EAZA Best Practice Guidelines

Black rhinoceros (*Diceros bicornis*)

Picture: Black rhino at Chester Zoo

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EAZA Preamble

Right from the very beginning it has been the concern of EAZA and the EEPs to encourage and promote the highest possible standards for husbandry of zoo and aquarium animals. For this reason, quite early on, EAZA developed the “Minimum Standards for the Accommodation and Care of Animals in Zoos and Aquaria”. These standards lay down general principles of animal keeping, to which the members of EAZA feel themselves committed. Above and beyond this, some countries have defined regulatory minimum standards for the keeping of individual species regarding the size and furnishings of enclosures etc., which, according to the opinion of authors, should definitely be fulfilled before allowing such animals to be kept within the area of the jurisdiction of those countries. These minimum standards are intended to determine the borderline of acceptable animal welfare. It is not permitted to fall short of these standards. How difficult it is to determine the standards, however, can be seen in the fact that minimum standards vary from country to country. Above and beyond this, specialists of the EEPs and TAGs have undertaken the considerable task of laying down guidelines for keeping individual animal species. Whilst some aspects of husbandry reported in the guidelines will define minimum standards, in general, these guidelines are not to be understood as minimum requirements; they represent best practice. As such the EAZA Best Practice Guidelines for keeping animals intend rather to describe the desirable design of enclosures and prerequisites for animal keeping that are, according to the present state of knowledge, considered as being optimal for each species. They intend above all to indicate how enclosures should be designed and what conditions should be fulfilled for the optimal care of individual species.

Preamble

These Best Practice Guidelines were based on ‘concept husbandry guidelines for Black rhino (*Diceros bicornis*)’ which were produced by Valentijn Assenberg and Thijs van den Houten for the final thesis of their Animal Management course at the Van Hall Larenstein Institute. The data to form the concept husbandry guidelines was collected by a literature study and a questionnaire. The literature was chosen from a number of sources. A full reference list can be found at the end of this document. The questionnaire was partly based on the AZA husbandry manual and partly on the EAZA husbandry guidelines for the greater one-horned rhino. The AZA husbandry manual was published in 1996 and covers all five rhino species and is made with the help of the International Rhino Foundation. The EAZA husbandry guidelines for the Greater one-horned rhino were published in 2002 by Basel Zoo.

We are very grateful to Valentijn and Thijs for this report. Also to the following people who assisted by completing the questionnaire: Andreas Knieriem (Hannover Zoo) Gerd Nötzold (Leipzig Zoo) Jiri Hruby (Dvur Kralove Zoo) Robert Zingg (Zurich Zoo) Ulrike Cyrus (Zurich Zoo) Helen Massey (NEZS Chester Zoo) Xavier Vailliant (Pont-Scorff Zoo). I am also grateful to Tine Griede and Ineke de Jonge from the Van Hall Larenstein Institute for supervising the project.

A review of these Best Practice Guidelines was completed in 2013 by Becca Biddle and Dr Mark Pilgrim. For this we are especially grateful to Dr Andreas Ochs of Berlin Zoo for his review of the Veterinary Section, Marcus Clauss and Jürgen Hummel for their work on the Nutrition Section, and Dr Sue Walker and Dr Katie Edwards for their additions regarding Black rhino endocrinology and its applications to captive management.

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Introduction

Once plentiful across Africa, the Black rhinoceros *Diceros bicornis* is now classified as Critically Endangered by the IUCN (IUCN Redlist, 2012). During the last 60 years the Black rhino population has declined by almost 90% reaching a low of 2,410 individuals in 1995. Since then and until recently numbers have slowly increased. This has been due to concerted conservation efforts to protect rhinos from poaching and to metapopulation management including founding or enhancing populations through translocation. At the end of 2012 the estimated number of Black rhinos left in the world was 5,055 individuals.

Diceros bicornis is the only species within its genus; there are four distinct subspecies recognised by IUCN/SCC African Rhino Specialist Group; The European Association of Zoos and Aquaria (EAZA) is working with two of the four subspecies (Eastern and Southern) and is managing them as distinct subpopulations. A viable European captive programme exists for Eastern Black rhinoceros (*D. b. michaeli*) and a non-viable European captive population exists for Southern Black rhinoceros (*D. b. minor*) of 1.1 animals. These are planned to be repatriated to Africa. There are no South western (*D. b. bicornis*) in zoos nor are there any North western (*D. b. longipes*) Black rhinoceros, which recently became be extinct in the wild (IUCN 2011). Globally there are now 799 *D. b.michaeli,* making it the rarest of the three remaining Black rhino subspecies (IUCN 2012).

Due to the Critically Endangered status, Black rhinos in European Zoos are under the most intensive level of management, an EEP. The purpose of this programme is to secure a genetically healthy and sustainable captive population which may serve as a backup population for the wild. An international breeding programme was set up in 1966. This breeding programme contains an international studbook and manages the captive Black rhino population (Dollinger and Geser, 2008). The goals of this international breeding programme are self-sustaining reproduction, demographic security and stability, genetic diversity adequate for animal fitness and population adaptability and target population sizes sufficient to achieve these genetic and demographic goals (Foose and Wiese, 2006).

Worldwide there are 240 Black rhinos in zoos, including 64 in 15 EAZA zoos (2012) and another 15 in 2 non-EAZA zoo's. EAZA members have established Taxon Advisory Groups (TAGs) for different groups of animal species that are kept in zoos and aquariums. One of the main tasks of a TAG is to develop Regional Collection Plans that describe which species are recommended to be kept. The TAGs also identify which species need to be managed in a European breeding programme called EEP (European Endangered species Programme) (EAZA, 2008).

The mission of the EAZA Rhinoceros TAG is:

'*To ensure all captive populations are healthy, self-sustaining and genetically viable and are capable of being an effective tool in support of rhino conservation in the wild.*'

The goals of the EAZA Rhinoceros TAG are:

Population management

- To ensure each EEP population is self-sustaining and genetically viable in the long term.
- To ensure each taxon has ambitious targets for the retention of maximum gene diversity (~ 90% GD per century).
- To work more closely with other regions to support effective population management.
- To work to overcome obstacles which impinge upon population and genetic management goals; e.g. international transfers and importation of new founders.

Husbandry and welfare

- To ensure each EEP drives ongoing welfare and husbandry improvement.
- To ensure Best Practice Guidelines are in place for all EEPs by 2012 and reviewed at least every second year.
- To develop an audit process to ensure all holders are compliant with Best Practice Guidelines by 2015.
- To identify and support research priorities which advance husbandry and welfare and support the development of Best Practice Guidelines.

Education and research

- To ensure the captive populations provide a significant educational and research resource capable of contributing to rhino conservation.
- To recruit an education advisor to the TAG.
- To measure the impact of zoo based education specific to rhino conservation and assist in the improvement of zoo based education.
- To set up a research advisor team to the TAG.
- EEP coordinators to identify research priorities prioritising projects conceived to improve captive management, reproduction and welfare.
- EEP coordinators to collate research activities.
- Research advisor to report on activities and facilitate TAG wide research activities.

Section 1: Biology and field data

1.1 Taxonomy

1.1.1 Order

All rhinoceroses are placed in the order of Perissodactyla. Perissodactyla comes from the Greek word 'perissos', which means odd number and 'dactulos' meaning finger or toe in Greek (Huffman, 2007).

1.1.2 Family

The order Perissodactyla is comprised of three families; the Equidae (horses), the Tapiridae (Tapirs) and the Rhinocerotidae to which the rhinos belong (Nowak, 1999).

1.1.3 Genus

There are four genera of rhinos within the family. The Black rhino is placed in the genus *Diceros* (Nowak, 1999).

1.1.4 Species

The genus *Diceros* has one recent species, *Diceros bicornis*, the Black rhino which was first described by Gray in 1821 (Nowak, 1999). The name *Diceros bicornis* comes from Greek and Latin, *Diceros* from the Greek "di", meaning "two" and "ceros", meaning "horn" and *bicornis* from the Latin "bi", meaning "two" and "cornis", meaning "horn" (IRF, 2008).

1.1.5 Subspecies

There are four subspecies recognised within the Black rhino*;* the eastern ssp. *(*D.b. micheali*),* the south-western ssp. *(*D.b. bicornis*),* the south-central ssp*. (*D.b. minor*)* and the western ssp. *(*D.b. longipes*),* (Emslie, 1999) which has recently been reported extinct (IUCN 2011).

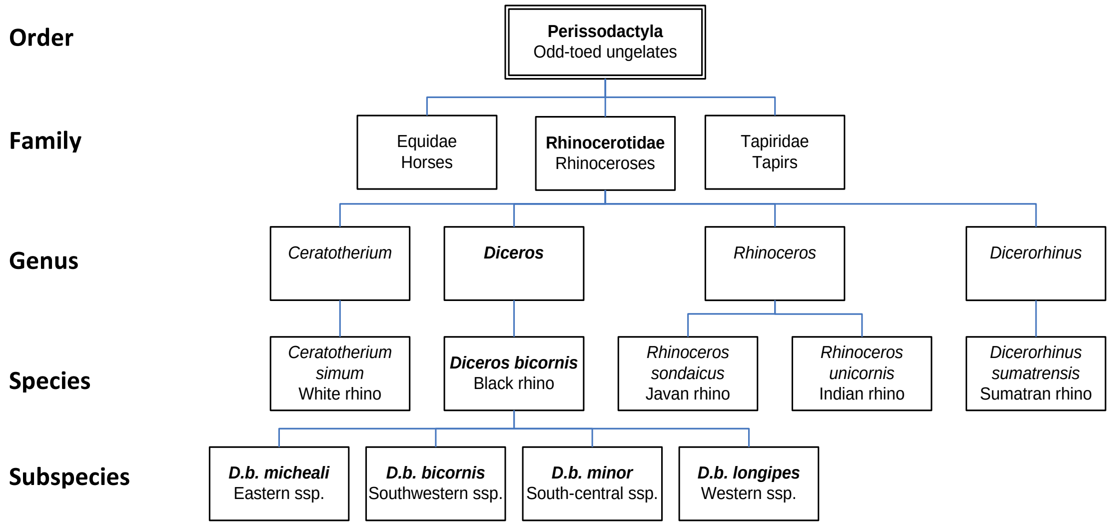

Figure 1.1. Classification of the Black rhino (*Diceros bicornis*) (Emslie, 1999; Nowak, 1999).

1.1.6 Common names

Black rhinos are actually not black at all. The name Black rhinoceros probably derives as a distinction from the White rhino (itself a misnomer), both species are grey. The White rhino having apparently derived its name from a variation of the early Cape Dutch word 'wijdt' meaning wide referring to the wide mouth of the White or Square-lipped rhino. Black rhinoceros can also refer to the dark-coloured local soil that often covers its skin after wallowing in the mud. Another common name for the Black rhino is prehensile or hook-lipped rhinoceros referring to the upper lip of the Black rhino which is adapted for feeding from trees and shrubs and it is its best distinguishing characteristic (Emslie, 1999; IRF, 2008).

Table 1.1: Translation of Black rhino into several European languages (Dollinger, 2008).

Languages	*Diceros bicornis*
Dutch	Puntlipneushoorn, zwarte neushoorn
English	Prehensile or hook-lipped rhino(ceros), Black rhino(ceros)
German	Spitzmaulnashorn
French	Rhinoceros noir
Spanish	Rinoceronte negro

1.2 Morphology

1.2.1 Body size

Adult Black rhinos have a body length of around 300 – 375 cm, and a height to the shoulder of approximately 80 – 140 cm. The weight of an adult Black rhino ranges between 800 and 1400 kg. Adult males are usually larger than females. The anterior horn is larger than the posterior horn; averaging about 50 cm in length. Sometimes the beginning of a third horn is present. The horns also differ between the sexes, with males tending to have chunkier horns and the females often longer and thinner ones. The longest recorded horn is 135.9 cm (Nowak, 1999).

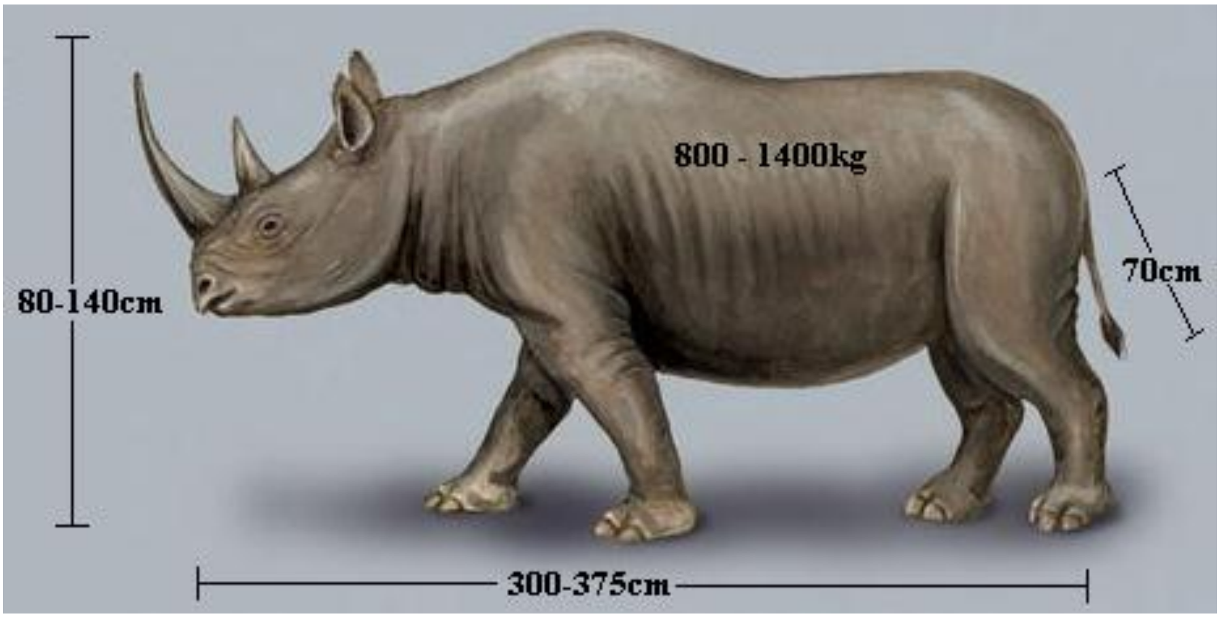

Figure 1.2 Body measurements Black rhino (Myers, 2006).

1.2.2 General description

The Black rhino’s skin colour varies between pale grey to dark brown to dark grey and is greatly influenced by the colour of the local mud in which it wallows. An external feature which more clearly distinguishes the Black rhino from the white rhino is the protruding prehensile upper lip (Nowak, 1999).

The dental formula of the Black rhino is; incisors: 0/0, canines: 0/0, pre-molars: 3/3 and molars 3/3 with a total of 24 teeth (Nowak, 1999). Figures 1.3 to 1.5 show the dentition of a Black rhino.

Figure 1.3: Black rhino skull taken from the right.

Figure 1.4: Upper jaw of a Black rhino.

Figure 1.5: Black rhino upper jaw molars.

Black rhinos have three toes with three stout nails, which leave impressions on the ground to the front and side of a softer wrinkled sole. The front feet are larger than the back feet (Adcock and Amin, 2006).

Black rhinos have two horns, which grow continually from the skin at their base throughout their life. The horn is continually worn away by rubbing. Each rhino develops its own rubbing habits and horn-wear patterns. Rhinos from different areas can have horns of different shapes (Adcock and Amin, 2006).

A Black rhino's sense of hearing is excellent, as is their sense of smell. These compensate for their poor eyesight which cannot easily detect an observer standing more than 30 m away. They can however detect movement at short distances (Adcock and Amin, 2006).

1.3 Physiology

The normal body temperature of a Black rhino ranges from 34.5 ^{o}C to 37.5 ^{o}C. The pulse is 30 to 40 beats per minute, and respirations are six to twelve breaths per minute (Fowler and Miller, 2003).

1.3.1 Horn

A rhino horn is comprised of thousands of compressed hair-like strands. The main component of the horn is keratin, making it extremely hard and tough, but it can be broken or split during fighting (Adcock and Amin, 2006).

1.3.2 Digestive system

The anatomy and digestive system in rhino species roughly resembles that of horses. Rhinos are monogastric animals with a hindgut-fermentation chamber. Microbial fermentation of plant fibre in the hindgut (cecum and large intestine) provides the main energy source for rhinos (Claus and Hatt, 2006). Figure 1.6 is a drawing of a rhino's digestive system.

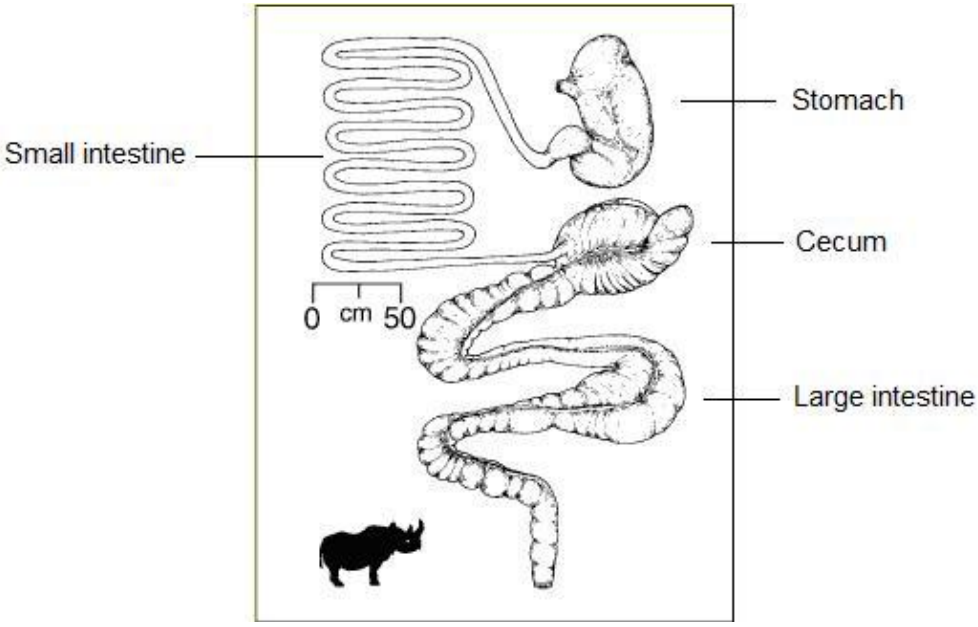

Figure 1.6: Black rhino digestive system (Stevenson and Hume, 1995).

1.3.3 Reproductive physiology – female

Female rhinos that have not bred have a hymen, the membrane that covers the vaginal opening which is often present cranial to the urethral opening. In several cases, a persistent hymen has been associated with a failure to breed, evidenced by the male's failure to achieve intromission after mounting. Rhinoceros have a long vagina characterised by longitudinal folds. These folds can make the opening of the cervix difficult to locate. The cervical canal is long and characterised by interdigitated folds. The uterus is bicornate, forming two horns after a short uterine body. The uterus of a pregnant rhino is characterised by diffuse placentation, meaning that almost the entire surface of the ventral outgrowth of the hindgut of the early embryo and the outermost membrane of the sac enclosing the foetus are used to form the placenta. The paired mammary glands are inguinal in position (Fowler and Millar, 2003).

1.3.4 Reproductive physiology – male

In the male Black rhino, the testicles are held close to the body along the preputial fold and are positioned horizontally as in the horse. The male reproductive tract includes vesicular glands, bulbourethral glands, and prostate. The relaxed penis is curved caudally, a position that results in the characteristic backward directed urination in male rhinos. The penis has notable horizontal flaps. Natural intromission may last up to 45 minutes (Fowler and Millar, 2003).

1.4 Longevity

In the wild Black rhinos can reach an age of 40 years. Black rhinos have the highest incidence among mammals of fatal interspecies fighting: almost 50 % of males and 33 % of females die from wounds. Fights between Black rhinos are usually for establishment and control of their territories. Why they are quite so aggressive is not known: in any event, rhino populations with high mortality rates recover only slowly. In captivity a male Black rhino has reached the age of 49 years (Felts, 2007; MacDonald, 2004; Nowak, 1999).

Field data

1.5 Zoogeography and ecology

1.5.1 Distribution

The Black rhino originally occurred throughout eastern and southern Africa and in the north ranged as far as north-eastern Sudan and at least as far west as north-eastern Nigeria (Figure 1.7). The extent of the former range in western Africa is not precisely known; suggestions are that prior to 1900 *Diceros* was found in the savannah zone as far west as Guinea (Nowak, 1999).

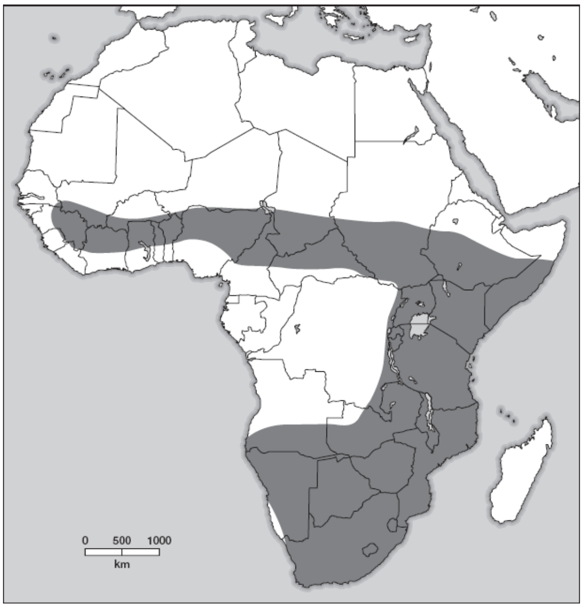

Figure 1.7: Probable distribution of Black rhino, circa 1700 (Emslie, 1999).

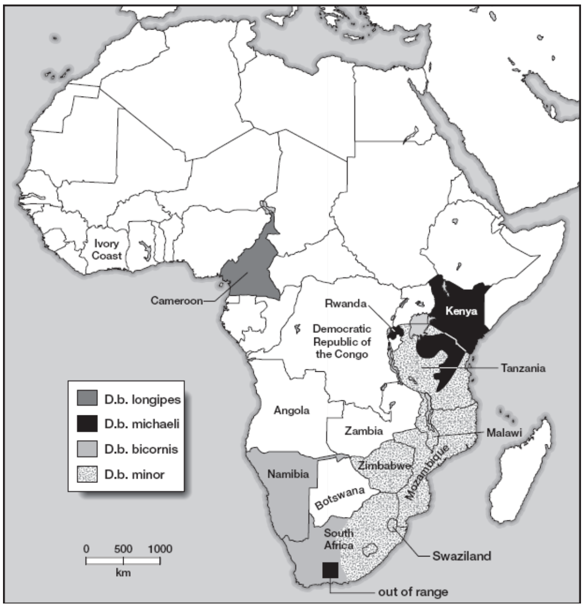

Figure 1.8: Distribution of the four Black rhino subspecies in 1997 (Emslie, 1999).

The Black rhino is divided into three extant and one recently extinct subspecies. The eastern ssp. (*D.b. micheali*) had a historical distribution from southern Sudan, Ethiopia, Somalia, and Kenya into northern-central Tanzania. The south-western ssp. (*D.b. bicornis*) had a historical distribution from Namibia, southern Angola, western Botswana and south western Africa. The original range for the south-central ssp. (*D.b. minor*) includes western and southern Tanzania, Zambia, Zimbabwe, Mozambique, northern and eastern parts of South Africa. It also probably occurred in southern Democratic Republic of the Congo, northern Angola and eastern Botswana. The western ssp. (*D.b. longipes*) recently reported to be extinct once ranged through west-central Africa *(Emslie, 1999).*

1.5.2 Habitat

The Black rhino is found mostly in the transitional zone between grassland and forest, generally in thick thorn bush or acacia scrub but also in more open country. It is not primarily a grassland animal but it favours the edges of thickets and extensive areas of short woody growth. Black rhinos exist wherever enough herbs and woody browse occurs in such sufficient amounts to support it. This spans a wide range of habitats covering deserts, semi-deserts, wooded savannahs, woodlands, forests and even sub-alpine heathlands. Black rhinos are restricted to habitat within about 25 km of permanent water (Nowak, 1999; Amin, 2006).

1.5.3 Population and conservation status

Early in the 19th century the Black rhino was the most numerous of all the rhino species with a total number in the hundreds of thousands. Due to poaching for their horn, numbers crashed to a low of 2,410 in 1995. Since then Black rhino numbers have been slowly increasing however well equipped, well organised crime syndicates have killed more than 2,050 African rhinos since 2010 (May 2013, taken from AfRSG, Traffic and CITES Rhino Working Group). South Africa alone lost 668 rhinos in 2012 (more than double the number in 2010) and trends suggest this number may be exceeded in 2013 (367 by May 2013). Minimum numbers of poached African Rhinos show a drastic increase in recent years, rising from 60 in 2006 to 745 in 2012. Most rhino horns leaving Africa are destined for Southeast Asian medicinal markets that are believed to be driving the poaching epidemic. In particular, Vietnamese nationals have been repeatedly implicated in rhino crimes in South Africa.

Despite these poaching losses, Black rhino numbers are up to 5,055 as of 31 Dec 2012 (from 4,240 in 2007). Even though this population growth is encouraging, unless the rapid escalation in poaching in recent years can be halted, African rhino numbers could once again start to decline. If the rate of increase in poaching seen 2011 / 2012 continues, modeling indicates that the tipping point when numbers start to decline could be reached as soon as 2015 (Emslie, IUCN / SSC AfRSG 2012).

In the year 2012 there were 799 eastern Black rhinos, 2,299 south-central Black rhinos and 1,957 south-western Black rhinos in the wild. The western Black rhino is extinct.

- The eastern Black rhino (*D.b. micheali*) is Critically Endangered, with a current stronghold in Kenya, with 631 rhinos as at the end of 2012. They live mostly within protected areas, (sanctuaries in both protected areas and on private land) and in free-ranging populations on county council land. Tanzania has *c. 100* eastern Black rhinos, mostly in free-ranging populations in unfenced protected areas and a few in sanctuaries. Rwanda and Ethiopia hold relict populations of one and two to four animals, in a protected area and on community land, respectively. At the end of 2012 South Africa had *c.* 68 eastern Black rhinos of predominantly Kenyan origin maintained on private land.

- Also categorised as Critically Endangered, the stronghold of the south-central Black rhino (*D.b. minor*) is South Africa and to a lesser extent Zimbabwe, with smaller numbers remaining in southern Tanzania. The south-central Black rhino is now thought to be extinct in Angola and Mozambique but small numbers have been reintroduced into Swaziland, Malawi and, more recently, Zambia and Botswana.

- Significant populations of the vulnerable south-western Black rhino (*D.b. bicornis*) have remained in the desert and arid savannah areas of Namibia and this country is the stronghold for the taxon, conserving 1,750 rhinos as at the end of 2012 with South Africa conserving a further 206 rhinos. There are no south-western Black rhinos in captivity.

- The population of the western Black rhino (*D.b. longipes*) was reduced to only a few scattered animals remaining in northern Cameroon with some animals believed to be seasonal visitors to Chad. The last extensive survey of possible rhino range in 2006 failed to find any rhino or signs of rhino, and there have been no sightings since 2006. This subspecies is now categorised as extinct (Amin, 2006; Emslie, 2006; IUCN, 2011).

1.5.4 Threats

The Black rhino faces a variety of threats. One of the main threats is poaching for the international rhino horn trade. Rhino horn has two main uses; use in traditional Chinese medicine, and ornamental use (for example, rhino horn is a highly prized material for making ornately carved handles for ceremonial daggers (Jambiyas) worn in some Middle East countries). During the 1960s civil unrest and the free flow of weapons in Africa had a significant impact on African rhino conservation efforts. Black rhino populations in Angola, Central African Republic, Chad, Mozambique, Namibia, Rwanda, Somalia, Sudan and Uganda have to varying degrees all suffered from the consequences of war and civil unrest since the 1960s. Some detrimental effects include trading of rhino horn and ivory for weapons, increased poaching due to increased poverty in times of civil unrest, and diminished levels of protection for rhino populations as funds are diverted away from wildlife departments. Habitat changes can also cause rhino populations to decline (African rhino specialist group, 2003).

Rhino horn has been an integral component of traditional Chinese medicine for thousands of years. It is ground down, mixed with water and ingested and believed to to be effective in reducing temperature, treating high fevers and convulsions, controlling haemorrhaging and assisting the liver in cleansing the blood of toxins resulting from the intake of alcohol or poison. Trade patterns detected by TRAFFIC indicate that the resurgent demand for rhino horn is driven primarily by users from Vietnam. Increasing prosperity in the Vietnamese economy has led to increased levels of individual disposable income and, sadly, use of rhino horn appears to be a way to demonstrate one’s affluence and high social status (Traffic 2012).

Threats to eastern Black rhinoceros: Some populations of the eastern Black rhinoceros in enclosed areas appear to be overstocked and are showing clear signs of density-dependent reductions in reproductive performance. In some cases competition from other browsers, such as African elephants *Loxodonta africana* and Giraffes *Giraffa camelopardalis,* appears to also be negatively affecting rhinoceros carrying capacity. Limited budgets for conservation are also a problem.

Threats to south-central Black rhinoceros: Conservative biological management appears to have limited metapopulation growth rates in some key populations. In parts of Zimbabwe, land transformation following re-settlement has negatively affected habitat in some areas and has resulted in a number of snare-related deaths. There is a plan to create an additional intensive-protection zone in Zimbabwe. Declining conservation budgets, an apparent increase in poaching and losses of animals to snaring, and the prosecution of rhinoceros offences under statues without deterrent sentences are of concern.

Threats to south-western Black rhinoceros: Illegal hunting has been blamed for the disappearance of the *s*outh-western Black rhinoceros from arid habitats in at least two range states (Angola and Botswana). Since 1979 conservation efforts in Namibia have stemmed poaching activities and the population has increased steadily. As in other range states, declining budgets for conservation are a problem.

Threats to Western Black rhinoceros: Poaching, lack of finance, limited anti-poaching efforts, limited local capacity for conservation management, failure of courts to give sentences that can act as a deterrent to potential poachers and genetic / demographic factors all pose serious threats to this subspecies.

1.5.5 Conservation actions

The Black rhino is, as a full species, classified as Critically Endangered by the IUCN. The south-central, eastern and western subspecies are also classified as Critically Endangered. The south-western subspecies is classified as vulnerable (IUCN 2012).

Black rhinos have been listed on CITES Appendix I since 1977. All international commercial trade in Black rhinos and their products have been prohibited. To help reduce illegal trade and complement CITES international trade bans, domestic anti-trade measures and legislation were implemented in the 1990s by a number of consumer states. Effective field protection of rhino populations has been critical. Many remaining rhinos are now concentrated in fenced sanctuaries, conservancies, rhino conservation areas and intensive protection zones where law enforcement effort can be concentrated at effective levels. Monitoring has also provided information to guide biological management decision-making aimed at managing rhino populations for rapid population growth. This has resulted in surplus animals being translocated to set up new populations both within and outside the species' former range. Following a decline in breeding performance in some areas, increased effort has recently been given to improving biological management with a view to increasing metapopulation growth rates. Increasing efforts are also being made to integrate local communities into conservation efforts. Strategically, Black rhinos are now managed by a range of different stakeholders (private sector and state) in a number of countries increasing their long term security. In contrast to southern White rhino, most Black rhino on privately owned land are managed on a custodianship basis for the state. In addition to local and national initiatives, there are a number of regional African rhino conservation initiatives: the South African Development Community (SADC) Regional Programme for Rhino Conservation, the SADC Rhino Management Group, the SADC Rhino Recovery Group, and the Southern African Rhino and Elephant Security Group. IUCN SSC's African Rhino Specialist Group is the continental coordinating body for rhino conservation in Africa (African rhino specialist group, 2003).

1.6 Diet and feeding behaviour

1.6.1 Food preference

The Black rhino is a browser, its main foods being the thin regenerating twigs of woody growth and legumes. A great variety of plant species are utilised, although acacia seems to be a favourite (Nowak, 1999). The natural diet of the Black rhinoceros is characterised by a high fibre and moderate to high protein content (Claus and Hatt, 2006). They eat a wide range of browse species in any given habitat, but while over 100 species may be ingested during a year's foraging, 90% of the diet is commonly made up from fewer than 20 species. Grass is generally only eaten incidentally while foraging for low-growing herbs, but new soft grass leaf growth is voluntarily taken (Adcock and Amin, 2006).

1.6.2 Feeding

Twigs are gathered with the prehensile upper lip, drawn into the mouth, and snapped off with the premolars. Drinking occurs every day if water can be reached and mineral licks are visited regularly (Nowak, 1999). Black rhinos are most active during the night-time when most of their foraging and drinking is done. Foraging also occurs in the cooler hours of the morning and afternoon, but wallowing and / or sleeping in a cool, breezy or shady spot is the main activity during the heat of the day (Adcock and Amin, 2006).

1.7 Reproduction

The sex determination system of XX / XY is applicable on Black rhino, where the males have XY and females have XX chromosomes. Black rhinos have a total of 84 chromosomes (Trifonov, 2003).

1.7.1 Sexual maturity

In wild males sexual maturity is reached between seven to nine years and in wild females at between four to six years. The first parturition of females in the wild is estimated around five years, but in some populations it might take up to twelve years. Research found that in two South African subpopulations of Black rhino, in which there were 0.1 / km^2, that in these two subpopulations females reached sexual maturity at a younger age then in a third subpopulation with a Black rhino density of 0.7 / km^2 (Adcock and Amin, 2006; Carlstead et al, 1999; Nowak, 1999).

Records from captive Black rhinos in the EEP show the youngest male at first reproduction was three years old. However, more generally (the youngest ten males at first reproduction, out of the total past EEP dataset) range between four and five years old. The average age at first reproduction is ten years and one month.

The youngest female at first reproduction was five years old, the youngest ten females at first reproduction ranged between five and six years old. The average age for a female at first reproduction is ten years and four months.

1.7.2 Seasonality of cycling

The recurring of the cycle in regular intervals is more important than the length of the cycle in days. Oestrous cycling begins while the mother is still nursing. Generally, breeding occurs throughout the year; however there may be mating peaks in some areas. In Kenya mating peaks occur during September till November and during March till April. In Zululand mating peaks are during October till November and during April till July. These indications suggest that most births take place in the rainy season (EAZA yearbook, 1995; Hutchins, 2003; Nowak, 1999).

Irregular oestrous cyclicity patterns are relatively common among captive black rhinos (see section *1.7.3 Reproductive cyclicity in females*), and in many females behavioural expression of oestrus can also be difficult to detect. It is a recommended that collections use hormone analysis as a management tool as this can allow the prediction of when a female will be receptive to the male based on her hormone profile, giving keepers extra information to help introduce the rhinos at the right time. Furthermore, once pregnancies have been confirmed, this allows the male to be separated and mixed with another female at an earlier stage of gestation.

As oestrous cycle length can be highly variable both between females and within an individual female over time, long-term sample collection is the best way to make accurate predictions. At Chester Zoo, since the initiation of the endocrine programme there have been six pregnancies and five births in six years, highlighting the impact this additional tool can have on facilitating breeding management.

1.7.3 Reproductive cyclicity in females

Black rhinos are polyoestrous, meaning that they will come into oestrus, i.e. the time of receptivity to the male, on multiple occasions throughout the year. Although there is some evidence of seasonality of births in the wild due to weather conditions (Hitchins and Anderson 1983), and perhaps in zoos due to management constraints, there is no evidence of seasonality in oestrous cycles.

The oestrous cycle has previously been characterised in this species using steroid hormone analyses, either using blood (Berkeley *et al.* 1997), urine (Hindle *et al.* 1992) or faeces (Brown *et al.* 2001; Edwards et al. 2013), and saliva could potentially also be used as a sampling medium. Changes in the ovary during the oestrous cycle have also been previously described using ultrasound (Radcliffe *et al.* 2001).

An average oestrous cycle length of the Black rhino has previously been described as around 26 days in length; however, research on wild (Garnier *et al* 2002), and captive rhinos in America (Brown *et al.* 2001), and more recently in Europe (Edwards *et al.* 2013) have revealed that oestrous cycles are more variable in length, with typical oestrous cycles lasting between 20 - 40 days. In addition, short (< 20 days), and long (> 40 days) cycles, and acyclic periods with no evidence of oestrous cyclicity are also relatively common. The causes of these different cycle types are yet to be fully understood, but early indications suggest that these may not reflect normal reproductive function, and to-date pregnancies have only been reported associated with 20 - 40 day cycle types. Therefore oestrous cycles of 20 - 40 days in length are considered to be normal, and although females may vary in their typical cycle length, within an individual cycle length tends to be relatively consistent. Indeed, the regularity of oestrus on an approximately monthly basis is perhaps more important than the exact length of the oestrous cycle.

Females in captivity may commence oestrous cyclicity between three and four years of age, with one female age three years eight months of age exhibiting clear oestrous cyclicity (Edwards *et al.* 2013). The oldest female to have reproduced in captivity was aged 32 years, but females may continue to cycle after this time, and in the wild have been reported to continue producing offspring after 30 - 35 years.

1.7.4 Reproductive hormones in males

Testosterone can be measured in blood (Christensen *et al.* 2009) and faeces (Brown *et al.* 2001; Edwards *et al*. 2013), and can be used as an indicator of reproductive function. Testosterone concentration increases with age (Edwards *et al.* 2013), and may also vary according to prior reproductive success (Edwards et al. 2013), or due to the sociosexual environment (Christensen *et al.* 2009).

1.7.5 Gestation period / birth rate

The gestation period is around 15 to 16 months or 440 to 460 days. The inter-birth period in the wild is somewhere around 27 months. The inter-birth period in captivity is 40 months. This may be due to delayed reintroduction to a mate postpartum (Carlstead et al, 1999; EAZA yearbook, 1995; Hutchins, 2003).

1.7.6 Birth

After the gestation period there is a single calf born, which weighs around 40 kg. Birth usually takes place in the early morning (*Nowak, 1999*).

1.7.7 Development

Nursing generally continues for over one year, and the older calf is driven away by the mother around the time that the next offspring is due. Some solid food may be taken within a few weeks of birth, weaning is completed after about two years; independence is achieved between two and a half and three and a half years old (Hutchins, 2003; Nowak, 1999).

1.8 Behaviour

The Black rhino is unpredictable in its behaviour and can be a dangerous animal, sometimes charging a disturbing sound or smell. It has tossed people in the air with the front horn and regularly charges vehicles and campfires. If a Black rhino catches the scent of humans, it usually runs away, sometimes for quite a distance before stopping (Nowak, 1999).

1.8.1 Activity

Black rhinos are more active (feeding, drinking and walking) in early morning and late afternoon to evening. Black rhinos are also active at night, often feeding, drinking, and walking outside their core areas and in more open habitat than during the day. The most intensive feeding takes place during early morning and evening.

Sleeping occurs either standing or lying down. Black rhino react swiftly when disturbed from rest, usually standing up and facing the source of disturbance. Because they have poor eyesight they may not locate the disturbance easily. Being curious animals, they will walk or trot forward to find out what is going on.

Black rhino usually run away if they catch a human's scent and only charge if they feel threatened. Black rhinos frequently wallow in shallow water holes. The water helps them to keep cool, they coat themselves in mud, probably to gain a protective coating against biting insects.

The horn is continually worn away by rubbing. Each rhino develops its own rubbing habits and horn-wear patterns. Rhinos from different areas can have horns with different shapes (Adcock and Amin, 2006; Jansa, 1999; Massicot, 2007; Nowak, 1999).

1.8.2 Locomotion

Black rhino can move extremely fast. They can run at 55 kilometres per hour change direction surprisingly quickly. They can run right through scrub and bushes (Adcock and Admin, 2006; Nowak, 1999).

1.8.3 Predation

Adult Black rhinos have no predators, although lions, leopards and hyenas may kill calves and sub-adults. Evidence of predator attacks are sometimes seen in the form of mutilated ears or missing tails. According to Brain, Forge and Erb (1999) sub-adult Black rhinos of a certain age appear particularly susceptible to lion predation in Etosha National Park. The sub-adults at this age have just left their mothers and are still relatively small. Brian, Forge and Erb (1999) also report that over a 13 year period in the Hluhlwe / Corridor / Umfolozi game reserve complex, there were no records of lion predation on Black rhinos, although there was strong evidence to suggest that there was spotted hyena (*Crocuta crocuta*) predation on small calves. It is estimated to be a 16 % loss of rhinos less than two years old to predation. Black rhinos are also capable of killing their predators. Reports vary from females with calves killing lions and a sub-adult female killing an adult hyena (Adcock and Amin, 2006; Brian, Forge and Erb, 1999; Law and Myers, 2004).

1.8.4 Social behaviour

Group structure: Black rhinos are predominantly solitary, the most commonly observed group structures being adult females with young. Other groups of various ages and genders occur, but they are usually temporary. The largest temporary group reported in one study included 13 Black rhinos. Recent studies indicate that Black rhinos are more social than previously thought and particularly around waterholes at night. Females are usually are found together with a calf and sometimes an older daughter. Females without young may temporarily join a neighbouring female. Sub-adults frequently associate with other Black rhinos. Only fully adult males become solitary, and even then they may form temporary groups that move and feed together. Male Black rhinos only become socially mature when they establish a set territory, in which they spend most of their time and do most of their feeding. Females settle into their own home range near the time of birth of their first calf. Female home ranges can overlap. Dominant bulls do not overlap with their home ranges (Adcock and Amin, 2006).

Relations and communication: Adult male Black rhino tend to live on their own, except when courting females. Among males, there are dominant and subordinate animals. Subordinate rhino are often sub-adults who must defer to an established territorial bull or risk a fight. Young bulls are often killed or injured in these interactions. Old males which can no longer defend their territories also die in fights, or become confined to a small area (Adcock and Amin, 2006; Massicot, 2007).

Black rhinos that share a part or all of their home range exhibit a familiarity with one another instead of the aggression that they exhibit to total strangers. Black rhino advertise their presence in their range to other rhino by spray-urinating and scraping their dung on the ground next to a path; and also by defecation on well-developed dung-piles. Male rhino spray-urinate and scrape more than females, and territorial (dominant) males keep more dung-piles in and around their range (Adcock and Amin, 2006; Nowak, 1999).

The explosive puffing snort of an alarmed Black rhino is the sound most clearly associated with this species by people who work with them. An appealing high-pitched whine or squeal is another sound made by this species. The high-pitched whine is used by calves to attract its mother’s attention, a male may use it to court a female, and all Black rhinos use it when in pain or in distress (Adcock and Amin, 2006).

1.8.5 Sexual behaviour

An adult male and female, with the female’s young if she has one, form temporary associations for mating during the female’s oestrus. For a few days, when the female is in oestrus, a pre-mating bond may develop between the bull and the cow, and the pair remains together during resting and feeding. They even sleep in contact with each other. Young are sometimes attacked by males during courtship. The young return to the female when the oestrus is over. Although at times several bulls may court a female simultaneously without apparent antagonism, serious fights and frequent deaths result from conflicts over females in oestrus. Rhinos are renowned for the extended duration of copulations, which last between 20 minutes and an hour or longer, with multiple ejaculations (Adcock and Amin, 2006; Hutchins, 2003; Massicot, 2007; Nowak, 1999).

Section 2: Zoo management

Introduction

This section suggests best practice management of Black rhino in the zoo environment. This topic is divided into the following chapters: Enclosure, Feeding, Social structure, Breeding, Behavioural Enrichment, Handling and Veterinary. The information for this has come from three main sources. Firstly, the opinions of experienced Black rhino managers were sought via a questionnaire and their opinions integrated into the first draft guidelines. These were: Andreas Knieriem (Hannover Zoo) Gerd Nötzold (Leipzig Zoo) Jiri Hruby (Dvur Kralove Zoo) Robert Zingg (Zurich Zoo) Ulrike Cyrus (Zurich Zoo) Helen Massey (NEZS Chester Zoo) and Xavier Vailliant (Pont-Scorff Zoo). Secondly, information was shared and compiled as a result of the discussions at a Black rhino husbandry workshop held in Doué le Fontaine, France in May 2010. Thirdly, information has been included from the AZA Rhinoceros Husbandry Resource Manual (Fouraker, M. and Wagener, T. 1996) additionally some other relevant sources of information have been used and these are referenced in the text.

A review of these Best Practice Guidelines was completed in 2013 by Becca Biddle and Dr Mark Pilgrim. For this we are especially grateful to Dr Andreas Ochs of Berlin Zoo for his review of the Veterinary Section, Marcus Clauss and Jürgen Hummel for their work on the Nutrition Section, and Dr Sue Walker and Dr Katie Edwards for their additions regarding Black rhino endocrinology and its applications to captive management. Many thanks to all involved.

2 Enclosure

In the temperate European climate Black rhino require indoor as well as outdoor facilities and each of these facilities is described separately. When designing an enclosure it is important that the zoo environment resembles the natural environment as closely as possible to maximise rhino health and reproductive success, when talking about indoor enclosures this may refer to temperature and lighting.

It is recommended that new holders plan for 2.3 rhinos. However if necessary this may be phased, with an initial build for a minimum of 1.2 rhinos and a commitment to be able to house 2.3 within five years.

2.1 Indoor enclosure

2.1.1 Indoor boundary

The recommendation is to build for 2.3 animals, with a minimum of six indoor enclosures. If the building is being phased the initial build must be for at least 1.2 animals with a minimum of four indoor enclosures. These indoor enclosures should be adjacent to and interconnected with each other allowing flexibility to combine enclosures to create fewer larger areas or split them to provide individual areas for each of the three adults and potential calves / juvenile. Adjacent enclosures should be accessible through at least two gates, giving the animals the possibility to roam without danger of being trapped (EAZA yearbook, 1995).

At least one of these areas should be suitable for the isolation of an animal (e.g. new animals or sick animals). This area should be off show to zoo visitors, protected from disturbance (such as noise) and should not allow direct contact between animals.

The boundary between the indoor enclosures separating the rhinos from each other should generally be solid walls with an area in the boundary to allow visual and limited physical contact between the animals. These have proved very useful in getting individuals used to each other prior to mixing and to allow keepers to better evaluate when animals are in oestrus using behavioural cues. The isolation pen should be of all solid walls and not allow contact between the animals.

The boundary between the rhinos and the keepers may be solid or bars. Bars have the advantage of giving the keepers an opportunity to habituate the animals to be touched all over their bodies. This facilitates health examinations and veterinary treatments, provides good opportunities for operant conditioning (training) and also promotes a good bond between keepers and rhinos (Figure 2).

Facilities should be designed so that each rhino can be checked over or trained on a daily basis should this be required. It is preferable that these training or examination areas are inside or undercover, however some collections do make use of outdoor training areas (either in addition or instead of indoor areas) to good effect (see *sections 2.7 and 2.8 on Handling and Veterinary 2.7*).

Figure 2: Indoor barrier between rhinos and keepers that allows keepers close contact to the rhinos. Note the low horizontal barrier that prevents a young calf passing through the bars.

Walls are usually made of concrete, concrete clad with wood, or wood of a suitable strength. Wood cladding on the concrete walls may help to prevent the rhinos from excessively rubbing their horns.

Bars are usually vertical, however horizontal and diagonal steel may also be used with great care taken to ensure that the bars are far enough apart to reduce the chance of rhinos trapping their horns and damaging them. Additional barriers may need to be required at low level when housing young calves to prevent them passing through. Vertical bars should be spaced 25 - 30 cm apart (Fouraker and Wagener, 1996).

Enclosure gates will often be the weakest points of the exhibit and therefore adequate hinge and lock strengths are very important. Interior doors are usually constructed of heavy-gauge galvanized steel or pipe that is hinged or sliding. Manual sliding gates are preferred to swing gates or hydraulic gates both for their speed of closure and to reduce the chance of keepers getting trapped or injured. Gates should be constructed to allow keepers to open and close them without entering rhino space. It is also important that keepers have good visibility either side of the gates in order to operate them safely. When sliding gates are used, the track must be kept clean in order to reduce the chance of them seizing, and care should be taken in the construction of the track to avoid injuring the feet of the animals as they run through gates during introductions. All gates must be firmly secured and designed so the animals cannot dislodge them with their horns, i.e. constructed in a way that the gate cannot be lifted off its hinges by a rhino.

2.1.2.Indoor substrate

Brushed concrete is most commonly used as the indoor substrate however tiles and rubber coated concrete may also be used. It is recommended that animals have access to both hard and softer substrates in their enclosure; these could be either inside or outside. If sand or other loose substrates are used, avoid feeding on these as this may promote accidental ingestion of the substrate and subsequent digestive problems.

2.1.3.Indoor furnishing and maintenance

Some collections provide the animals with straw bedding (Figure 2.1). Rhino urine can be sticky and corrosive and therefore enclosure furniture should be able to be scrubbed clean. No delicate equipment should be stored within spraying distance of the rhinos.

Figure 2.1: Straw bedding on a brushed concrete floor.

Indoor enclosures should be scrubbed clean with water on a daily basis. Cleaning products such as disinfectants with a strong smell should be avoided for routine cleaning as these are disliked by rhinos (Carlstead et al 1999).

Wooden rubbing posts should also be available. Horizontal pieces of wood have proved popular in one institution as they allow the rhinos to mimic the behaviour seen in wild rhinos where animals rub both the inside and outside curvatures of the horn. Availability of these horizontal wooden bars seems to promote more similar shaping of the horn to that seen in the wild.

2.1.4.Indoor environment

An indoor temperature of 18 - 20^{o}C should be maintained. Indoor temperatures of greater than 30^{o}C should be avoided. Various heating methods have been used successfully including radiators, hot water pipes, radiant panel heaters, hot air blowers and under-floor heating. If under-floor heating is used it is advisable for it to be installed in the area of the enclosure most likely to be used for sleeping, but not throughout the whole facility. This provides the animals with a thermal gradient and gives them a degree of choice as to where they are most comfortable sleeping. Some institutions have reported increased dust and ammonia fumes in houses where under-floor heating is used, so increased ventilation may be required to compensate. Indoor facilities should be maintained with good ventilation while avoiding draughts (EAZA yearbook, 1995).

Some institutions have used heavy duty plastic hanging strips on the doorways allowing animals free access to both indoors and outdoors whilst minimising heat loss and draughts. Animals can be habituated to these by installing one strip at a time. Most animals accept them within two - three weeks.

Thermal imaging cameras can be a useful tool in assessing surface temperatures in different parts of the indoor enclosure and identifying and eliminating cold spots. This is particularly useful when preparing an enclosure for a winter calving. One institution has experienced hypothermia in a newborn calf due to it lying on a cold floor. Use of deep straw (30 - 40cm) to cover the floor around the time of birth and during calf rearing is another method of reducing cold spots and recommended by one highly successful institution.

Natural daylight cycles seem to be adequate for rhinos. However, if an animal is to be held indoors for more than twelve hours (e.g. during winter in cold-climate institutions), facilities should provide artificial or natural light sources to stimulate natural cycles. Skylights or windows providing natural light should be included whenever possible.

Water should be available in each indoor area. Both standing water and wall mounted self-filling water troughs have been used. In order to avoid drowning when young calves are present, any water troughs set into the floor should either be replaced with one off the ground or drained of all but a few centimeters of water.

2.1.5.Indoor dimensions

It is recommended that in northern and central Europe where rhinos are likely to spend a great deal of time indoors, indoor enclosures should be no less than 60 m^2. In southern climates and if the animals have access to outdoor areas, the indoor pens can be smaller, but never less than 30 m^2.

2.2. Outdoor enclosure

2.2.1 Outdoor boundary

At least five outside areas are required for 2.3 animals, or three outside areas for 1.2 animals. These areas should be adjacent to and interconnected with each other allowing flexibility to combine enclosures to create fewer larger areas. The area used for mixing rhinos should offer multiple escape routes for the animals and good observation points for the keepers so that the animals can be separated quickly and safely if required.

A variety of methods have been used as primary barriers, including horizontal metal rails (Figure 2.2), vertical posts of either metal or wood and solid walls of concrete, masonry or stone (Figure 2.3). Black rhinos may climb and a primary barrier should be a minimum of 1.5m (5ft) high and non-climbable. There have been a couple of reports of particularly adventurous individuals getting just their front legs over horizontal rail fences of this height. Care should also be taken that animals cannot move items of cage furniture (such as logs) up to the fence line and use these as steps to climb over.

Posts connected by horizontal heavy gauge wire (hawser wire) have also been used however care should be taken with these as some rhinos will use them to rub their horns leading to the development of deep groove or even amputation of the horn. This behaviour may be reduced if plenty of alternative horn rubbing opportunities are available.

Figure 2.2: An example of a horizontal metal rail barrier.

Figure 2.3: A 2.0 meter high concrete barrier on a gently sloping ditch.

Both dry and water filled moats have also been used. Dry moats have a preference above water filled moats because of danger of drowning, especially if young calves are kept in the enclosure. Where dry moats are used, steep drops should be avoided as there have been cases of animals falling or being pushed into them. Ditches with vertical walls are considered dangerous and are not recommended, especially not in areas where animals may be introduced to other rhinos (Figure 2.4). The recommendation is that where existing vertical walls have ditches then these ditches be modified to a gradual slope with exits on either side of the ditch being provided (EAZA yearbook, 1995).

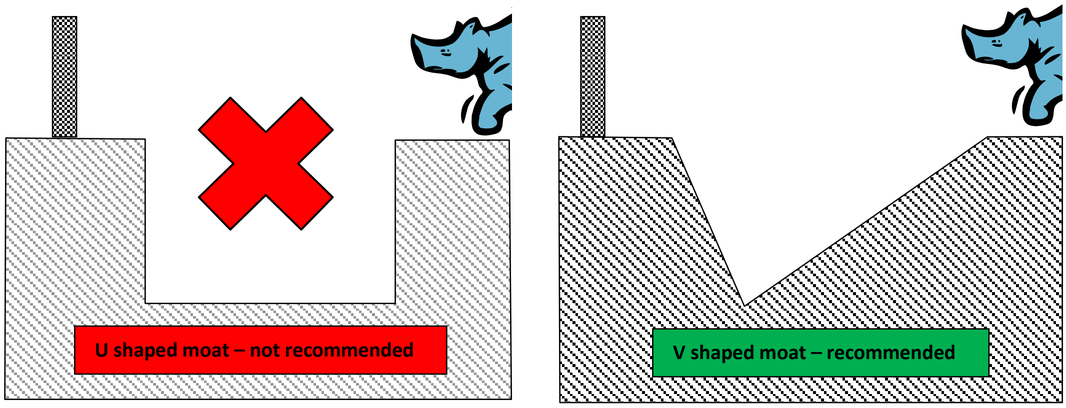

Figure 2.4: U shaped moat is not recommended, V shaped moat is fine.

Any moat used should have sloped areas to allow animals to exit easily if they enter them. It may be necessary to drain or otherwise prevent access to water moats or other areas of deep water in winter as there is a danger of animal attempting to walk on ice and slip or fall through.

It may be useful to have an area of bars (preferably vertical ones) where the keepers can examine and train the animals outside as well as inside as this will facilitate care of the animals without the need to bring them inside. Vertical pipes or posts should be spaced 25 cm to 30 cm apart (Fouraker and Wagener, 1996). Open bars and gates between the rhino paddocks are also important to allow socialisation of the animals prior to mixing. It is recommended that all introductions of animals be undertaken outside. Note, if bars are used, care should be taken to ensure that they are far enough apart to reduce the chance of rhinos trapping their horns and damaging them. If using poles, each should be about 30 cm thick and set into the ground in concrete. Bars or poles should be connected vertically to prevent the fence from being uprooted by the animals. If calves are kept outdoors, adequate measures are to be taken to prevent escaping. It is important to consider fence spacing and keeper access respectively in order to provide an exit in cases of emergency. When poles are used they must be treated with non-toxic compounds only.

Secondary barriers used to protect planting or provide escape areas for other species sharing the enclosure include fallen logs, large rocks (Figure 2.9) and lengths of primary barrier fencing. Electric fencing has been used with mixed results. Some animals respect the fence after their first shock; others seem to be irritated by it and will just rip it out. It certainly would not be a sufficient deterrent to stop a frightened or excited animal. Aprons of small sharp rocks have been tried to discourage rhinos from walking on particular areas however these do not seem to have worked and are not recommended. Triangular profile metal strips set over a shallow pit (cattle grid) have been used in one institution to restrict access to part of a paddock. This would almost certainly not deter an excited or frightened rhino and are also not recommended. To prevent individuals from being hurt, barriers should have no sharp edges (EAZA yearbook, 1995).

A variety of gate systems have been used. As with the gates used in the indoor enclosures, in general manual slides are preferred outside to swing gates or hydraulic gates both for their speed of closure and to reduce the chance of rhinos getting trapped or injured. It is important that keepers have good visibility of as much of the paddock as possible either side of the gates in order to operate them safely. When sliding gates are used, the track must be kept clean in order to reduce the chance of them seizing. All gates must be firmly secured and designed such that the animals cannot dislodge them with their horns (e.g. cap hinges so that rhinos cannot lift gates off their hinges with their horns).

It is important that animals are adequately protected from excessive disturbance from visitors. Carlstead (1999) showed that mortality since 1973 correlated positively with percentage of public access. It is difficult to make recommendations as to a suitable maximum percentage of the perimeter to which visitors be allowed access as this will depend on topography and size of the enclosure, availability of visual and auditory barriers, number and behaviour of the visitors and the temperament of the individual animals. However as a general rule it would be inadvisable to allow public to access the whole perimeter and it is important to monitor the animal's behaviour closely and to be able to make adjustment as necessary. Public should be prevented from touching or feeding rhinos, suitable standoff fencing is probably the most effective way of achieving this.

2.2.2 Outdoor substrate

Grass, soil, sand, concrete and stone have all been used successfully. It is recommended that animals have access to both hard and softer substrates in the range of enclosures to which they have access. If sand or other loose substrates are used, avoid feeding directly on these as this may promote accidental ingestion of the substrate and subsequent digestive problems. If grass or soil is used it should be taken into account that this surface may not be suitable when very wet or cold and an outside area with a suitable all weather surface should be available. Sand or concrete aprons around houses, the fence line, gateways or other high use areas are recommended to prevent these areas becoming eroded and excessively muddy. When using an area for mixing of animals, make sure that the ground conditions provide a good footing and avoid waterlogged or very slippery surfaces.

Figure 2.5: A large outdoor paddock with a sand substrate at Zoo de Doue la Fontaine, France.

2.2.3 Outdoor furnishing and maintenance

The way an enclosure is furnished can make a big difference to its suitability for mixing rhinos. Visual breaks either using the natural topography of the paddock or by planting or building artificial barriers is important to allow animals to get out of view of other animals and visitors.

A fresh water source should be constantly accessible (EAZA yearbook, 1995). Water should be changed daily. Drinking water should be offered in a water trough which should be concrete, automatic-fill or a continuous-flow device. Regular cleaning should occur at a rate that inhibits the growth of algae and bacteria. Water devices should be substantially constructed to prevent injury, upset, spillage or leakage (Fouraker and Wagener, 1996) and preferably be placed in the corner of the enclosure. It is important that these water containers have no sharp edges or corners that could injure a rhino.

Black rhinos need and enjoy access to pools and or mud wallows for skin health, temperature regulation and behavioural enrichment (Figure 2.6). The size of mud wallows should be gauged by the number of animals in the exhibit to allow ample room for each individual. Rhinos will construct their own mud wallow when given a helping start by digging out a wet area of the paddock. Mud wallows should be renovated periodically to prevent contamination.

Figure 2.6: A young Black rhino keeping cool in a mud pool.

Pools are occasionally used by Black rhino in very warm conditions, however generally they prefer mud wallows rather than water pools. Any pools used should be shallow, between 0.3 m to 1.0 m deep. To allow the rhino to safely access the pool, ramps are preferable to steps and should have a slope not greater than 20^{o}. Ideally ramps in and out of the pool should be in place around the entire perimeter, or at least in two locations around the pool. Multiple entry sites into a pool prevent it from being a dead-end in the enclosure. In the design of slopes or steps, keeper access for cleaning should be considered. The pool substrate should be broom-swept concrete to prevent it from being too slippery. Pools should be located in areas that are shaded for at least part of the day (Fouraker and Wagener, 1996). Rubbing posts may be particularly effective if placed near mud wallows or pools. Post material must be non-toxic to rhinos.

Free access to shade is essential, so sun shelters or shade in the form of trees or other vegetation must be provided. It is recommended that a number of adequate shady zones are provided, which can be natural or built structures. Sun shelters should also be usable as rain shelters; trees rarely serve this purpose (Figure 2.7).

Figure 2.7: An outdoor rain / sun shelter

Indoor enclosures are not acceptable for sun protection unless they are accessible at all times. In parts of the enclosure protection from the wind should be provided (EAZA yearbook, 1995).

Any new exhibit should include the capability for video recording systems indoor and outdoor. In addition, a scale for weighing animals is desirable and strongly recommended. A restraint device or an area for restraint should be included in the design of every facility (Fouraker and Wagener, 1996).

Natural substrates should be spot-cleaned and raked daily, and hard-surfaced areas that are not exposed to the elements should be dry-cleaned or hosed daily (Fouraker and Wagener, 1996).

Where rhinos create an outdoor midden as wild rhinos do, this should be left in place. Care needs to be taken to ensure that this does not produce hygiene problems especially during wet periods.

Thick ropes coiled round tree trunks have provided adequate protection for large trees against horn damage with smaller trees needing protection from being pushed over.

Figure 2.8: Rope protecting large trees.

Figure 2.9: Large rocks protecting a small live tree.

2.2.4 Outdoor environment

The minimum outside air temperatures suitable for rhinos to be shut outside will vary considerably with weather conditions for example with wind chill, sunshine or shade etc. It is recommended that enclosures be designed such that animals may be kept outdoors as much as is possible. However, it is not recommended that animals are shut outside for long periods at temperatures < 10^{o}C unless it is sunny and they have access to shelter. Considerably lower temperatures can be tolerated for short periods and many animals even enjoy playing in the snow! Generally it is not recommended to allow animals outside in temperatures less than -5^{o}C even for a very short time.

2.2.5 Outdoor dimensions

It is difficult to recommend appropriate outdoor enclosure size as it will vary greatly depending on the topography, furnishings and configuration of the paddocks as well as the individual animals. A range of different paddock/outside space configurations is recommended to allow flexibility to cope with the variety of situations that will arise when managing a breeding group. For example a minimum area of $1000m^2$ would be recommended for mixing animals, whereas all-weather outside yards of $500m^2$ or more may be suitable for short term use by individual animals.

2.3 Feeding

An essential consideration in the welfare of zoo animals is to provide a good diet that meets the natural feeding ecology as close as possible. Nutrition takes a big role in longevity, disease prevention, growth and reproduction (EAZA, 2006).

2.3.1 Basic diet

As mentioned in paragraph *1.3 Physiology* the anatomy of the digestive system roughly resembles that of horses. That's why the nutritional requirements for horses and ponies are also used for the Black rhino. The minimum nutrient requirements are listed in the table below (Dierenfeld 1996).

Table 2.1: Nutrient concentrations in total diets for horses and ponies (Dierenfeld, 1996).

Nutrient	**Growing**	**Mature/Maintenance**	**Pregnant/Lactating**
Dig. Energy (Mcal/kg)	2.45 - 2.90	2.0	2.25 - 2.60
Crude protein (%)	12 - 15	8.0	10 – 13
Ca (%)	0.6	0.3	0.4
P (%)	0.3	0.2	0.3
Mg (%)	0.1	0.1	0.1
K (%)	0.3	0.3	0.4
Na (%)	0.1	0.1	0.1
S (%)	0.15	0.15	0.15
Fe (mg/kg)	50	50	50
Mn (mg/kg)	40	40	40
Cu (mg/kg)	10	10	10
Zn (mg/kg)	40	40	40
Se (mg/kg)	0.1	0.1	0.1
I (mg/kg)	0.1	0.1	0.1
Co (mg/kg)	0.1	0.1	0.1
Vitamin A (IU/kg)	2000	2000	3000
Vitamin D (IU/kg)	800	300	600
Vitamin E (IU/kg)	80	50	80

Hay: Rhinos are large herbivores that are adapted for gaining energy from the fermentation of fibrous plant material. Black rhinos can be fed only grass hay. If this is done, and the protein content of the grass hay is not being monitored by laboratory analyses, the addition of legume hay (also known as alfalfa hay) to the grass portion of the diet (20% of the grass hay offered) is recommended in order to ensure adequate protein levels. However, generally a mixture of 1:1 grass hay and legume hay is recommended for the Black rhino to mimic the nutrient composition of the natural diet. There is speculation that a high proportion of grass hay may lead to excessive tooth wear in Black rhinos (Taylor *et al.*, submitted). There is no published evidence, but the exclusive use of lucerne hay for Black rhinos is discouraged. When freshly cut grass is available this could be fed as well but, when the grass is cut too short it can cause constipation of the hindgut (Clauss and Hatt, 2006). In studies of intake, digestion and passage in zoo herbivores, dry matter (DM) intakes of approximately 1% of body mass when Black rhinos were fed grass hays, and slightly higher levels (1.2 to 1.6% of body mass) when fed legume hay (Dierenfeld, 1996). According to Clauss and Hatt (2006) the maintenance requirements of hindgut fermenters should be 0.6MJ digestible energy per $kg^{0.75}$ metabolic body mass.

Browse: For browsing rhino species, the addition of fresh and / or frozen browse may be essential to dietary health. Browse may contribute required nutrients that have not yet been quantified and may also be of benefit to dilute a captive diet that is too digestible (Dierenfeld, 1996). Browse that could be fed to black rhinos are:

- Willow (*Salix spp*)
- Beech (*Fagus spp*)
- Hazel (*Corylus spp*)
- Ash (*Fraxinus*)
- Birch (*Betula spp*)
- Oak (*Quercus spp*)
- Hawthorn (*Crataegus spp*)
- Robinia (*Robinia spp*)
- Poplar (*Populus spp*)
- Apple (*Malus spp*)
- Cherry (*Prunus spp*)
- Prune (*Prunus spp*)
- Pear (*Pyrus spp*)
- Wild rose (*Rosa spp*)
- Blackberry (*Rubus spp*)

For feeding browse in the winter browse can be preserved by silaging (Clauss and Hatt, 2006). Browse should be fed 7 days per week. A browsing Black rhino can be found in Figure 3.

Figure 3: Black rhino browsing on fresh Hawthorn (*Crataegus* spp).

Concentrates: When feeding concentrates the pellets should be smaller than 1 cm in diameter for a proper intake of the pellets (Dierenfeld, 1996). The portion of pelleted compound feeds (or other forms of concentrates) in the diet should not exceed one-third of the overall calorific value. It should be possible to deliver sufficient amounts of energy and protein while providing a substantially lower proportion of pelleted compound feeds or concentrates in the diet. Pelleted compound feed may be used to balance mineral, vitamin and in some cases protein requirements. Pelleted compound feeds should only be used to satisfy energy needs when adequate roughage is not available. A pelleted compound feed based on lucerne meal, with a high concentration of vitamins and minerals (except iron) is recommended so that only small amounts need to be fed. When pelleted compound feeds are used it is recommended that it has high-fibre content (crude fibre 20% and acid-detergent fibre (ADF) of 25% of DM) (Clauss and Hatt, 2006). The proportion of concentrates in the diet should be between 1 and 10%.

Supplements: A possible vitamin-E deficiency has been suggested but not confirmed in zoo rhinos; current recommendations based on natural browse composition suggest that diets should contain 150 to 200 IU vitamin E/kg dry matter (a value usually surpassed in pelleted compound feed). If grown in an area prone to soil selenium (Se) deficiency, forage should be tested routinely for determination of Se content to provide data needed for balancing rations (Dierenfeld, 1996).

Iron: Over supplementation of iron is of particular concern in Black rhinos because this can cause several uncommon diseases. The recommended amount of 50 mg iron/kg DM for horses will probably be exceeded by the hay mixes described, and also by most pelleted feeds used. The use of tannin might reduce the excessive iron absorption. But there is no quantitative proof regarding supplementation of tannin in captive rhinos. According to Clauss and Hatt (2006), who assessed the results of studies regarding the effect of tannin supplementation in other species, the iron absorption will probably reduce by increased dietary tannin content. Extra supplementation with iron is not recommended (Clauss and Hatt, 2006). One Black rhino collection uses supplementation of tannin.

Fatty acids: The supplementation of linolenic acid (n-3) could be necessary to balance the amount of linoleic acid (n-6) and linolenic acid (n-3). This could be done by feeding fresh forage like freshly cut grass and browse, by increasing the proportion of grass or lucerne hay in the overall diet, by using concentrates that are based on lucerne meal rather than grain or soy products or by including linseed or linseed oil in the concentrates (Clauss and Hatt, 2006).

Salt lick: Black rhinos have been found to have higher endogenous faecal sodium losses. To counter sodium deficiency salt licks (suitable for horses) should be available *ad libitum* (Clauss and Hatt, 2006).

2.3.2 Special dietary requirements

Calves: Rhino calves can be hand reared in captivity. Please refer to chapter *2.5.6 Hand rearing* for information about milk formulas, hand rearing and weaning.

2.3.3 Method of feeding

The concentrate portion of the ration should be given in at least two feedings daily for better utilisation. When practical, a small feeding of hay should be encouraged prior to each concentrate feeding (Dierenfeld, 1996). Feeding can either occur at a fixed time and fixed feeding place, or hidden around the enclosure.

Food should be offered on a concrete pad / livestock troughs / hay racks or simply a pile in the corner. Sand impaction has previously been documented in rhinos; therefore, feeding directly on the ground is not recommended. To reduce competition for food if animals are kept together, individual feeding stations or adequate space at communal feeders is recommended (Dierenfeld, 1996).

If Black rhinos have any problem with eating, animals can sometimes be encouraged to consume less palatable forages if hays are soaked in water or sprinkled with molasses.

Apple sauce had proved to be helpful in administering unpalatable medications and / or supplements (Dierenfeld, 1996).

2.3.4 Body condition scoring

To decide when to increase or decrease the amount of food, the body condition score should be used. Body condition scoring (BCS) involves the visual assessment of specific parts of the body for muscle and fat content, and can be a useful indicator of general health and condition of an individual. A standardised body condition scoring system has previously been developed for Black rhinoceros (Reuter and Adcock 1998), which assesses seven key areas of the body, ranging from BCS 1.0 (emaciated) to 5.0 (heavy). Ideally, a 0.5 point scale can be used to assess these areas, to give a representation of the relative condition of individuals. It should be noted that the Reuter and Adcock (1998) BCS was designed for wild living rhinos where they consider a score of 5 to be excellent. In zoos however we see overweight animals and therefore consider a BCS of 4 to be ideal.

Reuter and Adcock (1998) have made a list of criteria for each body condition score. These criteria can be found in Appendix I (Reuter and Adcock). Another way to assess the nutritional status of the rhinos is by regularly weighing the animals and recording this data (Clauss and Hatt, 2006). Weighing of the diet is recommended.

Body condition may be of relevance to reproductive performance, particularly in females, as recent research has indicated that non-breeding females tend to have higher BCS than breeding females (60% of non-proven females were scored 4.5 compared to only 6% of proven females) (Edwards et al. 2013).

It is recommended that a side view and rear view colour photograph (not taken in bright sunlight) are sent to Becca Biddle at Chester Zoo (b.biddle@chesterzoo.org) annually, to validate the body condition score.

A rhino with a good body condition score can be recognised by:

- The neck appears thick across the top, and is well muscled, with a smooth gradation between the neck and the shoulder blade.
- The shoulder (scapular region) is well covered, and slightly rounded but not bulging.
- The ribs (costal region) are covered with thick skin folds, especially just behind the shoulder and elbow region.
- The spine (vertebral region) appears rounded and the long back muscle and fat deposits fill the gap between the ribs and the spine.
- The bony points of the rump (gluteal region) are covered and the rump appears flat as opposed to rounded.
- The abdomen (abdominal region) appears filled and taught.
- The tail base should be rounded but not bulging.

In Figure 3.1 you can see the different body regions, and in Figure 3.2 the different body condition scores 1 – 4 are displayed (Reuter and Adcock, 1998).

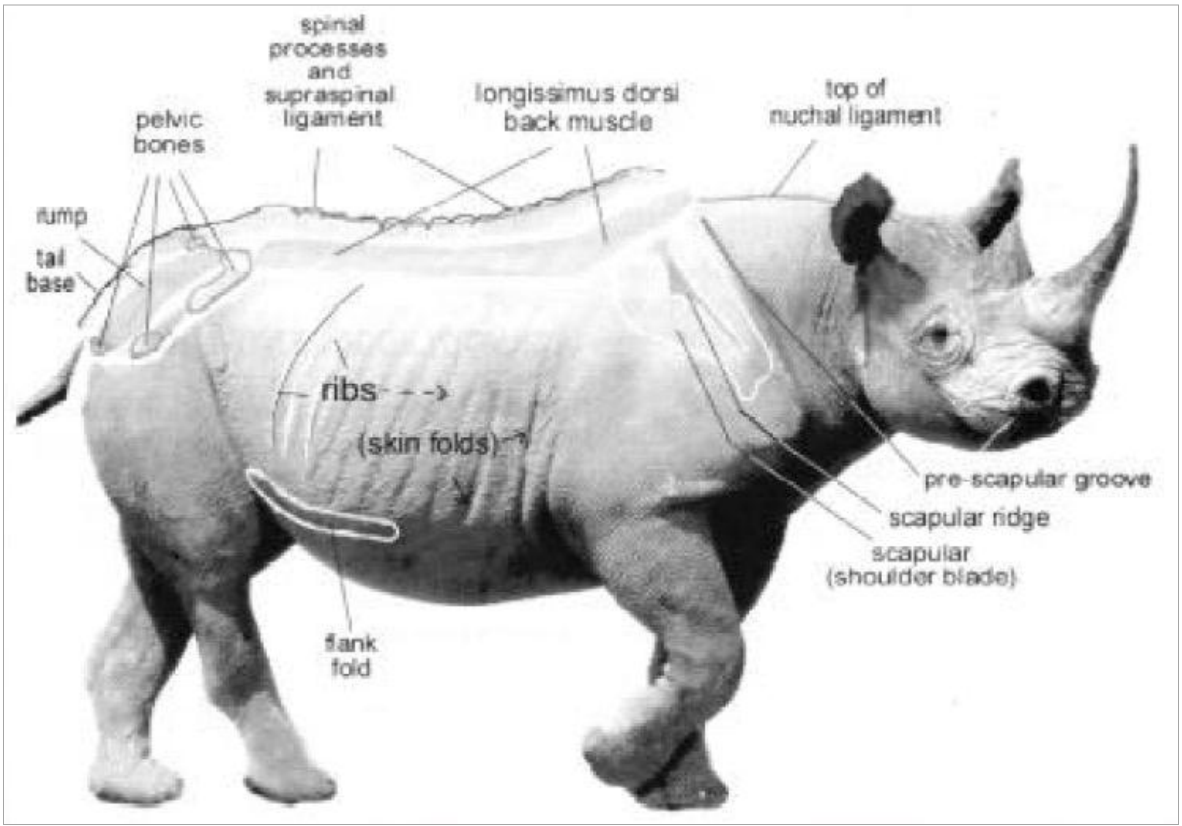

Figure 3.1: Black rhino body regions (Reuter and Adcock, 1998).

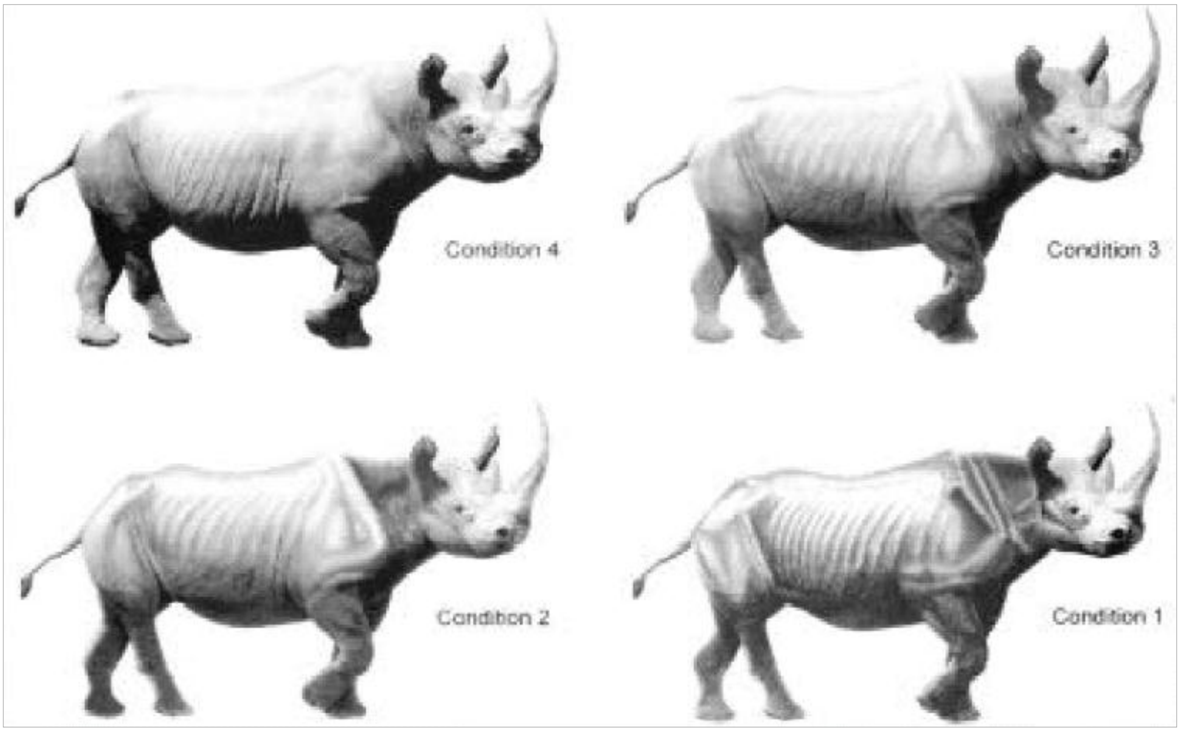

Figure 3.2: Black rhino body condition scores (Reuter and Adcock, 1998).

2.3.5 Water

Water should be freely available at all times (Dierenfeld, 1996; EAZA yearbook).

2.4 Social structure

The social organisation of the Black rhino in the wild is described in chapter *1.8.4 Social behaviour.*

The following chapter describes the social structure and introduction procedures in captivity.

2.4.1 Basic social structure

Black rhinos should be kept individually, in pairs, or in a trio of one male and two females. Keeping Black rhinos in larger temporary groups may possible particularly when young, however this varies between individuals and will depend on the size of the enclosure and the number of visual breaks. Two adult females together often works well but as all Black rhino groups they need careful observation.

The optimum group composition is 1.2. Since Black rhino males are territorial, each bull should have his own enclosure. Adult males in the same exhibit are not recommended and barriers must be solidly built, with no visual contact. Adult females in the same exhibit may be possible. An adult male and an adult female in the same exhibit is often possible but may need separating at times. Allowing compatible individuals to spend as much time as possible with each other may prove beneficial. Almost all of the institutions holding Black rhinos house them in pair situations, although they have also been exhibited in trios (1.2). If this is being considered, animal managers should monitor the behaviour of the dominant female closely, as female suppression has been recorded in select cases (EAZA yearbook, 1995; Fouraker and Wagener, 1996; Rieches, 1999).

2.4.2 Changing group structure

When introducing rhinos it is important to provide auditory, olfactory and visual contact between the individuals. Tactile contact through bars should be provided. Introductions should be made after careful acclimatisation of the animals. Experienced staff members should always be present for introductions and a plan for separating them should be in place, should this be necessary e.g. high pressure water hose (EAZA yearbook, 1995). It may be necessary to keep the animals separately when they are inside at night.

Steps in the introduction process - *this applies to all introductions, the animals should be kept separated until step 4.*

1. Animals in the same indoor enclosure or multiple outdoor enclosures should have olfactory and auditory exposure to each other. If the animals are not housed near each other (i.e., enclosures on opposite sides of the zoo, etc.) they should be moved to the same exhibit area.
2. Animals should be given visual contact with each other in addition to the above sensory modalities. If at any point during this process the animals display symptoms associated with stress (e.g., pacing, diarrhoea, excessive vocalisation) for more than two to three hours, the introduction should return to the previous step.
3. If animals are not already positioned adjacent to each other, they should be moved closer together (e.g., to adjacent stalls or adjacent outdoor enclosures).
4. The actual introduction (full tactile exposure) should take place in the largest enclosure available. Preferably, the enclosure should be familiar to the least dominant animal and include ample “run-arounds”.
5. Within institutions in which rhinos can be left together 24 hours a day, they should be separated during the first several nights or until they show only minor aggression (Fouraker and Wagener, 1996).

Aggression in Black rhinos ranges from ritualised to true aggression. Face-to-face staring is often seen at the beginning stages of ritualised aggression and may be an opportunity for the participants to “size each other up”. Ritualised aggression may subsequently proceed to fencing or sparring and then charging with or without an open mouth threat. Aggression becomes more serious as one animal begins chasing the other, which also include or lead to horn strikes and gores. With this kind of aggression problems arise in small enclosures and in dry moat ditches. Another indicator for aggression is coming into oestrus. Excited animals run with their tail up and very often start to be aggressive (Fouraker and Wagener, 1996).

Tranquilisers are only recommended when aggressive animals are involved or / and when an animal is very nervous. Two experts reported experience with the use of tranquilisers during an introduction, there are different methods described in chapter *2.7.3 Catching / restraining.*

When introducing more animals together, at first start with the introduction of the females and with the male as the last animal to be introduced. The introduction of a male and a (post-partum) female is described in chapter *2.5.1 Mating*.

Introduction of a new female to an established male-female group: Female Black rhinos generally do not tend to form strong pair bonds. Therefore, a new female should be introduced to an established male-female group one individual at a time, but it is not necessary that she is introduced to all females before being introduced to the male (Fouraker and Wagener, 1996).

Introduction of a new male to an established female group: As previously discussed, female Black rhinos do not generally tend to form strong pair bonds. However, if a multiple-female group is established and managers perceive that the females have formed strong bonds, the new male should be introduced to the females as a group rather than to one female at a time. If the females are not compatible, but an introduction is necessary (EEP recommendation, breeding, etc.), the new male should be introduced to each female individually (Fouraker and Wagener, 1996).

Introducing a female to a female: Two females may be kept in the same enclosure depending on the characters of both animals. Initially, the females should be familiarised with the enclosure. The females should have contact through bars when they are indoors. When they no longer show aggression toward each other, the rhinos can be introduced. Close observation is necessary after the introduction (EAZA yearbook, 1995).

2.4.3 Sharing enclosure with other species

The critical factors for sharing the enclosure with other species are space and refuge availability, including visual barriers. In a non-breeding situation rhino species have been successfully mixed with birds and hoof stock. In all cases the dispositions of the individual animals, as well as adequate space and exhibit structure are important considerations prior to attempting a mixed species exhibit. Black rhinos have been reported to be problematic in mixed species exhibits. Both sexes have attacked and killed neonate and adult ungulates (EAZA yearbook, 1995; Fouraker and Wagener, 1996; Guldenschuh and von Houwald, 2002; Rieches, 1999).

Black rhinos can be mixed indoor with several small bird species. Another species mentioned is the ostrich however this mix has failed. Black rhinos can be mixed with (water) birds. According to the Rhino Keepers' Workshop 2001 Husbandry Survey one zoo, Disney Animal Kingdom, has a mixed enclosure involving Black rhino, Pink backed pelican (*Pelecanus rufescens*) and Yellow billed stork (*Mycteria ibis*) *(Mehrdadfar, 2002).*

Red-billed ox peckers (*Buphagus erythrorhynchus*) have been mixed with Black rhinos in an indoor exhibit. The rhinos and ox peckers were only mixed when the rhinos were indoors. During the day the rhinos were locked outside of the house. Other species present in the same exhibit were Double-toothed barbet (*Lybius bidentatus*), Violet turaco (*Musophaga violacea*), Cattle egret (*Bubulcus ibis*), African grey hornbill (*Tockus nasutus epirhinus*), and Dinemelli's weaver (*Dinemellia dinemelli*). The Black rhinos were highly intolerant to the ox peckers. The Ox peckers were feeding on the wounds of the Black rhino and they were making new wounds. Only half of the time the rhinos managed to chase the Ox peckers away. It is recommended observing both species when mixed (McElligott et al, 2004).

2.5 Breeding

Breeding success is related to the size of the zoo enclosures where courtship and mating occurs. This is important because of the quite aggressive pre-copulatory behaviour. The primarily requirement for successful breeding Black rhinos is pair compatibility. Research of Carlstead *et al*, suggests that "a compatible pair is one in which the female is relatively more aggressive and assertive, and the male more submissive and adaptable" (Carlstead et al, 1999).

2.5.1 Mating

Breeding success may be enhanced by separating males from females as little as possible. For general information about mixing Black rhinos please refer to chapter *2.4.2 Changing group structure.* Specific information about male and female introduction is described below. This followed by a description of behaviour for female and male in relation with mating.

Introducing a male to a female: It is often easier to introduce a male to a female when she is in oestrus. Some collections have reported that for some pairs it is better to introduce them before the female is in oestrous as the male may get too excited when introduced when the female is in oestrus. This is very dependent on the individuals involved. Some females do not express clear behavioural signs of oestrus, even if they are cycling regularly and so should in fact be receptive. This has been reported to be a particular problem in previously non-proven females (Edwards *et al*. 2013), where females may be cycling based on hormone data, but often do not express overt behavioural signs of oestrus. Hormone analysis can then prove useful by allowing prediction of the period of female receptivity, even if behavioural signs are relatively absent. When introducing a male to a female both animals should be familiarised with the enclosure. The introduction should occur in the largest paddock available, following the general introduction steps stated in chapter *2.4.2 Changing group structure*. If a single large paddock is not available, adjoining paddocks should be opened to form a large area for the introduction. If the latter strategy is used, care should be taken to modify any resulting dead ends in the exhibit where a rhino may become trapped during an aggressive interaction. This method is proven to be successful. If it is not the usual enclosure of the male, he should have been given time to mark the enclosure. This should not be cleaned out. Before the introduction the contact through bars (indoors) must be sufficiently long until no more aggression is shown. Observation should continue when needed. In any case but especially when aggressive situations arise, preferred feed should widely be distributed throughout the enclosure (fresh greens, browse, carrots, etc) (EAZA yearbook, 1995; Fouraker and Wagener, 1996).

Introducing a postpartum female to a male: The reintroduction of a male to a female immediately postpartum is not recommended. If the calf is still born or does not survive it is recommended that after four months, reproductive hormone monitoring is resumed to determine the next oestrus. This combined with a judgement of the female's health dependant on the difficulty of the birth, can be used to decide when to reintroduce the male for mating. If the calf survives, it is recommended that reproductive hormone monitoring resumes by seven months to determine the next oestrus.

It has been reported in the wild female Black rhinos that cyclicity resumes as early as three months after parturition (Goddard, 1967). There has been one report from a captive female at Hannover zoo who was observed to be in oestrus 20 days following giving birth (Dittrich 1967), she was then reported as cycling regularly every 25 - 30 days (Goddard 1967) until conception approximately twelve months after giving birth. Hormone analysis on captive females by Brown *et al*. (2001) has confirmed that females generally resumed cyclicity within three to ten months post-partum.

If cycling occurs, and it is possible to separate the mother from the calf for a long enough period of time, the male can be reintroduced for mating when the calf is separated. The possibility of training the calf to be separated from the female after the age of seven months, to allow re-mating of the female, depends highly on the character and behaviour of both adult and calf, but can help to reduce the interbirthing period.

Studies have shown interbirthing periods in wild Black rhinos to be highly variable; from as short as 20 months, to 89 months, the mean interbirthing period of Black rhinos was 44 months in Hluhluwe National Park and 30 months at Umfolozi National Park (Bertschinger, 1994). In captivity the shortest interbirthing period reported is 16 months (Smith and Read 1992), indicating potential conception during first post-partum oestrus, however this is relatively rare.

The average interbirthing period in the current living EEP (Oct 2013) is 46 months (calculated from 40 separate cases where the female's previous calf survived more than four years). This is longer than seen in the wild. In almost half of these cases (48%) the interbirthing period was less than 40 months meaning the calf was separated at less than two years old to allow re-mating of the female. In 13% of these cases, the calf was less than 18 months when separated from the female to allow her to be re-mated. The shortest interbirthing period from these 40 cases was 26 months. The female in this case was 'Nane.' If Krefeld zoo, and if the gestation period was fully served, her calf must have been separated from her at ten months old to allow mating to occur. In Nane's case, short interbirthing periods are common; she has four surviving offspring born at intervals of 27, 26 and 36 months. This may be due to a combination of factors including her confident character, dominance over the male, and very peaceful mating (always the same male). Nane gives clear behavioural indicators that she has come back into oestrus between three and four months after parturition allowing the keepers to decide the best time to re-introduce the male.

Female behaviour: At the peak of oestrus the female shows the following behaviour: *positive male solicitation, presentation of hindquarters, aggression towards the male, running from the male, ignoring the male, copulation as well as refusing to copulate. Vulva changes include: squirting of white or cloudy urine, vulva swelling with occasional mucosal discharge prior to mating* (Fouraker and Wagener, 1996). At least two collections have succesfully used the hormone treatment *Regumate* to induce ovulation.

Male behaviour: Although, males in captivity may show interest in females outside of peak oestrus, greatest interest is seen during the peak of oestrus. At the peak of oestrus the male frequently exhibits the following behaviour: *erection, genital inspection and flehmen response, head resting, chasing, mounting, copulation, failing copulation attempts* (Fouraker and Wagener, 1996).

Ideally a male will show interest in a female one to three days prior to oestrus. The couple may need to be separated for the night. Copulation normally occurs the next day. Copulation will last 20 to 45 minutes with multiple ejaculations. Mating may occur for 24 hours (Fouraker and Wagener, 1996).

2.5.2 Reproductive endocrinology as a management tool

Using hormone analysis as an additional tool to manage introductions can be very useful, particularly in females where behavioural signs of oestrus are weak or unreliable. Non-invasive approaches are preferable as they minimise the disturbance to the animal and can allow long-term sampling as part of the keepers' daily routine. Faecal samples are often preferable to urine samples, as collection is often easier, and requires no additional training of the animal, instead fresh samples can be collected when the animal is let outside first thing in the morning. Samples should be frozen immediately after collection, and stored frozen until shipping to a laboratory for analysis.

Samples collected at least every other day are necessary for characterising oestrous cycles in females, and can be used to determine a females' typical oestrous cycle length. This can then be used to predict when she will next be in oestrus, and therefore give keepers extra confidence when deciding when to introduce a pair (or trio) for breeding purposes. See Figure 3.3 for an example of results of hormone sampling to predict cycling. Samples collected on a weekly basis are sufficient to investigate differences in testosterone between males (see Appendix III for faecal sample collection protocol).

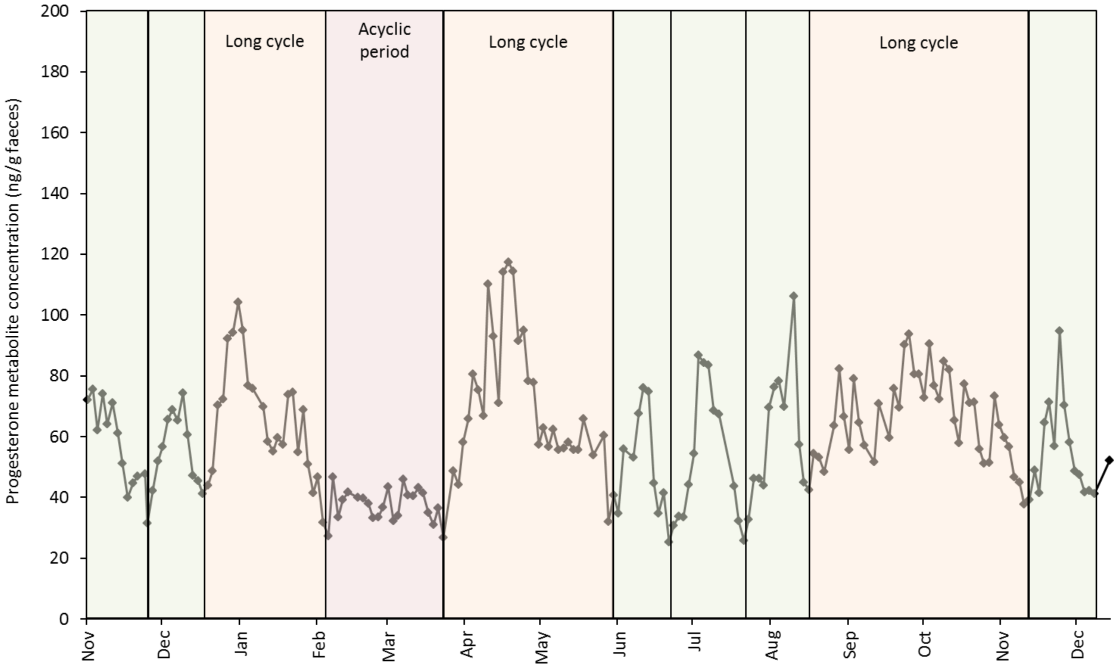

Figure 3.3: Progesterone metabolites (♦) measured in faeces collected from a female Black rhino can be used to characterise oestrous cycles. However, the length of oestrous cycles can be highly variable, with a normal cycle lasting between 20-40 days in length (green sections). However, shorter cycles less than 20 days (not shown) and longer cycles greater than 40 days in length (orange sections) are also relatively common, and periods of acyclicity (red section) are also observed.

2.5.3 Pregnancy

The gestation period is around fifteen to seventeen months, more information about the gestation period is found in chapter *1.7.5 Gestation period / birth rate.* It is recommended to test for pregnancy. Pregnancy testing can be done by faecal steroid sampling, urine steroid sampling, blood steroid sampling and by ultrasound. For blood sampling and using ultrasound the animal involved needs to be trained, while faecal and urine sampling involve less effort (EAZA yearbook, 1995).

Pregnancy diagnosis can also be performed using hormone analyses, samples collected every other day can be used to distinguish an increase in progesterone metabolite concentration, which occurs at around three months post-mating – patterns of progesterone secretion during this first three months are non-conclusive, and seem to show individual variation. Based on this progesterone metabolite increase, parturition can be approximated (see Figure 3.4 for an example of a female Black rhino hormone profile during pregnancy).

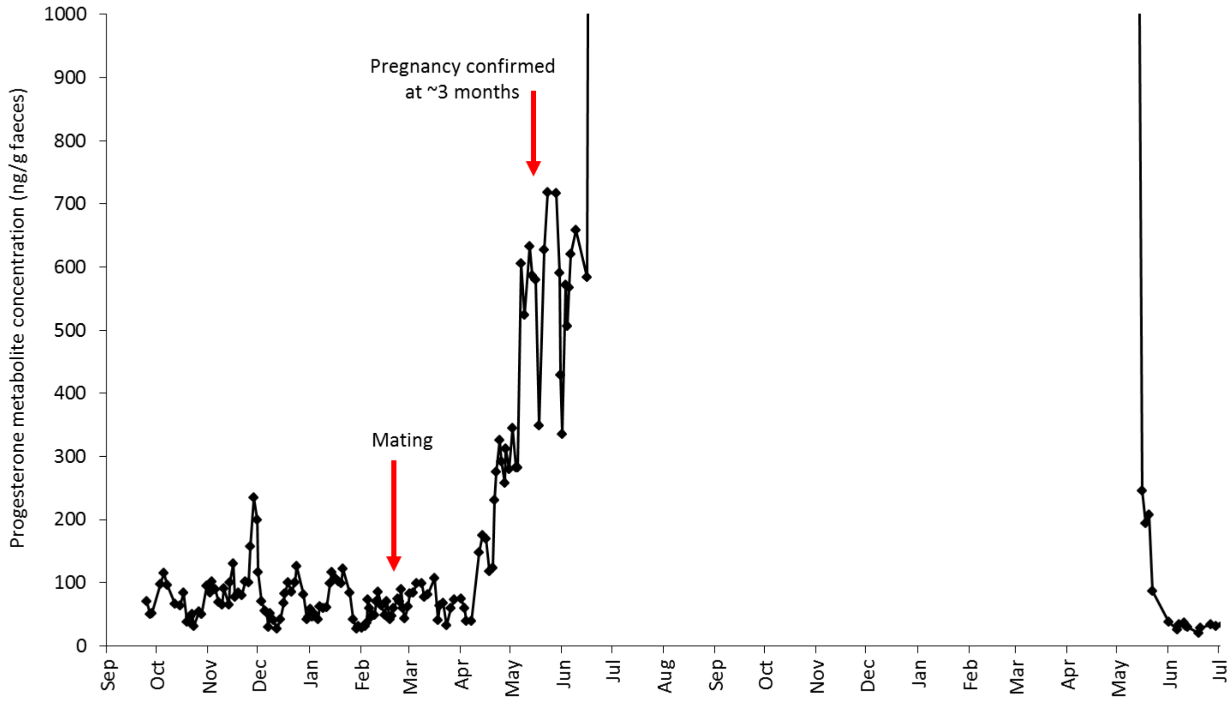

Figure 3.4: Example hormone profiles of a single Black rhinoceros pregnancy, in this case lasting 449 days. (a) Routine hormone monitoring was used to predict oestrus and time introduction to the male. After a successful mating it took approximately three months before pregnancy could reliably be confirmed, as individuals differ in their progesterone profile during this initial period.

2.5.4 Birth

The duration of delivery is short, usually lasting between one and three hours. Births often take place at night or in the early morning. It is not necessary to observe the birth, but if that is the case it is recommended to watch remotely on a TV monitor. Distocia is very rare. See Appendix IIII for an example birthing plan for female rhino *Ema Elsa* at Chester Zoo.

Indicators prior to birth:

- *Thirty days:* The female Black rhino teat size may increase and milk production begins. When pressure is put on the teats milk may be expended. It might be that the female prolapses vaginally when defecating (Fouraker and Wagener, 1996).

- *Two weeks:* The nipples of the female rhino may enlarge and the nipples develop wax plugs. The vulva will start swelling as well (Fouraker and Wagener, 1996).

- *Twenty-four to forty-eight hours*: The female Black rhino becomes irritable and aggressive to stimuli, including to the staff, also she may lose her appetite. The udder will increase dramatically in size; mucus plug forms and increased vulva dilation occurs. The female will lie down more often and is restless. Other behaviour mentioned is frequent urination (Fouraker and Wagener, 1996).

Additionally, if daily hormone analysis is performed leading up to birth, a drop in progesterone metabolite concentration can precede parturition by 24 - 48 hours, allowing prediction of parturition in cases where this may be deemed necessary (Figure 3.5).

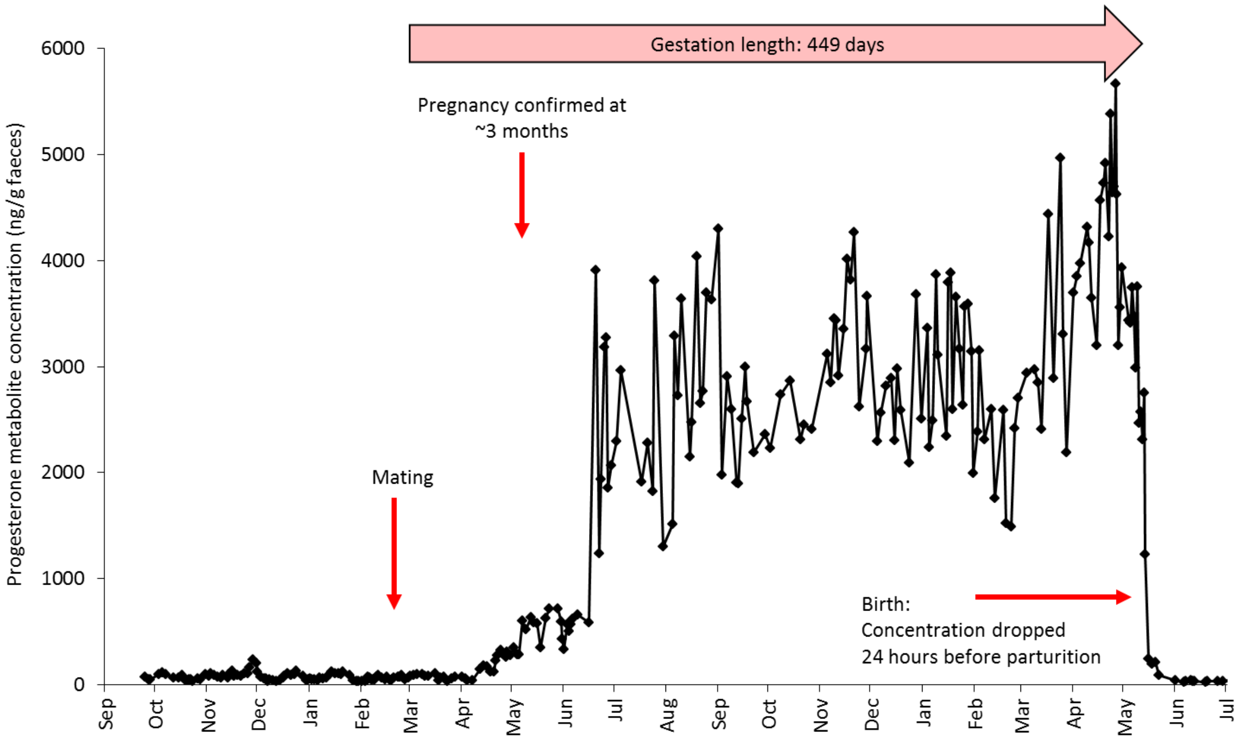

Figure 3.5: Example hormone profiles of a single Black rhinoceros pregnancy, in this case lasting 449 days. (b) At three months, progesterone concentration increases dramatically, and can be 10-20 fold higher than peak cycling concentrations. Parturition was predicted using hormone analyses as progesterone metabolite concentrations dropped 24 hours prior to parturition.

2.5.5 *Development and care of the calf*

Calving usually occurs during the night or the early hours of the morning. When the calf is born its weight is around 40 kg (Nowak, 1999). Immediately following birth, the new-born calf is usually cleaned by its mother. First standing up will be seen from 15 minutes to one or two hours after birth. A new-born calf may require traction material to help steady itself. Traction materials may include sand, gravel, straw, hay or rubber matting. These additional materials should be added well in advance of calving to give the female time to get used to them. In all cases, both the dam and calf should be monitored closely but without disturbing them to allow them to bond. Monitoring using closed circuit television equipment may be preferable for a nervous or first time mother. A calf should begin suckling within one to two hours of standing. The mother will nurse the calf standing or lying on her side (Fouraker and Wagener, 1996).

A new-born rhino should be up and walking within approximately one hour after a normal birth. Following a dystocia, or breach birth, the dam may be too exhausted to clean and care for the calf immediately. Similarly, calves weak from a dystocia may also take longer to stand. This, however, does not mean that intervention is necessary. It is advised to keep close monitoring. Suckling should be seen within the first five hours and should be frequent. While sleeping the calf should be with the dam constantly and touching her. A problem is indicated if the calf is seen alone for extended periods, appears weak, or is having trouble keeping up with the dam. If a problem is suspected and the dam and calf can be safely separated, daily weights of the calf should be obtained. Normal daily weight gains are representative of nursing success. When calves are under optimal weight, they should get supplemental bottle feedings. For bottle feedings check chapter *2.5.6 Hand rearing*. The faeces should be grey or yellowish grey in colour and the consistency of stiff putty. A healthy calf may not have a stool for the first 24 to 48 hours. For all calves with diarrhoea, a faecal sample should be submitted for culture of enteric pathogens and internal parasite screen. Daily observations of the calf's stamina are important, as its condition may decline rapidly. Any deviation from the normal body temperature of 36.9 -37.8^{o}C is likely an indication of poor health (Gage, 2002).

A Black rhino calf should be separated from the mother between the ages of two and four years. It should not be sooner than 18 months.

2.5.6 Hand rearing

Hand rearing should only occur if absolutely necessary.

There are successful experiences with the hand rearing of Black rhinos. Hand rearing is only recommended when there is no other possibility. The infant should always remain with the mother and (when needed) be additionally bottle fed. Hand rearing could become necessary when the young is rejected by the mother or medical problems with the mother and / or infant exist, or when the infant fails to nurse. The hand rearing of an infant has to be considered very carefully and intensive care of one or two keepers will be needed (EAZA yearbook, 1995).

Record keeping: Accurate record keeping is extremely important. Maintain a daily log of formula intake, body weight, body temperatures, and faecal output and consistency. It is helpful to have this information in a format such that one month's statistics may be viewed on one page. Note exercise times and behavioural changes each day. Rhino calves should urinate large amounts daily without the assistance of physical stimulation. Weigh the calf at the same time each day for two to four weeks, every other day until two months of age, twice a week until three months of age, and once a week until five to six months of age. Weights may be obtained by leading the calf onto a platform scale while it is nursing on a bottle (Gage, 2002). All information should be sent to the EEP coordinator.

Equipment: The following items should be on hand:

- Pliable, one litre polyethylene laboratory bottle with a narrow mouth
- Artificial lamb's nipple with a crosscut opening
- Calf bottles and calf nipples
- Large containers with screw tops for storing formula
- Large cooking pot, hot plate, large refrigerator, sink, disinfectant and bottle brush

- Measuring cups, gram scale, walk-on platform scale
- Large stuffed animal to serve as a companion
- A radiant room heater, or other safe heating system, and an electric blanket may be needed (Gage, 2002)

Housing: For a healthy calf the air temperature should be between 15-30^{o}C. If the air temperature is expected to drop below 15^{o}C, for example during the night, a heater will be needed. When the calf is hypothermic or debilitated a constant temperature of 26-30^{o}C is recommended. For bedding substrates like soft hay should be used. Wood shavings should be avoided due to possible intake, when the calf lies with its lips on the ground (Gage, 2002).

For mental and physical development a large exercise yard is important. A healthy new-born should be walked for half an hour, twice a day. The first few days the calf is allowed to explore the yard on its own with a keeper nearby for emotional security. A rhino calf loves to run and will do so after three-four days of age. This daily exercise encourages normal defecation (Gage, 2002; Wagner and Edwards, 2002).

For comfort and companionship a large stuffed animal may be placed with the calf. If another large ungulate neonate is available, it may be placed with the rhino calf after it has reached one week of age, replacing the stuffed animal. When there is no neonate ungulate available a young or adult sheep or goat can be used. Developing the bond between the two animals might take a week. First when one of them is nervous, it may be necessary to separate them during the night, putting the stuffed animal back in with the rhino calf. A companion animal also discourages the rhino from becoming dependent on keepers for security and companionship (Gage, 2002; Wagner and Edwards, 2002). Before introducing any companion animals it is essential that you know the full health history of the animal and the group it came from. Also, the animal must be healthy and parasite free. It is recommended that you check with your own veterinary advisor before any introduction to ensure that disease is not transmitted from the companion animal to the young rhino.

Toys should be provided at an age of one or two weeks. The toys should provide the calf to exercise its natural behaviour of head butting. It is important that the animal does not practise this behaviour on the keepers, due to potential danger, this behaviour should be discouraged. Suitable toys are: two electric cart tires bolted together in order to keep them upright and rolling, large plastic trash cans, boomer balls, or any other object which can be pushed around without the risk of the young rhino wedging its head inside the object (Gage, 2002).

Milk composition and formula selection: Based on available data, rhinoceros milk is more dilute than milks of other ungulate species. It is low in solids, low in protein, very low in fat, and high in sugar compared with milk of equids, bovids and cervids (Dierenfeld, 1996).

Though rhinoceros' milk is different from cow's milk, the latter may still be appropriate for hand rearing rhinos if used in combination with other ingredients, like extra iron, vitamins and lactaid. There is also special artificial milk available, and horse milk can be used as well. Cow's milk is low in iron; consequently, an iron source should be added to the formula at two drops per 100 g of formula.

In addition, infant vitamins should also be added to the formula at two drops per 100 g of formula. Some infant vitamins may contains added iron.

The animal may also benefit from the addition of lactaid at one drop per 100 g of formula. Lactaid aids in carbohydrate digestion and helps prevent possible gastrointestinal distress *(Dierenfeld, 1996).* If the neonate is less than 24 hours old, colostrums diluted 50% with water or an electrolyte solution for ungulates, such as Replenish, should be administered for the first 24 hours. Though species-specific is preferred, cow colostrums may be used. To avoid gastrointestinal distress, a diluted formula may be offered beginning on day two. The formula can be gradually increased to full concentration depending on the animal's health, including weight gain and stool condition. Table 2.2 shows an example for formula and feeding regime used in San Diego Wild Animal Park. This method was successful for hand rearing Black rhino calves (Dierenfeld, 1996; Gage, 2002).

Table 2.2: Rhino formula and guidelines used at the San Diego Wild Animal Park (Gage, 2002; Wagner and Edwards, 2002).

Age	**Formula**	**Ratios**	**Feedings per day**[1]
1 day old	100% Cow's colostrum		7 times, every 2 hrs
2 days old	NFC:LFC:Lactose:H_2O w/ 50% colostrum	27:9:1:1[2]	7 times, every 2 hrs
3 days to 1 month Early lactation form.	NFC:LFC:Lactose:H_2O w/ 10% colostrum	27:9:1:1	7 times, every 2 hrs
1 to 3,5 months Early lactation form.	NFC:LFC:Lactose:H_2O	27:9:1:1	5 times, every 3 hrs[3]
3,5 to 6 months Mid-lactation form.	NFC:LFC:Lactose:H_2O	27:9:1:2	4 times
6 to 9 months Mid-lactation form.	NFC:LFC:Lactose:H_2O	27:9:1:3	3 times
9 to 12 months Mid-lactation form	NFC:LFC:Lactose:H_2O	27:9:1:4	3 times
12 to 15 months Late lactation form.	NFC:LFC:Lactose:H_2O	27:9:1:6	2 times
15 to 16 months Late lactation form.	NFC:LFC:Lactose:H_2O	27:9:1:8	2 times

Note: NFC = liquid non-fat cow's milk (skim milk); LFC= Liquid low-fat cow's milk (1% fat); lactose powdered, edible grade dextrose (reagent grade) may be substituted for the lactose.

[1]Day consists of a twelve-hour period from 6 a.m. to 6 p.m.
[2]27 parts NFC to 9 parts LFC to 1 part Lactose to 1 part water
[3]At roughly two months of age the calf can go to 4 times per day.

Feeding regime: Hygiene is very important in order to avoid contamination of the milk. The calf should be preferably bottle fed instead of feeding with a bucket to avoid hasty drinking. Fresh water should be available at all times.

Quantity fed should range from 10% to 13% of body weight. Rhinos do not need to be fed around the clock. Animals should be fed every two hours. Because infants suckle during daylight hours, feeding should be equally spaced in a twelve hour period not to exceed 3% of body weight at any one feeding. It is recommended that feeding begin with 10% of body weight split equally into seven feeds two hours apart during daylight hours.

The quantity of formula fed should be adjusted daily based on the animal's weight. Animals should be weighed at the same time each day. During the first weeks feeding should also be during night, with an interval of two to three hours. If diarrhoea occurs, the quantity of formula fed should be decreased or the formula diluted until stool condition returns to normal. If diarrhoea is persistent, an electrolyte solution can be used to dilute the formula, replacing some or all of the water. In addition, the number of feedings can be increased to lessen the quantity fed at any one time.

Formula can be prepared ahead of time and warmed as needed. Water should be boiled to decrease possible contamination due to pathogens, and then refrigerated before being added to the formula. The formula should be refrigerated and used within 72 hours.

Prior to feeding, the formula should be warmed to the animal's body temperature. Calf nipples work well. Bottles should be boiled before use. Diluted bleach may be used as a disinfectant. Formula left over from each feed should be discarded (Dierenfeld, 1996; EAZA yearbook, 1995; Gage, 2002).

Weaning: Weaning may begin as early as six months and should be completed in one year. Weaning is a slow process involving carefully monitoring body weight and solid food consumption. Animals should have access to solid food at all times.

A nutritionally complete pelleted diet such as Calf Manna, horse feeds or high fibre ungulate pellets, in addition to alfalfa hay, is appropriate. Formula may be decreased by gradually elimination the number of feeds or decreasing the amount offered per feed and gradually decreasing the number of feeds (Dierenfeld, 1996; Gage, 2002).

2.5.7 Population management

There is an international studbook for the Black rhino. In Europe there are only two subspecies of the Black rhino in captivity, the eastern Black rhino (*Diceros bicornis micheali*) and the south-central Black rhino (*D. b. minor*). The Regional collection plan of the EAZA rhino TAG from 2002 recommends the eastern Black rhino for EEP management. The south-central ssp. is not recommended to keep. The target population for the eastern Black rhino is 65/100/100 over 10, 50 and 100 years period. The EEP coordinator will determine the need for new specimens for the region. Other breeding programmes for the Black rhino are managed by AZA (SSP) and JAZA (SSCJ) (Lindsay, 2002; Foose and Wiese, 2006).

2.5.8 Sustainability of the EEP population

A recent EEP supported research project established to investigate population performance in Europe has used studbook data to predict the future growth of the population (Figure 3.6). These data suggest that unless reproductive output is increased, growth of the population is projected to be between one to two percent per year. In comparison, average growth rates of free living Black rhinoceros populations are around five percent per year, a target that could be achieved in the zoo population if reproductive output could be increased.

In the zoo population, average age at first reproduction is slightly later, and average inter-birth intervals (IBI) and are slightly longer than in the wild, but due to management constraints in captivity these may be difficult to reduce dramatically. However, the two main factors limiting population growth in Europe are a low proportion of females breeding per year (11.3% 2001-2010 and 15.7% 1986-2010 compared to 23.7% in the wild), and an unequal reproductive skew, with 42.1% males age 7-32 and 48.6% females age 5-32 yet to reproduce as of 31st December 2010. This latter factor could also have an impact on the genetic diversity of the population in the long-term.

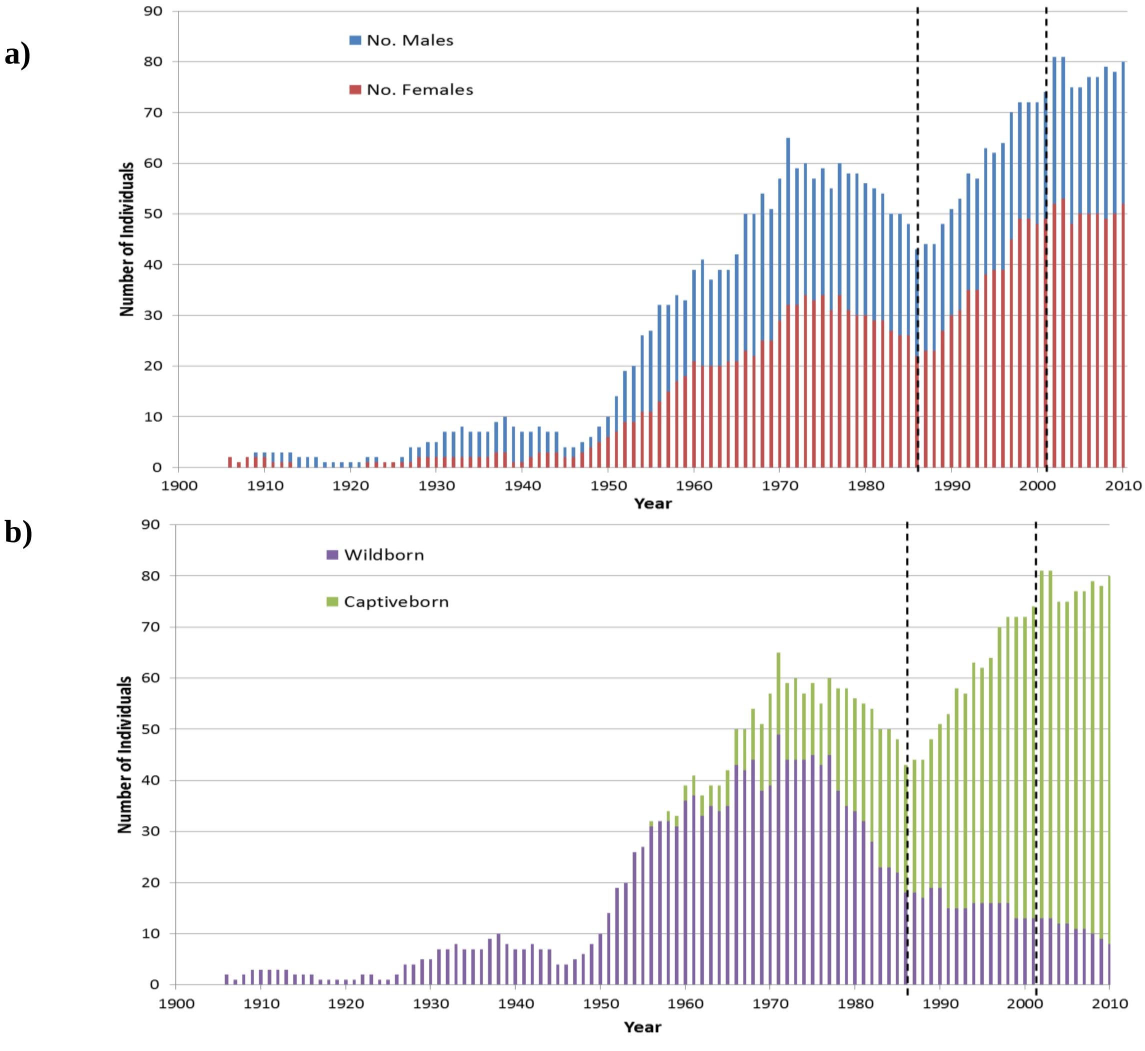

Figure 3.6: Census of the EEP Black rhinoceros population from studbook records indicating a) the number of females (red) and males (blue), or b) the number of wild caught (purple) and captive born (green) which make up the total population size each year. Dotted lines indicate the last 25-year and last 10-year time periods, where growth appears to have slowed.

2.6 Behavioural enrichment

Providing captive animals with opportunities to display a range of species appropriate behaviours and to make behavioural choices that give them some control over their lives is among the goals of an animal husbandry approach known variously as behavioural enrichment. Enrichment research in both laboratories and zoos has shown the importance of providing animals with an ever changing or rotating array of stimuli and behavioural opportunities (Ben-Ari, 2001). It is important that environmental enrichment encourages natural and not unnatural behaviours.

Enrichment may serve various functions like (1) improving well-being by reducing the levels of abnormal and injurious behaviour, increasing exercise, satisfying behavioural needs and optimizing the level of stimulation that animals receive, (2) educating zoo visitors by increasing the levels of natural and interesting behaviours, visibility and activity levels and (3) conserving endangered species by improving the success of captive breeding and reintroduction programmes. A simple way of behavioural enrichment is variability in enclosure topography and vegetation. Because rhinos can be aggressive towards each other, planting (protected from rhinos), rock piles, dirt mounds and other forms of visual barriers (mentioned in chapter *2.2.1 Boundary*) may help ease social tension by partially blocking rhino sightlines. Mud wallows and rubbing posts are other simple enrichment items and are particularly important for skin health (Fouraker and Wagener, 1996).

Designing indoor holding so that each rhino must pass through a common area prior to its individual stall allows rhinos to consistently sniff and mark another's dung-piles. Within reason, it is recommended that dung piles not be totally removed during cleaning. This again allows rhinos to obtain information about each other using their well-developed olfactory ability. With vet and management approval, dung may be exchanged with other zoos and placed in the rhino enclosure. The novel dung may stimulate sexual activity or increase territory marking behaviour. If the institution houses more than one rhino species or subgroups the same effect may be obtained by exchanging dung in-house (Fouraker and Wagener, 1996).

Rhinos may also benefit from addition of various objects. Enrichment items must be designed with the following criteria in mind:

- Can not be swallowed
- Can not be torn or ripped
- Can not be crushed or broken
- Can not trap or entangle the animal
- Can not cut, poke or scrape the animal

Objects that can be used as enrichment items are listed in table 2.3.

Table 2.3: Enrichment items (Felts, 2007; Fouraker and Wagener, 1996).

PVC Pipe	Spool with treats	Barrel
Waterbath (pool)	Moose stool	Bamboo contraption
Boomerball	Bison stool	Logs
Hanging boomerball	Pronghorn stool	Hanging logs
Street brush	Spinna	Sprinkler
Keg	Top soil	Stumps
Spool	Smashed barrel	Browse
Complex enclosure	Rubbing posts	

Another form of enrichment is having miscellaneous food items hidden in the substrate. By varying the time and location of food this will help to keep the animals occupied. Be sure to hide food on hard surfaces that are covered with mulched branches to avoid ingestion of sand or pebbles while the animals are digging for the hidden items (Gulenschuh and von Houwald, 2002).

An old tyre could be used as an enrichment item as well, it should be cut open so that the tire cannot get stuck around the nose, horn or neck of the rhino.

2.6.1 Training

There are several learning principles, namely positive reinforcement, negative reinforcement, "positive" punishment and negative punishment. We refer to Gatz (1998) for more information about these principles.

Training or operant conditioning programmes may also serve as a form of enrichment. Numerous examinations, as well as numerous nutritional, reproductive and veterinary research projects, often require hands-on contact with rhinos. An alternative to manual or chemical restraint of an individual is an operant conditioning programme that utilises positive reinforcement. Such a programme has many benefits, including reduced stress to the animal, more reliable sample collection, reduction of any effects of stress on the samples and less need for structural modifications to restrain animals. There are several institutions that have successfully trained rhinos to do procedures like blood collection, ultrasound and skin and food care (Fouraker and Wagener, 1996).

Before beginning any training the first step is to establish training programme goals and requirements. Training rhinos will require much coordination among staff members including keepers, curators, veterinarians and zoo management. Exhibit schedules may be modified during training. All parties must understand that consistency in routine is inevitable for training. Modifications will undoubtedly be made to the pre-training routine to accomplish the training programme goals (Fouraker and Wagener, 1996).

When training programme goals and requirements have been established the training of the animals can begin. The training process will generally include three basic steps:

- habituating of the animal to the trainer
- constructing and introducing targets, or visual areas of ideal placement for the rhino
- establishing the commands necessary to steer the animal to these target areas

It is recommended that training starts with one individual as the primary trainer. Additional personnel may be included once the rhino reliably executes the desired behaviours of the primary trainer. The goal is that ultimately, given the appropriate stimuli, the rhino will execute the desired behaviours for a number of different personnel. It is recommended that the training initially be performed in a specific area of the enclosure, but later on, flexibility is important so that the rhino will perform the desired behaviours in more than one area if necessary. Training commands, targets and rewards should only be used during training sessions. Training commands and targets should be carefully evaluated prior to beginning the training programme. Most used commands in training sessions are: (Fouraker and Wagener, 1996)

- Move up; when a rhino needs to move forward
- Back; when a rhino needs to move back
- Over; when a rhino needs to do a side step
- Steady; when a rhino needs to hold its position
- Foot; when a rhino needs to present its foot
- Come; when a rhino needs to approach the trainer
- Target; when a rhino needs to place is head or body part at a specific area (e.g. on the target)
- All right; when the training is over and the rhino can do what it wants

Specific training areas and objectives will vary across institutions. Closed stall, free-stall and chutes work well for medical procedures, provided there is ample access to the animal and the safety for personnel (Fouraker and Wagener, 1996).

To habituate the rhino to the presence of the trainer, regular ten minutes training sessions may be effective. It should be emphasised that the amount of time required will depend on the tractability of the individual. The primary objective of these sessions is to establish trust. By noting generalised behaviours and body positions of the animal, the trainer should be able to notice when the animal is relaxed in the trainer's presence. At this point the trainer can begin shaping the desired behaviour. Each successive approximation of the desired behaviour should be rewarded with a command like "good", which serves as a bridge to link the behaviour to the reinforcement, which is given concurrently. A positive reinforcer should increase the frequency of the desired behaviour. Successful reinforcers are food (e.g. apple, bananas or bread) and, to a lesser extent, tactile stimulation (e.g. belly scratching). The bridge and reinforcer should only be given for the approximation of the desired behaviour. Otherwise, additional behaviours performed in conjunction with desired behaviour will also be reinforced (Fouraker and Wagener, 1996).

After successful completion of the approach behaviour the trainer can introduce a target, or object easily visible to the rhino. At this point the trainer should encourage the rhino to the target on command using the same basic procedure of reinforcing approximations of the desired behaviour. At this point training sessions should last about 10 to 30 minutes. Alignment with both head and body targets places the rhino in position for all kind of procedures like blood collection or rectal temperature readings. When the rhino successfully targets the target the next step is to encourage the rhino to remain stationary for a given period of time (using the command steady). Once these behaviours have been established the final step is to desensitise the rhino to medical equipment. Additional personnel who will be performing the procedure can be introduced to the training. Initially the collection area should be manipulated (e.g. touching and pinching or cleaning the colon of faeces). Any medical materials that will be used should be slowly introduced. These introductions should continue until the rhino shows no reaction to the equipment. The final stage prior to the actual procedure may include pressure from a blunt needle or insertion of a reproductive probe until the rhino shows no reaction (Fouraker and Wagener, 1996).

If at any point during the training there is regression, the trainer should revert to a point in the training where the rhino is comfortable and then slowly proceed again. This may add time to the total time needed for conditioning but the probability of the overall success is increased. Once the procedure is routine for the rhino the trainer should periodically lead the rhino in performing the desired behaviours if they are not otherwise performed regularly. In the absence of regular performance, this variable reinforcement will help prevent the behaviours from extinguishing (Fouraker and Wagener, 1996).

2.6.2 Crate training

Crate acclimation can require two to six weeks, however some institutions have reported that they have successfully crate-trained their rhinos in seven days or less. Training should be completed by a method of approximation.

The first step is to introduce the crate into an interactive part of the animal's environment, for example the door way, allowing the animals to go in and out and get used to it. Place the rhino's food in the crate to encourage this behaviour. Gradually introduce the front metal bars of the crate, and move the rhino's food gradually closer and closer to these, getting the animal used to them.

If the animal acclimates to the point of completely entering the crate and will allow the door to be shut, the door should be left closed for short acclimation periods under close observation, however this can take a long period of time (longer than six weeks) so it is recommended just to close the door shut on the actual day of transport. If the rhino does not completely acclimate to entering the crate, partial immobilisation (standing restraint) may need to be utilised for shipping. In situations in which crate training is not possible, immobilisation should be incorporated. Forced crating without training or immobilisation is strongly discouraged (Fouraker and Wagener, 1996).

2.7 Handling

2.7.1 Individual identification and sexing

Recommended methods for identification are documentation of characteristic marks like wounds and scars, and the use of microchips. Individual traits can be documented through photographs. If an animal is transferred, these records, or copies of them, should go with the animal to the new facility and the EEP coordinator. Microchips are also recommended as a primary identification method. They should be placed behind the left ear. Transponder identification numbers need to be reported to the studbook keeper (EAZA yearbook, 1995; Gulenschuh and von Houwald, 2002). Recommended method of sexing is by visual means.

2.7.2 General handling

Black rhinos can be kept hands-off or hands-on depending on the facility's policy and personality of the rhino. Preferably Black rhinos are maintained to allow a day-to-day, set-routine interaction which will facilitate medical and foot care, introductions, births and separations. It is not recommended that keepers enter the same enclosure space as Black rhino and that handling occurs through a barrier the gives the handler protection (Figure 3.7). For information about safety precautions we refer to paragraph *2.7.5 Safety*.

Figure 3.7: Handling should occur through a barrier ensure keeper protection at all times.

2.7.3 Catching / restraining

For physical exams as well as nutritional, reproductive or veterinary research projects physical restraint devices can be very valuable. Numerous institutions have constructed permanent physical devices to restrain their rhinos when necessary. These physical restraint devices are also called chutes. In general, it is highly recommended that institutions modifying rhino exhibits or constructing new ones incorporate a physical restraint area or device into their design considerations. Several physical restraint designs are effective for rhinos. In general, major restraint chute design considerations include strength, durability, type and function. It should be noted, that available space and animal's size and disposition vary across institutions and should be individually addressed. Both captive managers and researchers emphasise that the general restraint area should be an active component of daily rhino management. Methods to accomplish this vary. A restraint chute or restraint area can be designed so that the rhinos must pass through it to exit the barn into the yard. If rhinos are fed indoors, part of the feed can be offered in the chute area. Rhino chutes should be manufactured out of steel or a combination of steel and steel-reinforced wood. Steel-strength aluminium has also been used. Aluminium is lighter and more manoeuvrable than steel, as well as potentially less stressful to rhinos because of lower sound properties than steel (Fouraker and Wagener, 1996). It is important to give the rhino time to get used to the chute so that it is calm and relaxed while being restrained. Depending on the temperament of the individual this may take many months to accomplish.

Restraint chutes: Permanent pass-through indoor restraint chutes are especially effective for rhinos. The chute should allow restraint of the animals when it is passing through in either direction so that shifting routine of the animal is not interrupted. The width of the chute should limit side-to-side movement while still allowing the animal to comfortably lie down. However, animals can become wedged in tight-fitting chutes if the side cannot be released. To alleviate excessive forward movement of the animal when it lowers its head, two vertical bars that push in from sides of the chute to the shoulder of the rhino may be utilised. Quick release of these shoulder bars often relieves agitated animals without having to release them completely (Fouraker and Wagener, 1996).

High-walled chutes: High-walled chutes or bars over the top keep the animals from climbing or rearing up. Horizontal bars in the chute's entry gates and sides are hazardous for examiners when the animal lies down. Vertical bars on the sides can trap researchers' arms if the animal can move forward. If the animal's movement forward and side-to-side mobility can be limited, vertical bars or walls on all sides are recommended. The distance between these bars along the sides of the chute should be great enough to prevent the animal's foot from becoming wedged if the animal rolls on its side in the chute. For researcher safety, this distance can be divided with removable vertical bars (Fouraker and Wagener, 1996).

Closed chute: A closed chute is another option that has been used successfully. A typical closed chute has both front and back gates. The back gate restricts the rhino's movement by sliding forward. The hind end of the rhino is supported by a v-design that prevents it from lying down. This design also allows additional safety for the staff while working with the animal. In many respects, a closed chute does not depend as strongly on conditioning of the rhinos as does a squeeze chute, though acclimation is recommended prior to attempting any treatments within the chute. The design of a closed chute might necessitate an outdoor location in most cases, thus the use of this type of chute may be limited by weather (Fouraker and Wagener, 1996).

Free-stall chute: A free-stall chute can be used for animals more sensitive to a confined enclosure. The design of this type of chute allows the rhino to enter or exit at its will and thus may help to keep rhinos calmer during procedures. Because there is free access rhinos must be conditioned to target or stand still. A free-stall design can easily be incorporated into an existing pen or stall, indoor or outdoor. As stated, the open back of this type of chute allows the animal to enter and leave the structure at will. Protection of staff when working with the rhino is important, and a partial back wall constructed of vertical pipes allows staff to step out of the way (Fouraker and Wagener, 1996).

Sliding gates: Sliding gates are safer than swinging doors because rhinos may slam swinging doors. A rectangular opening in these gates for performing palpation should not pin the arm of an examiner when the animal is shifting. The distance between the vertical sides of this rectangular opening must be wide enough for researcher safety while still limiting the space through which a rhino could squeeze. Also the horizontal bottom bar of this rectangle should be only a few inches from the ground, as animals frequently lie down. Solid doors on the outside of these gates can be used to stop rhinos, as they may attempt to charge even small openings. Additionally, good lightning and accessible electrical sources are useful (Fouraker and Wagener, 1996). Guillotine gates are not recommended.

Immobilisation: Besides early crate training prior to transportation, immobilisation offers a fairly simple way of crating a rhino. The usual pre-immobilisation should be observed for any procedure requiring the use of chemical immobilisation/tranquilisation agents. For rhinos, etorphine (M-99, Large Animal Immobilon) remains the drug of choice although several alternatives are available. The duration of immobilisation without administration of an antagonist may range from 30 minutes to two hours (Fouraker and Wagener, 1996). For more information about specific drugs and immobilisation we refer to section *2.8 Veterinary* (Fouraker and Wagener, 1996). Other drugs used for immobilisation in combination are butorphanol, detomidine, xylazine and ketamine.

Following crating, all rhinos should be held for 24 hours at the loading location for observation, or accompanied by a veterinarian during transport. This is necessary because renarcotisation is common in hoofed animals, especially rhinos, given opioids. Trained personnel should be present to administer the correct reversal agent(s) in the likely event of renarcotisation. Any other complications of crating can be managed more easily and effectively in-house rather than en route (Fouraker and Wagener, 1996).

Several principles should be followed to increase the safety of chemical restraint procedures for both the animals and personnel:

- When applicable, antagonists to the restraint drugs should be prepared prior to the initiation of the procedures and should be available for rapid administration.
- Careful monitoring of the patient (auscultation, ECG, pulse-oximetry, etc) will help to rapidly identify problems should they develop and allow early intervention.
- The large size of an adult rhinoceros may result in further complications during anesthetic procedures. Efforts should be made to maintain the animal in sternal recumbancy when possible to minimize respiratory complications, and if the procedures is to last more than 30 min. efforts should be made to “pad” the area under the animal (with mattresses, inflated inner tubes, straw/hay bedding, etc.) to minimise the effects of pressure on the limbs (Fouraker and Wagener, 1996).

The kind of catching/restraining that is preferred is physical restrain or chemical restrain. When a rhino is being caught/restraint risk of injuring the horn needs to be taken into account.

2.7.4 Transportation

It is recommended to transport Black rhinos by truck or airplane in a crate. Each method has its advantages and each should be carefully considered and evaluated concerning the distance to be travelled, the personnel needed and the temperatures to which the animals will be subjected (Fouraker and Wagener, 1996).

Typical problems that can occur in shipping include the following:

- Animals destroying and or climbing out of the crate top
- Animals becoming inverted in the crate and unable to right themselves
- Animals destroying end panels or doors, resulting in eye, horn or facial injuries
- Prolonged excessive exertion resulting in hyperthermia and/or myopathy

Truck: When transporting Black rhinos by truck, open trailers should be protected from excessive wind, rain and sun. In extreme hot or cold temperatures an enclosed trailer is an option. In any case, the vehicle must be climate controlled.

Airplane: The International Air Transport Association (IATA) has made the IATA Live Animals Regulations (LAR). These Live Animal Regulations are a Worldwide Standards for transporting live animals by airlines. The objective of the IATA Live Animals Regulations is to ensure that all animals are transported safely and humanely by air, whether it is to transport a pet, an animal for zoological or agricultural purposes or for any other reason *(IATA, 2007).*

The IATA Live Animals Regulations are applicable to members of the International Air Transport Association according to the provisions of Cargo Services Conference Resolution 620 and to airlines being parties to the IATA Multilateral Interline Traffic Agreement-Cargo *(IATA, 2006).*

The IATA Live Animals Regulations are accepted by the Convention on International Trade in Endangered Species of Wild Fauna and Flora (CITES) and the Office International de Epizooties (OIE) as guidelines in respect to transportation of animals by air. These regulations have been used by the Council of Europe as a basis for its code of conduct for the international transport of farm animals. The European Union has adopted the IATA Live Animals Regulations as the minimum standard for transporting animals in containers, pens and stalls. As an increasing number of countries adopted or accepted these regulations as a part of their national legislation, shippers are warned that shipping live animals in violation of the regulations may constitute a breach of the applicable law and may be subject to legal penalties.

The IATA Live Animals Regulations container requirement 71 concerns the rhino species.

Transport box / crate: The IATA Live Animals Regulations container requirement 71 states the requirements of the transport container that is applicable to transport of Elephants, Hippopotamus and Rhino species (IATA, 2006).

Material and dimensions: Materials that can be used to construct a transport container, according to the container requirement 71, are metal and hardwood. The transport container should be big enough to restrict the movement as well as restrain the animal in question. The animal must be able to stand naturally without being cramped but must not be able to move freely (IATA, 2006).

Container dimensions should be determined by the animal's size. In general, the container should be 30 cm longer and wider than the animal when it is lying on its side. Approximate container dimensions are 350 cm in length, a height of 191 cm height, and 140 cm wide to prevent the animal from turning around (Fouraker and Wagener, 1996).

Frame and slides: The frame of the transport container should be made out of strong metal welded or bolted together depending on the weight of the animal. Solid hardwood sides, with no internal projections, must line the outer framework for extra strength. All woodwork must be secured with bolts and nuts that face the exterior so that they can be easily tightened from the outside. Spring steel weld mesh can also be used in combination with strong metal corner posts, together with a rigidly braced top and sides. In either case the lower part of the sides must be solid and leak-proof. A heavy plastic foil or tarpaulin covered with sufficient absorbent material which is tied up half a meter around the crate can be used (IATA, 2006).

Floor: The floor must be made of thick tongue and groove of at least 2.5cm (1in.) thickness or its equivalent and have a non-slip surface. It must be completely leak-proof (IATA, 2006).

Roof: The roof must be solid over the animal's head and shoulders and slatted over the loins and hindquarters to give good ventilation (IATA, 2006).

Doors: A series of metal bars must be bolted to the top and bottom of both the entry and exit of the container. Exterior to these bars sliding or hinged solid hardwood entry and exit doors must be made to completely cover the entry and exit. The doors must be fastened by a sufficient number of strong bolts which must be able to resist the weight of the animal. The upper third of both doors must have ventilation openings. Entry and exit must be clearly marked as such (IATA, 2006).

Ventilation: Through the slatted or louvered upper third of both wooden doors and the slatted portion of the roof there should be adequate ventilation (IATA, 2006).

Feed and water containers: The water container must be fixed in the front of the container. It must be made of strong metal and wide enough for the animal's muzzle to enter. The edges of the water trough should be smooth so the animal cannot hurt itself. For feeding, outside access can be from a low wooden flap, clearly marked "Feeding" at the base of the door. Food can be placed between the bars and the door. The access flap must be securely closed when not in use (IATA, 2006).

Forklift extrusions: Forklift extrusions must be provided as an integral part of the design (IATA, 2006). An example of a container that can be used for rhinos is shown in Figure 3.8.

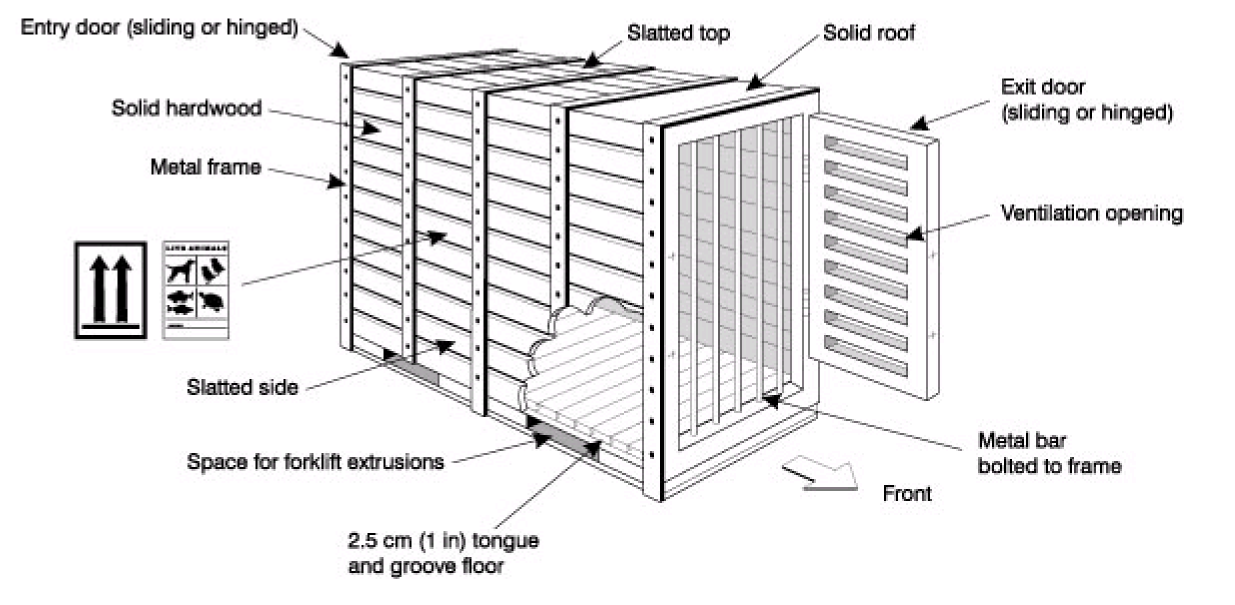

Figure 3.8: Example transport container for rhinos (IATA, 2006).

An ample amount of absorbent material such as wood shavings is required for bedding. The animal must be watered before shipment. Animals do not normally require additional feeding or watering during 24 hours following the time of dispatch. If feeding or watering is required due to an unforeseen delay, instructions supplied by the shipper must be followed (IATA, 2006).

It is recommended that all shipments of these species be accompanied by a person/veterinary and go through a crating training well before dispatch (IATA, 2006). We refer to section *2.6 Behavioural enrichment* for more information about crate training. A light sedation is recommended for transports taking longer than a couple of hours (Fouraker and Wagener, 1996).

Markings on transport container: The markings on the transport container must be durable and printed or otherwise marked on or affixed to the external surface of the live animal container. English must be used in addition to the language which may be required by the state of origin (IATA, 2006).

Unless otherwise specified in these Regulations, each live animal container must be marked, durably and legibly on the outside of the container, with each of the following:

- The full name and address and contact number of the shipper, consignee and a 24-hour contact (if it is not one of the aforementioned persons responsible for the shipment).
- The scientific and common name of the animal(s) and quantity of each animal contained in the container, as shown on the shipper's certification.
- Containers carrying animals which can inflict poisonous bites or stings must be boldly marked "POISONOUS". Aggressive animals or birds that can possibly inflict injury through the bars or ventilation openings of the container must have an additional warning label "This Animal Bites".

- Affix special feeding and watering instructions to the container.
- In general, tranquillisation is not advocated for the transportation of live animals. However, certain wild species require the use of such medication. Whenever used, they must be administered under competent supervision and the name of the sedative, time of administration and the route of administration must be clearly marked on the container and a copy of the record must be attached to the documents relating to that shipment. Any further medication administered must be recorded and accompany the shipment with the name of the sedative, time of administration and the route of administration (IATA, 2006).

It is mandatory to attach at least one IATA “Live Animals” or one “Laboratory Animals” label or tag, properly completed, to each live animal container, unless otherwise stated in the individual container requirements. Animal containers may have the appropriate labelling imprinted (IATA, 2006). The label for live animals should have the following header “Live Animals”, the colour should be bright green on a light background. The minimum dimensions of the label are 10 cm x 15 cm and letters of 2.5 cm (IATA, 2006). In Figure 3.9 the label for live animals is shown.

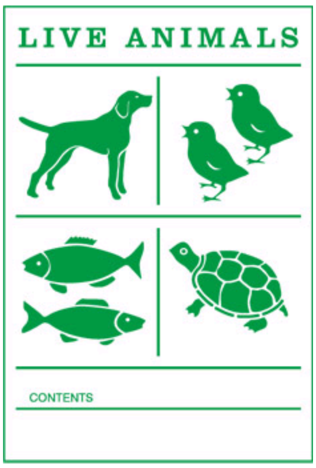

Figure 3.9: Live animals label (IATA, 2006).

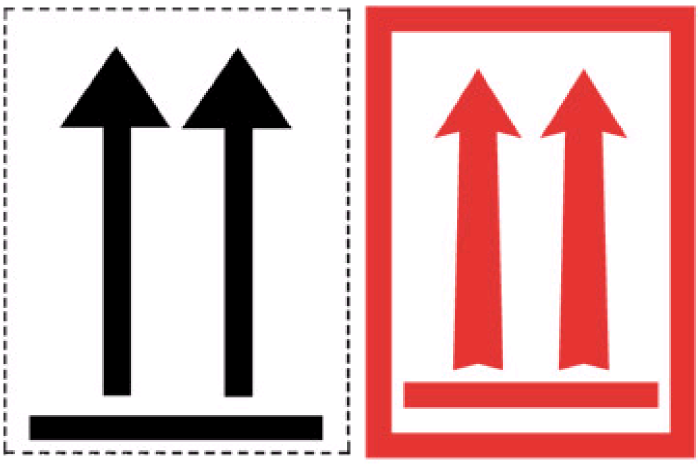

Figure 4.0: This way up label (IATA, 2006).

In addition to the “Live Animals” label, it is mandatory that the “This Way Up” labels or markings be placed on at least two opposite sides. Labels may be imprinted on the container. The label for “This way up” should be black or red on a contrasting background. The minimum dimensions of the label are 74 mm x 105 mm and letters (IATA, 2006). In Figure 4.0 the label for “This way up” is shown.

2.7.5 Safety

In order to ensure safety and to properly meet the requirements of management it is recommended that more than one keeper be responsible for these animals on a daily basis. Keeper interaction should be restricted to designated areas and should be conducted in accordance with institutional protocols. Consistency of routine is vital in daily interaction (Fouraker and Wagener, 1996). Keepers should always carry a radio and / or a mobile phone when working with rhinos.

2.7.6 Stress

Moving animals between institutions is an important aspect of population management, but could also be considered as a potential stressor, both in terms of the transport involved and the novel social and physical environment into which the rhino is moved. Hormone analysis can also be used as a tool to investigate the role of potential stressors on adrenal activity, by measuring the steroid hormones glucocorticoids. The adrenal response to a stressor in the first instance is not a bad thing, but demonstrates that the animal is responding accordingly to a potential threat. However, if stressors are particularly severe, or are prolonged, then negative consequences on health and reproduction could result.

To investigate short-term or long-term effects of translocation, hormone analysis was conducted following the translocation of four male and five female black rhinos between European institutions between 2008 and 2012. Although an adrenal response to translocation was observed in some individuals following inter-zoo transfer, this was not apparent in all individuals. Furthermore, following these translocations, there was no evidence of oestrous cycle disruption; three out of five females were sexually mature and oestrous cycles continued post-translocation, one has since produced a calf. The remaining two females were not yet cycling prior to translocation, but one of those since commenced regular cyclicity and has now produced a calf. In males, no consistent differences in testosterone concentration were observed post-transfer, and one male sired a calf approximately twelve months post-transfer.

Behavioural indicators for stress are animals with head up and looking around or running with tail up, pacing during the night; which is evident by the night bedding being spread around the night area, abnormal amount of moving and pacing and aggression against fence, person etc.

There are several causes for stress in Black rhinos and these should be avoided:

- inability to escape entirely from other Black rhinos
- boredom
- inability to display all of their natural behaviour patterns
- stressed by their environment by public, machinery, etc.
- lack of visual barriers
- permanent separation of mother and calf before age of 18 months

Stereotypical behaviour mentioned by the experts are walking back and forth, caused by not enough hiding possibilities and running on one path only, it is not known what causes this behaviour.

2.8 Veterinary: Considerations for health and welfare

In captivity Black rhinos occasionally display unusual disease syndromes not described in the wild (Dennis et al., 2007). Black rhinos appear to be more susceptible to a variety of diseases (Fowler and Millar, 2003). Regarding illness the behavioural repertoire of rhinos is often quite limited. Depression, inappetence and loss of body condition are often the only signs of major disease problems (Fouraker and Wagener, 1996; Reuter and Adcock, 1998). In this chapter, basics of medical procedures, diseases and disorders concerning the Black rhino are described.

2.8.1 Medical procedures

Blood collection: blood collection from rhinos can take place from several sites. The most commonly used is the ear vein, which is sufficient for collecting small quantities of blood and for intra venous (IV) injections. A larger medial (radial) vein in the hind leg has been identified and allows collection of larger amounts (one to eight 8 litres) of blood (Fouraker and Wagener, 1996). Best preferred is the medial aspect of hind legs (vena metatarsalia) in well trained animals in a training cage. For blood values we refer to Appendix II. In Figure 4.1 a picture of blood collection can be seen.

Figure 4.1: Blood collection (Göltenboth and Klös, 1995).

Neonatal examinations: although it is often impossible, neonatal examinations could be performed. These should include weight, a dipstick blood glucose, total solids, CBC, sera chemistry profile, sera/plasma for vitamin-E levels and, when possible, stored sera. Examinations may include vitamin supplementation and the placement of an identification transponder (Fouraker and Wagener, 1996).

Catheter: as the cervical canal is long and tortuous and characterised by interdigitated folds, the passage of catheters is extremely difficult, although it has been achieved in Black rhinos under ultrasonic guidance. At least one attempt to dilate the cervix with prostaglandins and estradiol was unsuccessful. Examination of the female reproductive tract in adult rhinos may be attempted with standard ultrasound units and aided by appropriate equipment modifications (Fowler and Millar, 2003).

2.8.2 Diseases

Following is a list of reported rhinoceros diseases; tuberculosis, gastrointestinal torsion and impaction, encephalomyocarditis infection, oral and skin ulcers, haemolytic anemia, fungal pneumonia, encephalomalacia, tissue accumulation of iron, creosote toxicosis, skin problems, mouth and nasal bleeding polyps, nail and foot problems, and leptospirosis.

Gastrointestinal torsion and impaction: gastrointestinal torsion and impaction have been reported in Black rhinos with signs similar to those of colic in the horse. Gastric ulcers are commonly seen in the post-mortem of old rhinos and in ill rhinos. Protective medication should be considered in chronically ill individuals (Fouraker and Wagener, 1996).

Encephalomyocarditis infection: death following infection with encephalomyocarditis virus has been noted in two Black rhinos. Vaccination for the equine encephalitides is not routinely practiced (Fouraker and Wagener, 1996).

Haemolytic anemia, idiopathic haemorrhagic vasculopathy syndrome (IHVS): the initial signs of haemolytic anemia or IHVS are usually limited to depression, sometimes lameness followed by hemoglobinuria and bleedings from nose and mouth. The progression of the cases is usually acute, death often occurring within 48 hours of the initial signs. Inducing this proposed multifactorial disease different factors as viral, bacterial, vitamin deficiency and stress are discussed (Göltenboth and Klös, 1995). Current research indicates that the red blood cell (RBC) of the Black rhino is inherently energy deficient and thus unstable and susceptible to haemolysis (Fouraker and Wagener, 1996). The diet of Black rhinos is suspected of attributing to the problem of haemolytic anemia (Norstrom, 2004).

Hemolysis: due to the predilection to hemolysis in the Black rhino, Dr Donald Paglia of the University of California at Los Angeles has suggested avoiding exposing them to drugs and compounds that are known to induce hemolysis in enzyme-deficient human populations. All of the following compounds should be avoided.

- Pharmaceutical compounds: antimalarials, sulfonimides, sulfones, nitrofurans, acetanilide, chloramphenicol and some vitamin-K analogs
- Chemical compounds: wood preservatives, rodent-control poisons and other pesticides, strong cleansers particularly those containing naphthalene
- Food: favabeans

Other drugs have been associated with hemolysis but with an uncertain or doubtful role. These drugs include aspirin, phenacetin, aminopyrine, acetaminophen, probenecid, vitamin C, dimereaprol, p-aminosalicylic acid and 1-DOPA. Any exposure to creosote should be avoided. In view of the hemolysis induced in horses by the consumption of certain oak and red maple leaves, as well as wild onions and members of the Brassica (kale) family in other domestic species, consumption of these species should be avoided (Fouraker and Wagener, 1996). A long term treatment with chloramphenicol in Black rhinos had no impact on urine quality or a haemolysis. Continuous vitamin E and C supplementation is recommended as prophylaxis especially during the winter (Göltenboth and Klös, 1995).

Salmonella: Salmonella has been reported to cause both enteritis and sepsis in Black rhinos. Treatment has been attempted in several cases with parenteral antibiotics and fluids but has not often been successful. *Pseudomonas ssp*. and *E. coli* have been reported as the cause of enteritis. *Pseudomonas pyocyanea*, *Campylobacter coli* and coliform infections have been reported in hand-reared rhinos. *Yersinia pseudotuberculosis* has also been reported in young, hand-reared rhinos to cause enteritis and mesenteric lymph node enlargement (Fowler and Millar, 2003).

Leptospirosis: leptospirosis has been associated with some cases of primary hemolytic anemia. In at least one Black rhino leptospirosis caused renal compromise in addition to haemolytic anemia and responded to treatment with oral ampicillin (Fowler and Millar, 2003). The only vaccination routinely recommended is the biannual administering of Black rhinos with either a 5-way leptospiral bacterin (containing *Leptospirosis interrogans serovar icterohaemorrhagiae, - grippotyphosa, - pomona, - canicola, - hardjo)*, or a 6 way bacterin containing a 5-way leptospiral bacterin and *Leptospirosis interrogans serovar bratislava*. It should be noted that injection site abscesses are relatively common (5 to 10%) (Fouraker and Wagener, 1996).

Clostridial disease: although there are only two reports of clostridial disease (one case of tetanus and one of *Clostridium sordelli*) vaccination for these diseases may be considered (Fouraker and Wagener, 1996). There is debate over whether is recommended to vaccinate or not for clostridial disease.

Tuberculosis: infections with both *Mycobacteria tuberculosis* and *Mycobacteria bovis* have been reported in captive rhinos. The presentation of tuberculosis is emaciation, although coughing and dyspnoea may occur before death. At post mortem testing examination the respiratory system is most commonly affected with both focal and diffuse granulomes. Pre mortem testing may be attempted with intradermal tuberculin in the eyelid, tail fold, or the skin at the base of the ear or pinna. Comparative testing in the tail fold, repeated ten days later in the neck if a suspect reaction occurs in the tail is recommended (Fowler and Millar, 2003). In a male Black rhino at Berlin zoo the ElephantTB STAT-PAK Assay (CHEMBIO Diagnostic Systems, Medford NY, USA) antibody test kit was used after collecting blood with the help of assassin bugs (Ochs, 2013 unpublished).

Rabies: it would be anticipated that all rhino species are susceptible to rabies. Although rabies has been reported in an Asian rhino in India the lack of subsequent reports suggests that this is not a common occurrence (Fowler and Millar, 2003).

Fungal pneumonia: fungal pneumonia had been reported in at least nine Black rhinos. Nearly all the cases have involved infection with *Aspergillus sp.*, and at least five of these have followed corticosteroid therapy, sometimes even relatively low doses administered over short treatment periods. Fungal pneumonia should be considered in all Black rhinos with signs of respiratory illness *(Fouraker and Wagener, 1996).*

Encephalomalacia: encephalomalacia has occurred in four young (two months to two years of age) female Black rhinos. In three cases, it presented as acute and profound stupor. Two of those calves died within four days of onset, but a third lived and became a “dummy” calf that was later euthanised. The fourth rhino, a two year old, became hyper excitable and then depressed. The histological lesions were those of profound leucoencephalomalacia, and the etiology remains unknown. These cases emphasise the importance of collecting brain and central nervous system tissue on all rhino necropsies (Fouraker and Wagener, 1996).

Tissue accumulation of iron: adult Black rhinos appear to accumulate iron, particularly in their livers. These lesions are not those of a primary iron-storage disease, but similar to those of chronic iron exposure. Further studies may help determine whether the iron results from chronic sub clinical haemolysis or from dietary causes (Fouraker and Wagener, 1996).

2.8.3 Disorders

Parasites: parasites have been of low frequency and are usually not associated with clinical signs in captive rhinos. In newly captured rhinos, consideration should be given to hemic and skin parasites as well as fecal ones. A biannual fecal examination for parasites is adequate in rhinos established in captivity. In newly arrived rhinos blood examinations should be performed for hemic parasites (e.g. *Babesia sp*., trypanosomes, theileriasis and leishmaniasis). Skin lesions in wild caught Black rhinos should be biopsied and examined for the presence of *Stephanofilaria dinniki.* The most commonly found endoparasites have been tapeworms. In rhinos established in other captive collections other endo- and ectoparasites have been unusual. If fecals contain parasites treatment should be based on accepted horse parasiticides that possess a wide safety margin. If hemic parasites are found, treatment should be based on those employed previously. Treatment of ectoparasites in newly arrived rhinos has consisted of the application of coumaphos (Fouraker and Wagener, 1996).

2.8.4 Common injuries and treatments

Skin: Skin lacerations are relatively common occurrences and unless deep and/or badly apposed, they normally heal well without significant medical intervention. As noted above it is difficult to differentiate abrasions over points of wear from early skin ulcers. They should be closely monitored.

Horn: Horn avulsions occur with some frequency. The use of horizontal bars under which rhinos can hook their horns should be avoided. There can be notable haemorrhages from the base of the horn, but these usually stop with little or no intervention. Treatment is generally limited to the topical application of antibiotics and non-toxic fly repellents (Fouraker and Wagener, 1996). In Figure 4.2 an example of a partial horn avulsion can be found.

Skin conditions: skin conditions have been noted in Black rhinos. A syndrome of oral and skin ulcers of unknown etiology had been frequently noted in captive Black rhinos. Fractured or avulsed horns are a frequent entity. Treatment generally consists of topical antibiotics and non-toxic fly repellents, and the haemorrhage generally stops over a period of several hours. Abscesses at the coronary have been reported and responded to the application of hot water and topical treatment (Fouraker and Wagener, 1996). In one case, when rhinos were housed in an enclosure with no 'mud bath' dry skin spots developed, these were treated by routinely applying baby oil.

Foot problems: the feet of a rhino should be carefully monitored. When enclosures are not cleaned properly urine and faeces remains can cause problems between the nails and the sole. Deep cracks may occur between the sole and the central nail. These cracks can take a long time to heal and could cause paralysis in the foot. In severe cases the sole could rib (Göltenboth and Klös, 1995). In Figure 4.3 an infected hoof can be seen.

There was no specific information found about the euthanasia of Black rhinos. For more information about immobilisation of Black rhinos we refer to paragraph *2.7.3 Catching / restraining*. For more information about diseases and medical procedures we refer to Göltenboth and Klös (1995) and Fowler and Miller (2003).

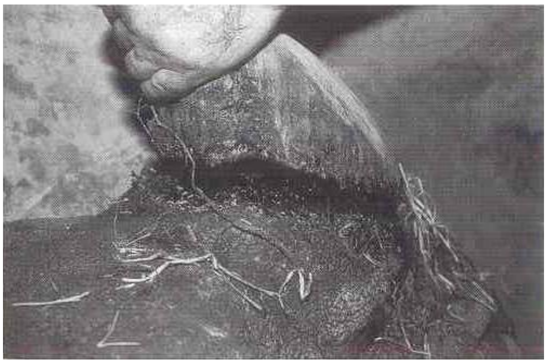

Figure 4.2: Partial Horn avulsion (Göltenboth and Klös, 1995).

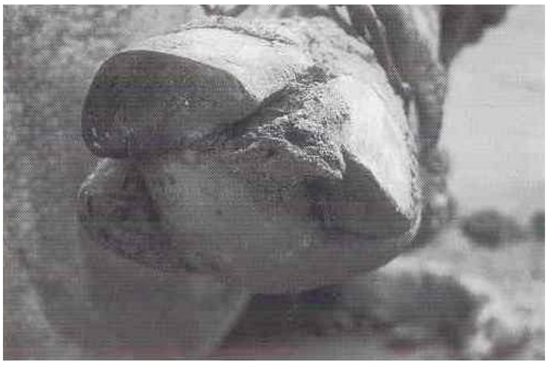

Figure 4.3: An infected hoof (Göltenboth and Klös, 1995).

3 Glossary

AZA	American Zoo Association
CITES	Convention on International Trade in Endangered Species
EAZA	European Association of Zoos and Aquaria
EEP	European Endangered Species Program
IUCN	International Union for Conservation of Nature
JAZA	Japanese Association of Zoos and Aquaria
IATA	International Air Transport Association
SADC	South African Development Community
SSCJ	Species Survival Committee in Japan
SSP	Species Survival Plan
TAG	Taxon Advisory Group

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Appendix I: Criteria for the body condition scores.

	CONDITION Assessment site	Numerical scale Descriptive scale	5 excellent (heavy)	4 good (ideal)	3 fair (average)	2 poor (thin)	1 very poor (emaciated)
A	Neck	General appearance	thick, well muscled, rounded	well muscled, rounded	rounded	flat, narrow neck; nuchal ligament visible	narrow, angular (bony) neck; nuchal ligament prominent
		Prescapular groove		-	slightly visible	obvious	deep groove very obvious
B	Shoulder	General appearance	well-muscled, rounded	rounded	flat	flat, slightly angular (bony)	angular, bony
		Scapula	covered	covered	spine visible	obvious	very obvious
C	Ribs		well covered (skin folds)	covered (skin folds)	visible	obvious	very obvious
D	Spine	General appearance	rounded	slightly angular	back groove back visible	groove deep obvious	back groove very obvious
		Spinous processes	covered	slightly visible	visible	prominent	very prominent
E	Rump	General appearance	well rounded	flattened	slightly concave	concave	obvious depression
		Bony protuberances	covered	slightly visible	visible	prominent	very prominent
F	Abdomen	General appearance	distended, taught	filled	slightly tucked in	tucked in	tucked in
		Flank-fold	none	sometimes slightly visible	slightly visible	visible	obvious
G	Tail base		rounded (bulging)	rounded	narrow	slightly bony	very thin and bony

Appendix II: Blood values

Table II.1: Hematology values in the Black rhino (Fowler and Millar, 2003).

Parameter	Value
WBC x 10^3/ul	8,42
RBC x 10^6/ul	4,01
HBG g/dl	12,0
Hct %	33,4
MCV Fl	85,7
MCH pg/cell	30,5
MCHC g/dl	35,7
Platelets x 10^3/ul	284
Nucleated RBC/100 WBC	0
Reticulocytes %	1,6
Neutrophils x 10^3/ml	5,24
Lymphocytes x 10^3/ml	2,48
Monocytes x 10^3/ml	0,43
Eosinophils x 10^3/ml	0,25
Basophils x 10^3/ml	0,17
Neutrophilic bands x 10^3/ml	0,27

Table II.2: Serum mineral values and blood gases in the Black rhino (Fowler and Millar, 2003).

Parameter	Value
Ca mg/dl	12,7
P mg/dl	4,8
Na mEq/L	133
K mEq/L	4,7
Cl mEq/L	96
Bicarb mEq/L	23,3
CO^2 mEq/L	25,4
Iron ug/dl	227
Mg mg/dl	3,34

Table II.3: Blood chemistry values in the Black rhino (Fowler and Millar, 2003).

Parameter	Value
BUN mg/dl	13
Creatinine mg/dl	1,1
Uric acid mg/dl	0,5
Bilirubin mg/dl	0,3
Glucose mg/dl	69
Cholesterol mg/dl	102
CPK IU/L	255
LDH IU/L	595
Alk Phos IU/L	80
ALT IU/L	16
AST IU/L	85
GGT IU/L	27
Total protein g/dl	7,6
Globulin g/dl (electrophoresis)	4,9
Albumin g/dl (electrophoresis)	2,6
Fibrinogen mg/dl	104

Appendix III: Faecal collection protocol

Faecal Collection Protocol for Routine Hormone Analysis

For samples to be useful for reproductive monitoring, it is extremely important that they are collected regularly.

- For reproductive monitoring in **females, every other day** is required.
- For reproductive monitoring in **males, weekly** samples are sufficient.

Collection of Faeces

Identification:

The most important requirement is that that you know which animal the sample came from.
- If the animals are not housed separately you will need to observe the animal defecating and collect the sample as soon as possible.
- If you think you may have a problem with identification of faeces it is possible to mark the faecal samples by feeding a marker.

Contamination:

Avoid faeces that are contaminated with urine
- Urine has hormones too, and this interferes with measurements of faecal hormone concentrations.
- Please also ensure that the faeces are not contaminated with another individual's sample (faeces or urine).

Collection:

Once you have properly identified the sample, try to collect the sample as soon as possible following defecation
- Hormone concentrations in samples left exposed to the environment for extended periods will decrease the accuracy of measures.
- If animals are housed individually, it is not necessary to observe defcation – collecting the freshest sample from the over-night enclosure first thing in the morning is sufficient.
- There is no need to collect the entire faecal pile/bolus. Instead (as 'pockets' of hormone concentrations can be found within the faecal sample) turn zip-lock bag inside out and collect several (3-4) 'sub' samples from different areas of the same faecal sample.
- In total, collect approximately a handful sized amount of faecal material into the zip-lock bag.
- Label the bag using a <u>waterproof permanent marker</u> (i.e. Sharpie® pen) with:
  - **Animal's name**
  - **Species**
  - **Date (day/month/year)**
  - **Time (whether am or pm)**

Please note - sample labelling will be easier if zip-lock bags with a write-on panel are used – once samples have been frozen, writing directly onto the plastic bag or using an unsuitable pen can wipe off easily, making samples un-useable.

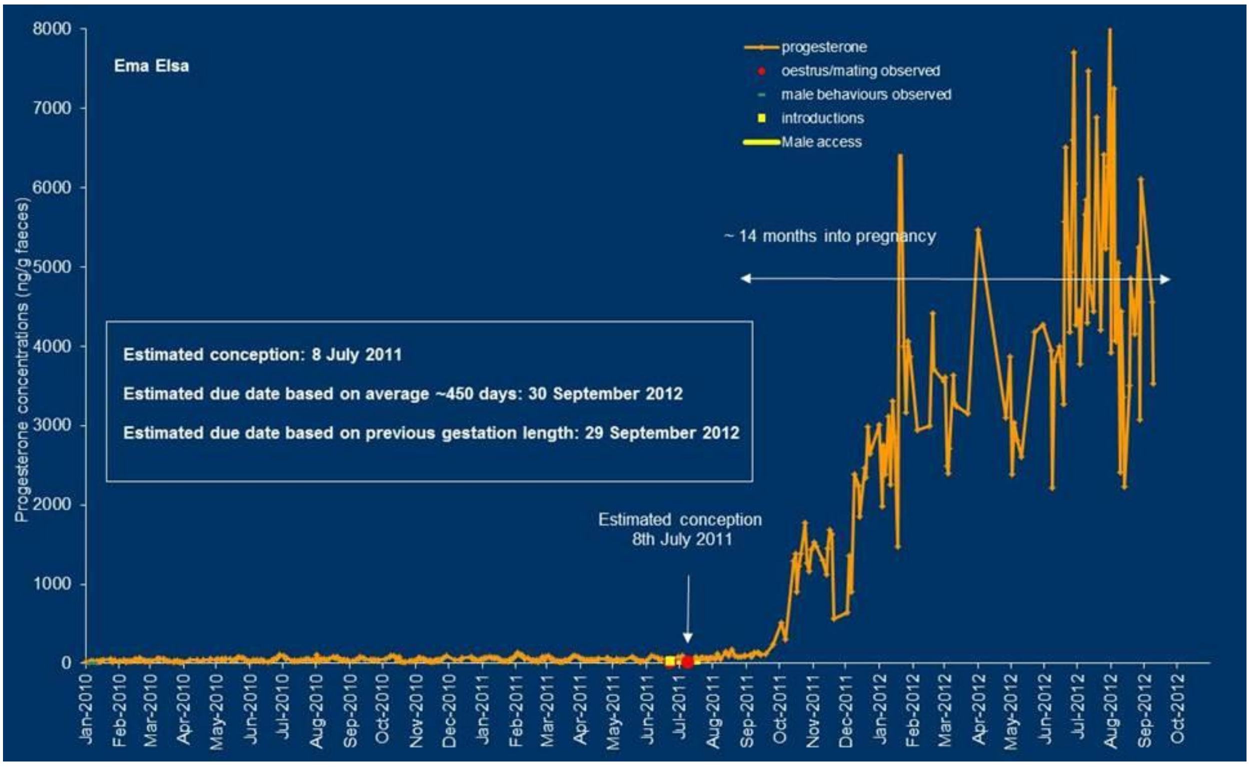
Storage:

Store samples as soon as possible after collection in freezer at -20 ^{o}C
- Hormones concentrations will degrade if samples are left out too long – please try to freeze within an hour of collection.

Appendix IIII: Black rhino birth plan: Ema Elsa, Chester Zoo, 2012

When is she due?

Timing of parturition, how do we know, what is the window?
Can we predict it more accurately (do we need to?)



Prior to the Birth

1. Enclosure preparation	Ema will have access to 2 pens both with beds, sand bed in the smaller pen
2. Changes in routine	No change except possibly shut in at night
3. Diet	No change until Calf born then increase Zebra pellet from 2kg daily to 4kg daily and ad lib lucerne

The Birth itself

1. Normal Birth Data	The following data was all obtained from the AZA husbandry manual 1996. • 3 days to a week, female starts bagging up • Within 24hrs area around eyes goes very dark and sweats – didn't happen with Kitani before either birth or Ema Elsa prior to her previous birth • Mother very restless 24 hrs before • Birth usually at night or in early morning.

	• Calf usually born within 10-12hrs of waters breaking (but can be longer in first time mothers) Generally around 4 hrs • Calf usually born head first • Birth weight 24-41kg • House won't be cleaned, food just quickly put in and water checked, house to be closed to public
2. Have problems been seen at CZ or other institutions?	No problems with parturition noted in the AZA manual or from Paignton's enquiries of other UK zoos with Black rhino.

3. What monitoring of the birth do we plan to do?	• As Ema Elsa previously had no problems with birthing, it would appear that there is little to be gained from staff being on site during the night. It was agreed by all present that we would not do night watches.
	• Recording footage of the birth however is desirable for two reasons:
	    • To gather data on the birthing process (with a view to contributing to the EEP husbandry guidelines and to get an accurate birth time to assist in decision making – see later)
	    • To use footage to publicise birth.
	• Cameras are installed and working in Old Rhino House pens. Low level lighting is used to improve image. (IR camera is available if required). All 3 camera outputs are continuously recorded on a hard drive.
	• Camera footage can currently be viewed live on monitor in old rhino house
	• During the day if it is not obvious calf has fed, they can be observed from a distance with binoculars as may not beable to see from the cameras

4. What are the indications, if any, for intervention during the birth	• All staff should stay away from the enclosure during the birth as it is quite likely that any disturbance will delay the process and may well lead to decrease chances of survival of the calf. • We can only envisage intervening if the calf was obviously stuck (ie part sticking out) and the mother visibly exhausted. – In this case we would need to consider anaesthesia to pull the calf out. We discussed use of the chute but if the cow was in distress it is unlikely she would enter it. Vet team to produce contingency plan for this eventuality. OR • If the dam's waters had broken >16hrs with no calf born (we may try and administer oxcytocin - vet team to develop contingency plan).

After the birth – rhino issues

1. Normal calf data	The following data was all obtained from the AZA husbandry manual 1996. • Birth weight 24-41kg • Calf normally stands 30min – 5hrs post birth. • Calf normally suckles 1-2hrs after birth (longest 16hrs) • Nursing may occur with the dam standing or lying on her side • Calves should be gaining about 4.5kg/day during the first 10days • In the first 2mths, calves usually suckle every hour. • After 2mths frequency drops to about every 2.5hrs • Weaning occurs naturally at about 2hrs. Weaning can commence at 6mths however it is recommended that it not be done until 1yr. • First defecation occurs 2-10days post birth • Passing of Placenta 1-2 days post birth
2. Have problems been seen at CZ or other institutions?	• Mothers have been seen to push babies around with horn, looks harsh but is normal • Port Lympne have found that several calves walk on their heels, usually lasts for a few days, has been longer in one baby but no interference was needed • One calf was lost as pushed into water bowl • One calf was hand-reared due to being distressed and weak, mothers milk had dried up, wasn't obvious from her mammary glands that this was the case, but we now have better facilities to observe this. • Kitani last July had problems with breech birth, Kitani had stopped pushing, left her for 24hrs. Unfortunately she must have stopped pushing as the calf would have already died
3. What problems might occur and what actions could be taken.	Possibilities include: • Aggression from mother – action remove mother and check calf. Try and reintroduce +/- sedation of dam. • Lack of interest from mother. – action remove mother and check calf. Treat calf (warm up, feed as necessary) If calf ok try and reintroduce and see if dam is now more attentive. • Calf trying to suckle but doesn't seem to be getting sufficient milk – This would become apparent if calf is distressed/restless and/or calf appears weak and/or calf is not gaining weight. Action consider supplementary or hand feeding. (see protocol below)
4. Hand rearing or supplementary feeding protocols	Indications for supplementary feeding: • Weak calf – needs a boost to get going – ie if week from birth and not standing after 4 hours • Not fed in first 12hrs – requirement to get some collostrum into it to ensure not immunosuppressed. – possibly put calf back with mum

	Indications for hand rearing: • Mum not sufficient milk – will keep a close watch on her mammary glands • Mum aggressive or not letting it suck Hand rearing protocol: • What bottles and teats/ are these ready? – One 2 litre bottle and one 3 litre bottle • What sterilisation/cleaning regime? – Milton and brush ordered, will be sterilised in large bowl • What milk formula? – Have formula in AHC from preparation for Kitani • For the first 24 hours, colostrum is fed – rhino is best, but either cow or horse can be used if required [both are in stock]. • For the 2nd 24 hours, the transition from colostrum to formula should be 50:50 [SanDiegoWAP] • Thereafter, milk formula is used – either rhino milk replacer or cow's milk (+iron/lactose) • Amount of milk given is determined by the calf's body wt. Start feeding a total of 10% of body wt. daily, increase to ~15-20% after first couple of weeks. This volume is divided by the number of feeds in a 24hr period. • For ease of use, I recommend we use Mazuri Rhino milk replacer* which has been formulated to mimic the composition of rhino milk, as described above. It already contains lactose and a vit/min mix so no further supplementation is required. • All calculations that follow use largest weight of black rhino calf at birth (40kg) and fastest growth rate (1 kg/day) [SanDiegoWAP]. NOTE – this is different from the birthing notes which state 4.5kg/day over first 10 days, which would double the calf's body wt in this time period. San Diego record this weight change over the first month, based on 4 animals; 2 white and 2 black rhino calves. • Mazuri Rhino milk replacer is a powder that needs to be mixed 1:10 with water. It comes in 10kg sacks and for guidance only I estimate 1 sack is sufficient for 21 days. [Calculations > 40kg calf – 10% of wt. as daily feed = 4 litres in total, spread over 7 feeds. Therefore each feed = 550 ml, can adjust for 'hungry' calf.] • Feed volume is adjusted to ensure adequate growth wt after this period, which will likely mean bags are used faster, but this can be assessed and calculated if required. • What frequency of feeding? – Every 2 hours to begin, including a late evening feed for first week (on a rota) then if strong enough 5pm feed and first thing in morning feed • What volume of feeding? - 1 litre every 2 hours to begin

	• How do you know if doing ok? (eg plot daily weight and compare) – will weigh daily • Does calf have to be removed from mum? (permanent, temp, during feeding only, potential for reintegration, etc) – yes we will remove calf from Mum permanently if hand rearing. Only put back in if it is just to give calf a boost • If calf removed what facilities required (location, bedding, extra heat, company etc???) – Small calving pen in old rhino house, hay as bedding to avoid straw impaction, will arrange extra heat if required, no company for first 3 weeks then will be discussed
5. Weaning and reintegration with other rhinos	Probably wean about 6-9 months depending on calf. Only would be integrated with another Calf so depends on ages of rhinos at the time

After the birth – managing information and the public

1. Who do we tell, when and how	• There is no reason to keep the birth a secret however in the first days information should be managed carefully so as to reduce possibility of disturbance to the mother and baby. • Staff can be told mother and calf have bonded well and the calf is suckling well. (Probably at least 48hrs). • PR will sort out press statements and how the story will be disseminated. If the birth goes well and mother and calf are well bonded a very limited photo shoot or filming may be allowed. This will be decided following the birth in conjunction with the rhino team.

Summary of the Birth of Asani 29/10/2008

- 1200 Kitani walking forwards and reversing
- 1545 Kitani gave birth
- 1745 Asani standing (2 hours post birth)
- 2210 Asani suckling (6 hours 25 mins post birth)

Summary of the Birth of Bashira 15/05/2009

- Unfortunately there was no data collected as calf was born overnight and cameras stopped working in the pen she gave birth in