush or blow any dirt from around the eyes and flush the eyes with saline, if necessary. Tying the blindfold properly takes two people—the secret is to start with a 4-m length of muslin cloth and place the middle point on the forehead directly behind the back horn. Both ends of the cloth are pulled tightly on either side of the head, making sure both eyes are closed in a normal manner. The cloth is wrapped under the jaw around the opposite side behind the back horn where it is tied securely. While wrapping, the cloth should be "spread" to cover the eye properly and hooked behind the jaw. Three heavy-duty cable ties 40 cm in length are used to secure the blindfold to the rhinoceros. Two ties are threaded through holes in the cloth fashioned above and forward of the eye and closed to form a loop. A third cable tie is threaded through the two loops in front of the back horn and pulled tight. The cable ties serve to pull the cloth forward securely over the eyes. If the cloth becomes loose during the journey, simply pull on a cable tie to tighten the blindfold.

Reversal and crating is then routine: a heavy hemp rope is secured around the head with blindfold and threaded through the front of the crate. This rope will provide the forward pull upon standing and will direct the rhinoceros into the forward part of the crate. A second rope is secured to one rear leg and is used as a "break rope" to simultaneously slow the momentum of the rhinoceros so it does not collide with the front of the crate, where horn or nasal trauma may result. A relatively high dose of diprenorphine (black rhinoceros adult, 1.5–2 mg; subadult, 1 mg) is given intravenously after positioning immediately in front of the crate door. Two minutes are allowed to pass while all staff is quiet and the animal undisturbed. Upon standing, the rhinoceros is pulled forward into the crate while the break rope on the back leg slows the rhinoceros. Once inside the crate, the rhinoceros is secured by sliding three pipes into place at the rear of the crate and the rear doors closed. This protocol consistently produces a lively and relatively awake rhinoceros inside the crate that remains calm because of the combined use of a secure blindfold and earplugs.

Rhinoceroses blindfolded with muslin cloth travel well and can be given other sedatives, including azaperone (60–100 mg) or additional opioids (etorphine or butorphanol) during transport as needed. From about 4 hours onward, an additional 0.05- to 0.1-mg etorphine (usually with about 60 mg azaperone) is administered, and is usually repeated every 2 hours. The veterinarian (or someone with a high level of experience with opiates) must remain with the animal for the entire trip to evaluate and top-up as needed. "Straightening" with a prodder applied lightly to the forehead, neck, or shoulder is often necessary in the first few hours. The mutton-cloth can be kept on for as much as 36 hours. It is essential to put the cloth on tightly and to make sure it is 100% clean and that no sand or dirt gets into the rhinoceros's eyes.

The beauty of the tight blindfold and blocked ears is that you can wake up the rhinoceros to a large degree (and therefore prevent pushing), but because the animal can't see or hear, it is very unresponsive and rarely gets excited—the effect is quite remarkable. However, the muslin cloth blindfold is inadequate by itself for crate transport, and additional tranquilization using repeated ultra low doses of etorphine and azaperone is essential along the road.

Tranquilization during Transport

All black rhinoceros require tranquilization during transport (even most crate-conditioned animals) to preclude excessive struggle and associated trauma (Table 54.3). Other rhinoceros species tolerate transport better than black rhinoceroses, but still often benefit from some sedation. The veterinarian must always travel with the rhinoceros 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 rhinoceros is alert and bouncing around, will risk unnecessary trauma to both animal and attendant. Additionally, a cool animal is generally more relaxed than an overheated one.

Rhinoceroses 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 excited if suddenly startled (i.e., from stopping, off-loading, etc.). The rhinoceros can be redosed 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. A 20-gauge, 1.5-in needle is inserted into the lateral muscles of the neck while avoiding the nuchal region. Beware of the head and horn during neck injections. For restless individuals, the gluteal region also works well. Once the rhinoceros has settled, attach the syringe and inject the drug. A pole-syringe can also be used, but hand injection is preferred because it precludes the startled response resulting from the jab of the pole. Beware of coring where the rhinoceros's skin may block the needle lumen. An intramuscular injection takes 5–10 minutes for first effect; for a faster response, an intravenous 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 rhinoceros's position and duration of recumbency. If the rhinoceros lies down while the vehicle is moving, the rocking and bouncing action of the truck helps to facilitate limb circulation. Beware, however, if the rhinoceros lies down for a long period (>60 minutes) in a stationary vehicle, unless you are very comfortable with its position. Rhinoceroses 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 or detomidine, and diazepam or midazolam, are useful agents to produce a calming effect in rhinoceroses during transport. Azaperone is the tranquilizing agent of choice at 100–250 mg per adult and can be repeated every six hours as needed (Kock et al. 2006; Rogers 1993a, 1993b). A forty mg per mL azaperone solution is a convenient preparation and mixes well with etorphine for intramuscular administration to a fractious, crated rhinoceros. The administration of opioids, either alone or in combination with intramuscular azaperone or diazepam, is the only effective way to preclude an excited black rhinoceros from traumatizing itself inside a crate (Table 54.3). Etorphine and azaperone (0.05–0.15 mg and 100–200 mg, respectively) are delivered by hand injection or pole-syringe with sedation achieved in 5–10 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 rhinoceros up to 400 mg) takes about an hour to provide sedative effects after administration, while perphenazine enanthate (Trilafon; 200–400 mg per adult) takes about 12 hours for first noticeable effects (Kock et al. 2006; Swan 1993; Table 54.3). Perphenazine works well for the translocation of black rhinoceros while caution should be exercised in white rhinoceros as its use has been implicated in anorexia (Kock et al. 2006; Portas 2004).

Black Rhinoceros Off-Loading from Crate

Black rhinoceros, and especially juveniles and subadults, are notorious for being aggressive and sometimes even self-destructive to themselves and the crating equipment (trucks, crates, etc.) at offloading sites. In Namibia, where rhinoceros are often moved from veldt to veldt without the use of bomas and

adaptation periods, the capture team uses a technique for off-loading that is effective and largely eliminates the aggressive phase. At the off-loading site, the rhinoceros is resedated inside the crate using a low dose of etorphine (adult, 0.1–0.2 mg) combined with azaperone (adult, 80–120 mg) and allowed to narcotize over a several minute period. Once the rhinoceros is head pressing or otherwise sedated, the muslin blindfold and earplugs are removed and the etorphine antagonist administered. A standard dose of 12–18 mg of diprenorphine for an adult animal is administered by intramuscular (not IV) injection and the crate doors are opened. As the rhinoceros regains first levels of awareness, it walks slowly from the crate even while still sedated and partially narcotized. These black rhinoceroses tend to walk directly away from the crate and into the veldt without the characteristic aggression and attack of the crate or related equipment. As the animal continues to walk away from the off-loading site, it becomes more fully aware of its surroundings and ambulates from the site in a normal fashion. This protocol has largely eliminated the aggression and self-trauma that is often characteristic of black rhinoceros at off-loading.

ALTERNATIVE RHINOCEROS ANESTHESIA TECHNIQUES

Antidote Choice

Following intravenous antidote administration, a rhinoceros will stand within 60–80 seconds. Response to the antidote is first noted 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 rhinoceros should always be moved into sternal recumbency before giving the antidote or it may *bash* its head on the ground as it attempts to rise from the lateral position. Intramuscular dosing of the antidote is often preferred for arousal of rhinoceros cow–calf combinations so that the pair awake slowly and have time to join together without dashing off in opposite directions. If intravenous dosing is desired with recovery of cow–calf pairs, the cow is injected first, thirty seconds before the calf.

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 20–50 times the etorphine mg dose and 90–100 times the carfentanil mg dose are considered standard for captive rhinoceros (Allen 1996; Kock et al. 2006; Swan 1993). Renarcotization has been reported in the white rhinoceros, but it is a rare occurrence in the black rhinoceros (Kock et al. 1990; Portas 2004). Field workers frequently use lower naltrexone doses (12.5 : 1 naltrexone to etorphine ratio) without a problem (Kock et al. 1995); however, sedative signs at these doses have been reported in white rhinoceroses and a minimum of 40 : 1 is therefore recommended to preclude renarcotization (Kock et al. 1995; Portas 2004; Rogers 1993a). While naltrexone is considered the agent of choice for complete reversal of narcotic anesthesia, a number of scenarios arise under both captive and field conditions where 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 rhinoceros are reversed into a crate with nalorphine, nalbuphine, or butorphanol, alone or in combination (Kock et al. 2006; Radcliffe & Morkel 2007: Table 54.2 and Table 54.3). In the boma, *Diceros bicornis* are reversed with naltrexone, although very nervous or aggressive individuals may benefit from reversal with diprenorphine for its sedative properties. *Diceros bicornis* are completely reversed in the field using naltrexone; however, because it is expensive, a combination of naltrexone and diprenorphine is often used for field reversal. In this case, the standard diprenorphine dose (2–2.5 times the etorphine dose; Swan 1993) is administered by intramuscular injection together with 50–100 mg of naltrexone.

In marked contrast to black rhinoceroses, white rhinoceroses are reversed into a crate using diprenorphine, with perhaps 1–2 mg of naltrexone. In the boma and in the field, *Ceratotherium simum* are similarly reversed with naltrexone. Diprenorphine is often used for translocation of *Ceratotherium simum*, since its partial agonist-antagonist actions provide significant narcosis during travel. However, diprenorphine has minimal agonist effects in *Diceros bicornis* and therefore should be used judiciously for transport in this species. For any partial antagonism in a crate situation, it is critical that the rhinoceros 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 rhinoceroses during travel.

Other Drugs and Immobilization Doses

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

- Carfentanil at 1–1.2 and 0.9 mg (captive adult white and black rhinoceros, respectively; Portas 2004; Rogers 1993a) and 3 mg for wild adult rhinoceros (De Vos 1978; Hofmeyr et al. 1975). 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 rhinoceros; Kock et al. 2006; Rogers 1993b).
- Fentanyl alone at 60 mg (black rhinoceros; Rogers 1993b).
- Thiafentanil can be mixed equally with etorphine. The adult rhinoceros 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 rhinoceros, the need may arise to deliver artificial ventilation. For emergency respiratory support in a rhinoceros, the animal is first pushed onto its side. A large person forces the knee and lower leg (with foot placed firmly on the ground) into the abdomen 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 intravenous 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 rhinoceros is ineffective and does nothing but fracture ribs and inflict unnecessary trauma.

Myopathies are common in rhinoceros that experience excessive chase periods or hyperthermia during capture. 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, 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 (Fig. 54.11). Use of the electric prodder on the head can often stimulate the animal to rise and stand, but avoid prodding the hindquarters as this can exacerbate the problem. If this does not work, consider prompt intravenous 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 rhinoceros develop an adverse reaction that the author refers to as the "fat nose syndrome" (Radcliffe & Morkel 2007). 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. This unusual response 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 who have led by exciting experimentation and hard-won experience (Harthoorn & Lock 1960; Kock et al. 2006; Player 1972; Young 1973). 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 et al. 2005). Various opioid receptor affinities and their pharmacologic 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, at least for rhinoceros capture specialists, the incorporation of mixed agonist-antagonist opioid cocktails as part of routine field capture methodologies for the African rhinoceros (Bush et al. 2005).

Recent work by Bush and colleagues in white rhinoceroses 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 54.2; Bush et al. 2005, 2011). 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 of etorphine and azaperone in the white rhinoceros. Butorphanol is a mixed opioid agonist-antagonist; its agonist κ receptor produces analgesia and marked sedation, while the weak μ receptor antagonism reduces respiratory depression and rigidity. The weak σ (non-opioid) 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. In black rhinoceroses, the butorphanol antagonism of μ-opiate actions will result in a lively rhinoceros not suitable for handling without additional sedation.

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, while blood pressures remain lower than with the standard pure opioid agonist protocols. Administering diprenorphine, a μ antagonist, intravenously 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. These discoveries may help to bring field rhinoceros capture into the realm of safety realized with captive animals where butorphanol-based protocols are now standard replacements for more potent opioids (Portas 2004; Radcliffe et al. 2000a).

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 (Gandolf et al. 2006; Langan et al. 2001; Radcliffe et al. 2000c). Due to 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 intravenous delivery or a slower induction by intramuscular administration, with both methods proving safe and effective for serial anesthesia (Gandolf et al. 2006; Table 54.4). The combination of the alpha-2 agonist, detomidine, along with the butorphanol was thought to enhance muscle relaxation and depth of anesthesia with intramuscular use in white rhinoceros calves. Complete reversal is achieved using naltrexone at four to five times the butorphanol mg dose and yohimbine HCl (Yobine) or atipamezole at 0.125 mg/kg for antagonism of the alpha-2 agent.

Cow and Calf Field Capture

Field immobilization of juvenile rhinoceros is not without inherent risk as calves may separate from their dams after darting or become recumbent at different times despite concurrent drug delivery (Fig. 54.12). Additionally, calves are more susceptible to capture stress, hyperthermia, and postcapture morbidity and mortality in boma situations (Kock et al. 1995). Translocation of cows with calves less than 18 months of age can be traumatic and is best avoided, while movement of very young calves 2–3 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 et al. 2006). The wild black rhinoceros cow is solitary by nature and usually retreats to a quiet spot to calve and will stay there for the first month afterwards. Therefore, if a black rhinoceros gives birth in a boma, she rarely manages to raise the calf.

Opioid doses lower than those reported for adult animals are utilized for juvenile rhinoceros, with subadults receiving approximately one-half the adult dose. For example, when combined with a tranquilizer subadult African rhinoceros (age ~2.5 years) should receive 1.75- to 2-mg etorphine while very young calves (age 2–3 months) can be immobilized with as little as 0.5–1 mg etorphine (Rogers 1993a, 1993b; Table 54.4). 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 rhinoceros calves run ahead of their mothers while black rhinoceros calves run close at their mothers' heels (Kock et al. 2006).

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, the cow is darted first, and about a minute, later the calf is darted (Kock et al. 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 rhinoceros must be avoided, and it is therefore mandatory to have experienced trackers as part of the ground team. When darting a cow-calf pair on foot, the calf will usually stay close to its immobilized mother. If approached carefully, the calf can be darted and will generally become recumbent close to its mother; note that black rhinoceros calves are skittish and run off more easily than white rhinoceros calves.

Wild subadult greater one-horned rhinoceroses have been immobilized using the same dosage as adult animals (2- to 2.5-mg etorphine plus 10-mg acepromazine; Dinerstein et al. 1990). However, subadult animals proved more difficult to capture and often evaded darting attempts by outrunning the trained elephants that are commonly utilized for field immobilization of greater one-horned rhinoceroses 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 et al. 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 tested the long-held belief—a conviction still strongly held by many since Durer's famous rhinoceros—that its skin was held together with rivets like a knights armor and impenetrable to any volley man could throw its way.

Table 54.4. Suggested doses for immobilization and anesthesia of rhinoceros calves in both *captive* and *wild* environments

	Captive Calves			Wild Calves		
Rhino Species	Protocol	Reversal	Reference and Comments	Protocol	Reversal	Reference and Comments
White rhinoceros	10- to 20-mg butorphanol (BT) IV for 66- to 159-kg calf (Dose 0.13–0.15 mg/kg IV)	Naltrexone at 5 mg per mg BT	Gandolf et al. (2006) Heavy sedation Light anesthesia Mild resedation noted 8 hours post-reversal in one calf	*Calf:* 0.1- to 1-mg etorphine (M99) *Juvenile:* 1- to 2.5-mg etorphine *Subadult:* 2.5- to 3.5-mg etorphine (*Note*: Above ranges represent calves of all sizes.) *Note*: All white rhinos, including calves, get IV butorphanol at 10–20× the mg M99 dose ASAP upon recumbency to provide respiratory support OR 1-mg M50:50 plus 10-mg Nalorphine IV	Diprenorphine at 2.5 mg IV per mg M99 for transport Naltrexone at 40 mg per mg M99	Kock et al. (2006) from SANP *Note*: Always dart mother rhino 30–60 seconds *before* calf (Primarily for black rhino who easily split. In the case of white rhino, the calf rarely leaves the mother's side so one can wait longer or until mother goes down before darting the calf)
	2.5- to 5-mg butorphanol + 1.5- to 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 et al. (2006) Surgical anesthesia	*Calf:* 0.1- to 1-mg etorphine + 5- to 20-mg azaperone *Subadult:* 2.5- to 3.5-mg etorphine + 30- to 60-mg azaperone	Diprenorphine at 3 mg IV per mg M99	Rogers (1993a)
Black rhinoceros	25-mg butorphanol IV for ~500 kg subadult calf	Naltrexone at 5 mg per mg BT	Radcliffe et al. (2000c) Heavy standing sedation	*Calf:* 0.1- to 1-mg etorphine *Subadult:* 2.5- to 3.5-mg etorphine *Calf:* 0.1- to 1-mg etorphine + 10- to 50-mg azaperone *Subadult:* 1.75- to 3.5-mg etorphine + 100-mg azaperone *Note*: Do not use the M50:50 plus nalorphine protocol in black rhinos as it will cause arousal *Instead*: 5-mg butorpahnol or nalorphine IV; titrate to effect	Naltrexone at 40 mg IV per mg M99 Diprenorphine at 3 mg IV per mg M99	Kock et al. (2006) from SANP Rogers (1993b) Kock et al. (2006) *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- to 1-mg etorphine + 5-mg acepromazine *Subadult:* 2- to 2.5-mg etorphine + 10-mg acepromazine	Diprenorphine at 2.5 mg IV per mg M99	Dinerstein et al. (1990) Same dose used for adult/subadult

Sources: Dinerstein E, Shrestha S, Mishra H. 1990. Capture, chemical immobilization, and radio-collar life for greater one-horned rhinoceros. *Wildlife Society Bulletin* 18(1):36–41; Atkinson MW, Bruce H, Gandolf AR, Blumer ES. 2002. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. *Journal of Zoo and Wildlife Medicine* 33(2):157–162; Gandolf AR, Wolf TM, Radcliffe RW. 2006. Serial chemical restraint for treatment of decubitus ulcers in two neonatal white rhinoceroses *(Ceratotherium simum)*. *Journal of Zoo and Wildlife Medicine* 37(3):387–392; Kock MD, Meltzer D, Burroughs R, eds. 2006. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services.

Figure 54.12. Anesthesia of rhinoceros calves is challenging, particularly under field conditions where darting of the cow–calf pair must be well coordinated in order to limit stress on both parent and offspring.

To the surprise of royalty and commoners alike, the rhinoceros quickly expired.

The future of the world's rhinoceroses will remain tenuous as human conflicts over shared resources escalate and rhinoceros horn continues to be cherished by traditional Asian societies for supposed unicorn-like mythical properties. Nevertheless, it is comforting to know that man—while solely responsible for the current crisis—is 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 rhinoceros—tailoring the dose for size, age, and condition of the animal. The rhinoceros should not be chased while the dart is being loaded. When darting from a helicopter, get the dart in quickly and back off until drugs considerably affect the rhinoceros.
- 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 subcutaneous 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 (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 to help sedate and calm the rhinoceros during transport.
- The addition of hyaluronidase, a hydrolytic enzyme that increases tissue permeability, greatly improves drug absorption and can markedly shorten the induction time.
- A lower opioid dose must be used for rhinoceroses 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 rhinoceroses.
- 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 rhinoceros is moving in a semi-narcotized state and thereby lessens the chance that the rhinoceros will injure itself by encountering a hazard. This is especially true when immobilizing rhinoceroses in rough terrain. A quick induction also limits the exertion and the physiological stress associated with increased body temperature, heart rate, oxygen consumption, and related physiologic changes. Caution must be used, however, as very rapid induction times are often associated with marked respiratory depression, especially in the more susceptible white rhinoceros.
- *Nalorphine (Nr), nalbuphine (Nb)*, and *butorphanol (B)* are useful in African rhinoceros (Table 54.2 and Table 54.3):
  - To *improve respiration*, give 5-mg *Nr* IV for black rhinoceros and 20- to 30-mg *Nr* for white rhinoc-

eros. Black rhinoceroses are very sensitive to *Nr* and *B*, so administer small incremental 5 mg doses given intravenously to effect. *Nb* and *B* may be used at approximately twice the *Nr* dose (20–40 mg) in a similar fashion for improving respiration in the white rhinoceros.

- To *walk a rhinoceros*, start with 10-mg *Nr* IV in black rhinoceros up to a total dosage of 20- to 40-mg *Nr* in 5 mg increments. For white rhinoceros standard practice is now to give *B* at 10× (up to 20×) the etorphine (M99) dose (10- to 20-mg *B* per mg M99). *B* can be incorporated into the initial dart or given at the time of loading. Others give 1-mg diprenorphine plus 20-mg *Nr* IV followed by small incremental doses of 10- to 20-mg *Nr* up to 75 mg.
- For *transport*, wake the black rhinoceros up into the crate with 10–20 mg of *Nr* IV per 1 mg of M99. May also need to give 1- to 2-mg diprenorphine IV if animal is pushing or collapsing in crate. Wake white rhinoceros with 10 to 20 mg of *B* per mg M99 or give diprenorphine at 2.4 times M99 dose (generally a "lively" wake up). Can also give 1–2 mg of naltrexone with diprenorphine or later in transport to prevent pushing in crate.

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