AN ABSTRACT OF THE THESIS OF

Carol A. Allen for the degree of Master of Science in Animal Science presented on October 1, 1999. Title: Preliminary Investigation of Artificial Incubation of Emu Eggs, and Alternative Feeds and Management Techniques for Emu Chicks up to Ten Weeks of Age.

Redacted for Privacy

Abstract approved: _____
James C. Hermes

A preliminary study was conducted to establish baseline data for emu artificial egg incubation and emu chick growth and management. Three experiments examined factors affecting hatchability of emu eggs that occur prior to and during artificial incubation. Characteristics analyzed were egg storage duration, hen age, egg weight, egg index, shell thickness, incubation temperature, incubator type, and egg weight loss. The six treatments among the three incubation experiments consisted of two AVN incubators with temperatures maintained at 36.0 C and 35.8 C, and four Jamesway 252 incubators maintained at 36.0 C, 36.6 C, 36.5 C, and 36.7 C. Three subsequent experiments involved six treatments with hatched chicks to test the effects of diet and pen size on chick weight gain, and growth of the beak, middle toe, and tarsometatarsus up to eight and ten weeks of age. Sand, grass clippings, and pine shavings substrates were also tested.

Analysis of duration of egg storage indicated that an increase in pre-incubation storage time was associated with a decrease in hatchability. The highest percent hatch

(63.8%) occurred for eggs held ≤7 days and decreased for each additional week stored. Analysis revealed evidence that as emu hen age increased, fertility increased ($p<0.05$). Also, as hen age increased, egg size increased ($p<0.05$). Age of hen did not influence ($p>0.05$) egg index or shell thickness. There was no indication that egg index had an effect on hatch. An inverse relationship was observed between hatchability and egg shell thickness. Higher incubation temperatures were associated with a decrease in hatchability. Temperatures ≤36.0 C resulted in an average hatchability of 64.3%, while temperatures ≥36.0 C resulted in only 47.2%. Incubation temperatures ≤36.0 C were observed to increase chick quality compared to temperatures >36.0 C. Eggs having weight loss of 11%-14.9% during incubation had higher percentage hatch than eggs with losses out of this range. The pattern for embryonic mortality was observed to be similar to the pattern in other domestic avian species; high mortality peaks at the beginning and end of incubation. However, a higher than expected mortality occurred between the two peaks.

Chicks fed a 21.4% protein broiler starter diet (BS) had higher weight gain ($p<0.05$) than chicks fed a 20.1% protein chick starter diet (CS). Feed conversion among chicks consuming the BS was significantly better ($p<0.05$) than the chicks consuming the CS. In a 2 X 3 factorial design experiment, chicks were raised in two different pen sizes, 0.9 m X 3.0 m and 1.8 m X 11.0 m, and fed three different commercial diets; an 18% protein all-in-one diet (AIO), a 23% protein emu diet (EMU), and a 28% protein turkey-gamebird starter diet (TG). Chicks showed no evidence ($p>0.05$) of interaction between pen size and diet for weight gain, feed conversion, or growth of the beak, middle toe, and tarsometatarsus. However, chicks fed EMU

consumed significantly more feed ($p<0.05$) than chicks fed TG. Pine shavings substrate was determined to be the least labor intensive and provided the best footing for the chicks among the three substrates tested.

According to this investigation, for farmers to achieve maximum production for artificial incubation of emu eggs, they should use breeder hens ≥4 years old, store eggs no longer than 7 days, and incubate eggs at temperatures ranging from 35.5 C-36.0 C. Until nutrition requirements are established for emu, there is no advantage to feeding emu chicks through 10 weeks of age a commercial emu diet compared to other commercial domestic bird diets.

Preliminary Investigation of Artificial Incubation of Emu Eggs, and Alternative Feeds and Management Techniques for Emu Chicks up to Ten Weeks of Age.

By

Carol A. Allen

A THESIS

submitted to

Oregon State University

in partial fulfillment of
the requirements for the
degree of

Master of Science

Presented October 1, 1999
Commencement June 2000

Master of Science thesis of Carol A. Allen presented on October 1, 1999

APPROVED:

Redacted for Privacy

Major Professor, representing Animal Science

Redacted for Privacy

Chair of the Department of Animal Sciences

Redacted for Privacy

Dean of the Graduate School

I understand that my thesis will become part of the permanent collection of Oregon State University libraries. My signature below authorizes release of my thesis to any reader upon request.

Redacted for Privacy

Carol A. Allen, Author

ACKNOWLEDGMENTS

Many generous people assisted with this research. I am particularly grateful to Dr. James Hermes for giving me the unique opportunity to study the emu, and for his expertise in incubation and management of production birds.

I wish to especially thank the members of the Oregon and Washington Emu Associations, individual emu farmers, Kropt Feed Inc., Grange COOP Supply Association, and Ratek Industry for their generous donations which made this research possible.

I am grateful to all those who expressed confidence in my ability to complete this graduate program by awarding me the Hansen Scholarship, OSU Folk Club Scholarships, and the Chester Wilcox Graduate Fellowship.

A special thanks to Dr. Harold Koenig for serving as my minor professor in business administration and his helpful comments in review of this material. I also wish to thank Dr. James Males and Dr. Neil Christensen for their willingness to serve as members of my committee and review this report.

Thanks are extended to Dr. David Thomas, Dr. Diane Carroll, Shaun Wulff, and Haribaskar Srinivasan for their assistance with statistical analysis.

I wish to express graditude and appreication to following individuals for their assistance and support. Kari Henschel, Larry Mirosh, Rosemary Schulte, Allen Feltman, Robert Williams, William Ross, Cliff Thompson, Dale Miller, Stephanie Greenway, Lonnie Quinlin Murphy, Richard Christian, Tina Clark, David Cuningham, Jason Gentemen, Richard Glenn, Derek McLean, and Sarah Allen.

Finally, I wish to express my sincere thanks to Julie Reeder and Alejandro Chavez for their continuous assistance and support, as well as good friendship throughout this research.

TABLE OF CONTENTS

TABLE OF CONTENTS (Continued)

TABLE OF CONTENTS (Continued)

LIST OF TABLES

DEDICATION

I dedicate this thesis to my son Corey, whose encouragement, great sense of humor, and love were gratefully appreciated throughout this study. Also to my dad, Nevin, who finds it difficult to understand why I do the things I do. Finally to the memory of my mum, Helen, who I'm sure knew.

Preliminary Investigation of Artificial Incubation of Emu Eggs, and Alternative Feeds and Management Techniques for Emu Chicks up to Ten Weeks of Age.

INTRODUCTION

Evolution

Emus (Casuariiformes, Dromiceidae, Australia & Tasmania), along with ostriches, (Struthioniformes, Struthionidae, Africa), cassowaries, (Casuariiformes, Casuariidae, Australia & New Guinea), kiwis (Apterygiformes, Apterygidae, New Zealand), and rheas (Rheiformes, Rheidae, South America), belong to a group of flightless birds identified as ratites. Similar morphological characteristics among the ratites are a palaeognathous palate (a distinct reptilian like configuration of the palatal bones) (Huxley, 1867; McDowell, 1948), and a keel-less sternum. Merrem in 1813 used the keel-less sternum to first classify ratites separate from all other living birds (McDowell, 1948).

Other similar features, found also in some other flightless birds and seen in early developmental stages of carinate birds (all living birds except ratites) include feathers without hooklets on the barbules, reduced or rudimentary wings, an obtuse angle of articulation between or fusing of the scapula and coracoid bones (Huxley, 1867), a reduced furcula (wishbone) (Jenkins *et al.*, 1988), an ilioischiatic fenestra (broad open area between the ilium and the ischium) (Cracraft, 1974), and, in some ratites, a sutured skull (deBeer, 1956; Dawson *et al.*, 1994). Additional features are solid metatarsus and

tibiotarsus bones (Grzimek, 1973) and the absence of the pretibial bone (McGowan, 1984).

Tinamous (Tinamiformes) are the only other bird group having a palaeognathous palate, however they are not included within the ratite group because they have a keel on the sternum. The presence of the palaeognathous palate along with the keel-less sternum separates the ratites from other bird groups including other flightless birds (Huxley, 1867).

For over a century there has been controversy whether the ratites evolved from flying or non-flying ancestors, and whether they are of a monophyletic or polyphyletic origin. Today the majority of investigators are in agreement that ratites derived from flying ancestors. Evidence used to arrive at this conclusion include the presence of structures adapted for flight: degenerate flight quills on the wings of the cassowary; a pygostyle (fused tail vertebrae); formation of a carpometacarpus in the wing skeleton; a similar type cerebellum (controlling equilibrium) to that found in flying birds; and in rheas, the presence of an alula on the wing (Feduccia, 1996).

Huxley's discovery of the palaeognathous palate was the beginning of the controversy of the monophyletic or polyphyletic origin of ratites and it persisted up to the geologic discovery of the drift in continents in 1963. Evidence of the break up of Gondwana provided strong support for studies proposing a monophyletic origin (Cracraft, 1974; Prager *et al.*, 1976). Thus, the debate was thought to have been resolved. However, two more recent reports by Houde (1986) and Dawson *et al.* (1994)

reopened the controversy over the ratite groups evolving to present day by convergence or divergence.

Peter Houde (1986) identified fossils from North America and Europe as being the ancestors of ostriches. Houde hypothesized that ostriches emigrated from the northern hemisphere to the southern hemisphere, the opposite route for the dispersal of ratites designated by continental drift. The absence of fossils of ostriches in the southern hemisphere, considered conspicuous by Houde, indicates a convergence of the ratites.

The study by Dawson *et al.* (1994) supported the theories of deBeer (1956), and McDowell (1948) that the ratites are not flightless as a result of evolution, but instead are a result of neoteny (arrested development). They thyroidectomized starlings at 4 days of age and discovered neotenous characteristics resulted in the adult. Plumage pattern, feather structure, juvenile type palate, and the presence of skull sutures rather than fused in adult thyroidectomized starlings were all similar characteristics to those identified in adult ratites. Thus indicating the possibility that ratites may not be primitive, but instead are probably recent, and independently developed neotenous characteristics from separate origins and converged by adaption.

It has been shown in the evolution of flightlessness among rails separated in geographic isolation that "when flightlessness occurs, the same general morphological features appear time and time again, convergently" (Feduccia, 1996). A thorough review of the studies investigating the phylogeny of the ratites has been presented by Sibley and Ahlquist (1990) and Feduccia (1996). The reader can refer to these references for a more detailed discussion.

Until the debate over monophyletic or polyphyletic origin is resolved, the question of the relationship among the ratite groups will go unanswered. Most investigators have concluded the emu and cassowary are of the same genera with the kiwi as their closest living relative and the ostrich along with the rhea being more distant (Sibley and Ahlquist 1990). Cho, *et al.* (1984) and Huxley (1867) reported several anatomical differences in features among the ostrich, emu, cassowary, and rhea. These differences included ostriches having only two toes, while the others have three, and the wings of the emu and cassowary are rudimentary in comparison to the ostrich and rhea. Also, the number of digits on the wings differ as do the number of clawed digits. The digestive tract differs among the ratites in structure of the tongue, proventriculus, gizzard, duodenal loop, and the length of the ceca and colon (Cho *et al.*, 1984; Herd, 1985; Fowler, 1991). Other features varying in appearance include the spleen, thymus, trachea, bursa of Fabricius, and color of the gonads (Cho *et al.*, 1984). Oliver (1945), Cracraft (1974), and Duke *et al.* (1995) compared the leg bones of the ratites and found differences in the joint structure of the femur, tibiotarsus, and tarsometatarsus. Dawson *et al.* (1985) and Withers (1983) noted different methods of water conservation in the ostrich and emu.

Natural History

The emu, *Dromaius novaehollandiae*, is the second largest living bird, dwarfed only by the ostrich, *Struthio camelus*. The emu is native to Australia and found in all

regions of the continent except in the rain forests, the driest deserts, and in areas of dense human population (Campbell and Lack, 1985). These birds are highly mobile with large ranges, wandering or migrating over great distances following the occurrence of food supply brought on by intermittent rains. The emu diet is diverse and rich in nutrients consisting of insects, caterpillars, fruits, flowers, berries, seeds, and new growth of herbage, grasses, and shrubs (Davies, 1978). Chicks feed mainly on insects for the first few weeks. Coprophagy has been reported where emus pick semi-digested food particles from fresh emu droppings. Chicks may benefit from this behavior when available food sources are too high above the ground for the chicks to reach (Davies, 1978; Campbell and Lack, 1985).

When the Europeans first arrived in the region, the subspecies *Dromaius novaehollandiae diemenensis* occurred on Tasmania and two smaller emu species, *Dromaius ater* and *Dromaius baudinianus*, were found on King Island and Kangaroo Island, respectively, located off the southern Australian coast (Garnett, 1992). Emus and their eggs were hunted as a food source by the settlers and sailors, and the oil taken from the carcass was used in lamps (Hill, 1967; Grzimek, 1973). After the islands and the mainland were settled and agriculture established, emus destroyed crops and surrounding fencing. They freely utilized water troughs and pasture grasses intended for domestic livestock. Farmers retaliated by killing the emu in massive numbers and destroying eggs found in nests (Burton and Burton, 1969). By 1865, the emu were extinct on King Island, Kangaroo Island, and Tasmania (Blakers, *et al.*, 1984; Garnett, 1992). As late as 1964 in western agricultural regions of Australia, the government still paid a bounty for

heads and eggs of emus (Hill, 1967; Grzimek, 1973). However, *Dromaius novaehollandiae* has managed to maintain a strong presence throughout the mainland with the help of reintroduction programs in eastern regions (Blakers *et al.*, 1984) and laws established to protect this national bird of Australia. In western semi-arid regions, emu populations have benefited from construction of dams and placement of livestock water troughs providing a permanent source of water (Campbell and Lack, 1985). Rather than killing the birds, an alternative strategy in western agricultural regions has been to put up hundreds of kilometers of fencing to keep emu out of crop and livestock pastures, away from water troughs (Grzimek, 1973).

Agricultural History

Past History

The first commercial farming of ratites occurred in 1838 in South Africa where ostriches were raised for feather production. Due to the popularity of the feathers and the high prices paid for them, farms soon followed in Algeria, Sicily, France, and the United States. Before World War I, farmers in South Africa could expect to receive up to $7500

for the sale of a proven breeder male. However, following the war, feathers were no longer stylish forcing most of the farms to go out of business. Only a few farms remained viable in South Africa (Grzimek, 1973).

Recent History

In the early 1970's, ostrich farms began to increase in number in South Africa, this time for the production of leather (Grzimek, 1973). An interest in ratite farming emerged in the late 1970's in the United States raising ostriches for meat, leather, egg, and feather production. The emu and the rhea were found on farms by the mid 1980's. The emu was also being raised for production of oil rendered from the fat. By 1988, the number of farms raising emus had increased enough to support the formation of the American Emu Association (AEA) in Breham, Texas with 50 charter members. The 1996 membership in the AEA spanned 43 states with an estimated 2650 members.

Most emu producers raise the birds as alternative agriculture, many on diversified family operations. From a 1994 national survey of members of the AEA, it was estimated that there were approximately 450,000 emus (all ages) being raised domestically in the U.S. (Ford, 1994). Commercial farming of emus in Australia was not approved by the Australian government until 1987. By 1995, a reported 84,000 emus were being kept on 1300 farms (O'Malley, 1995).

The only emus in the U.S. before the interest in farming emus were those kept in zoos or in small private exotic bird collections. The Wildlife Protection (Regulation of Exports and Imports) Act 1982 forbidding the export of avian species from Australia along with laws protecting emus within Australia left only a small genetic pool to generate an emu production industry in both the U.S. and Australia.

During the early production of ratites on farms in the U.S., what little was known about the production of ostriches was applied to the raising of emu. However, from the results of phylogeny investigations, and studies of anatomy and physiology referenced previously, it is clear that information concerning the production needs of the ostrich are not necessarily relevant to the needs of emu. Production requirements of ratites have been based mostly on trial and error of the farmers and little on controlled scientific studies. Most of the studies available on ratites pertain to ostrich, while studies of the emu as a farm production animal are limited.

The ratite industry functioned as a breeder market through the 1980's and into the early 1990's. During this time producers realized huge profits for the sale of their birds not unlike the high prices received prior to World War I. Because of the immense profits, farmers had little concern for production costs. The priority was set on the sale of the birds, to the extent of incurring high veterinarian charges to insure the sale of even one bird. In the past few years, the market has turned from a breeder market to a slaughter market and with it, a drastic drop in profits to the producer. Many of the entrepreneur farmers sold out as this changeover took place. The remaining growers are determined to make the industry a viable one. Other people considered the drop in market profits as an opportune time to enter into the industry, due to lower capital investment. However, current producers have become aware of the relatively high costs of production which was economically obscured by previous profits of a breeder market.

Justification of Research

Artificial Incubation

The parameters for artificial incubation of emu eggs have not been clearly defined. Zoos and producers have been the main source for emu egg incubation requirements due to the lack of scientific studies elsewhere. In Ford's 1994 AEA survey of members raising emus, the reported hatch rate of artificially incubated emu eggs averaged 48.65%. This figure indicates a high production cost for farmers in terms of lost product, cost of producing chicks, incubator space, and labor. The sale of an egg shell from an infertile or a non-developing egg does not cover the cost of producing that egg and labor costs from time it is laid to after it is determined to be a non-hatcher.

Some farmers report a higher percentage hatch based on the number of fertile eggs incubated rather than total eggs set. They admit they do not break out every unhatched egg to determine the number of true fertile eggs. It is virtually impossible to distinguish fertility if the eggs are not broken out for careful examination. Low percentage hatch rates reported by individual farmers and in the AEA survey strongly indicates an industry incubation problem and a need for research of emu egg artificial incubation.

Nutrition

One major production expense has been and still is the relatively high cost of feed. The first feeds commercially available to ratite farmers were generic "ratite" diets. After the differences in the digestive tracts among the ratite birds were acknowledged, separate specific diets for ostrich and emu became available, but still at a relatively high price. The ratite feeds have been formulated without the nutritional requirements of the birds having been determined. Diets have been based on the National Research Council requirements for poultry with varying increased levels of protein and some vitamins and minerals. These additives cause the feeds for ratites to be higher in cost than commercially available poultry feed.

Management

There are insufficient studies on other management practices for emu production. Assumptions have been made that ratites need large areas, due to their large size, for proper growth and development. The spatial requirements of ratites have not been established. The common practice that indoor-outdoor pens are required for young chicks and the remodeling of existing buildings or building of new facilities for indoor-outdoor pens has not been substantiated as a requirement. Large pens also increase investment in fencing materials and labor for construction. Indoor surfaces of several types have been tried by producers to provide secure footing for preventing leg problems in growing chicks. These have included such costly materials as indoor-outdoor

carpeting, grooved rubber matting, and extra fine masonite sand. Less expensive substrates, such as those used in broiler operations, have not been tested due to the assumption of impaction.

The highest mortality of live birds for an emu enterprise typically occurs within the first three months after hatch reaching losses of approximately 20%. Losses decrease substantially to about 4% from three to six months of age. After six months there is rarely any mortality (Ford, 1994) other than injuries resulting from aggressive breeding behavior.

Purpose

Due to the low hatchability of emu eggs, the high losses of young chicks, and the high cost of emu feed this study focused on incubation characteristics, embryo development, chick growth up to ten weeks of age, alternative diets, and management strategies. The main purpose of this study was to use controlled research to establish preliminary feeding and management guidelines for production of young emus.

LITERATURE REVIEW

Artificial Incubation Introduction

Artificial incubation of avian eggs, although appearing to be a simple process, is in fact quite complex. Many factors occurring before and during artificial incubation can influence the success of a healthy chick emerging from the egg at the end of the incubation period. The complexity is increased because several of these factors are interrelated (Lundy, 1969).

Researchers agree that three of the most important requirements for successful hatch of artificially incubated eggs are proper temperature, relative humidity, and the turning of the eggs during incubation (Landauer, 1961; Lundy, 1969; Tullett, 1990). These factors may also be important during the pre-incubation storage period, from the time the egg is laid until incubation begins (Landauer, 1961).

There is a voluminous amount of literature available on the factors affecting the hatch of artificially incubated eggs of domestic avian species. The result of these investigations have greatly contributed to the success of today's poultry industry. Extensive reviews of research concerning the factors affecting the outcome of artificial incubation of poultry eggs have been compiled by Landauer (1961 and 1973) and Lundy (1969).

Investigations on artificial incubation of ratite eggs are quite limited with the majority involving ostrich egg incubation. Published research for artificial incubation of

emu and rhea eggs is notably lacking. The few scientific studies on emu incubation available are supplemented by several publications (Minnaar and Minnaar, 1992; Parsons, 1994; Stewart, 1992, 1994, and 1996; Wade, 1992) based on personal observation rather than controlled research. These publications offer hatchery management recommendations for the artificial incubation of emu eggs.

The following review includes some of the individual and interrelating factors influencing the hatchability of artificially incubated avian eggs that may be relevant to the present study. Hatchability is defined as the percentage of eggs that hatch from the total number of eggs incubated (Landauer, 1961).

Pre-incubation Storage

Duration

Awareness that eggs could be stored up to ten days after the day the eggs were laid prior to incubating had been documented by the Romans in the first century A.D. (Landauer, 1961). More recently an actual benefit to holding eggs for a short period prior to placing (setting) the eggs into the incubator has been reported. Asmundson and MacIlraith (1948) concluded that turkey eggs held for several days prior to setting had better hatchability than eggs set the same day as laid. Similar results were reported for chicken eggs by Funk *et al.* (1950) and by Kosin (1954) for turkeys. However, both of the later studies found a progressive decline in hatchability for eggs held longer than seven days. A study on Japanese quail showed an initial drop in hatchability after only

one day followed by a slight continuous decrease through day 16 after which a significant drop in hatchability occurred (Chahil and Johnson, 1974).

Funk (1934) found eggs stored longer periods required a longer incubation period, therefore delaying the time of hatch. Chahil and Johnson (1974) confirmed this when they calculated that for each day of preincubation storage, the hatch of Japanese quail eggs is delayed by 1.1 hours. They suggested the duration of storage may significantly widen the interval of hatch time for a group of eggs.

Temperature

During preincubation egg storage the embryo should remain in a dormant state. Proper temperatures to maintain dormancy of stored eggs must be above freezing and below the physiological zero temperature for the species incubated. Physiological zero is the temperature below which no embryonic development occurs (Landauer, 1961). Funk and Biellier (1944) determined the physiological zero temperature for the chicken to be 27°C. Eggs stored above this temperature, but below normal incubation temperature, had abnormally slow development and a negative effect on hatchability. Olsen and Haynes (1948) determined the optimum storage temperature range for chicken eggs to be between 10.0°C and 12.8°C with temperatures closer to 10.0°C maintaining the high hatch quality in eggs held for longer duration. In a later report, Proudfoot (1969a) reported an advantage to using higher storage temperatures of 15°C - 16°C

when storing eggs seven days or less, and lower temperatures, 11°C - 12°C, for longer storage.

Humidity and Egg Weight (Water) Loss

Relative humidity greatly affects egg weight loss during storage (Proudfoot, 1969a). High relative humidity levels of 80-88% were reported to be optimum for chicken eggs during storage to maintain hatch quality (Funk and Forward, 1951). Proudfoot (1964) testing a wider range of relative humidities from 75%-90% found no significant variance in hatchability, however he did note humidity levels at the lower end of this range resulted in an improved hatch of chicken eggs stored 3 weeks.

A recent study by Fasenko *et al.* (1992b) indicated eggs stored for a period of ≥14 days versus eggs stored for ≤7 days resulted in lower embryo viability (liveability during the storage period), along with higher egg weight loss at the end of the storage period for eggs stored for the longer duration. In eggs broken open for examination prior to incubation, embryo viability was reported to be significantly related to the increase in weight loss, but a decrease in embryo viability was not significantly affected by the length of storage. However, the duration of storage had a significant linear relationship with the increased weight loss. From these results, they proposed an indirect effect of duration of storage on embryo mortality due to the highest mortality during storage occurring in eggs stored for a longer time.

Position and Turning

The storage position of eggs and the turning or not turning eggs during storage prior to incubation are two additional factors investigators have determined to affect hatchability. The conventional position for hatching egg storage has been in an upright position with the large (air cell) end up to maintain proper orientation of the blastoderm and yolk. Funk (1934) claimed turning chicken eggs while stored in the conventional position showed no benefit to hatchability. However, Funk and Forward (1951) suggested there may be a slight benefit to turning eggs if storage was greater than one week. When the eggs were stored with large end up longer than 2 weeks, Proudfoot (1966) confirmed it was beneficial to turn the eggs. Proudfoot (1969b) further presented evidence that hatchability improves when eggs are stored small end up and not turned, even for periods extending 2 to 4 weeks. In the same study he showed evidence of a similar hatchability when eggs were stored horizontally, but turning was necessary. A study with turkeys resulted in eggs stored with small end up for 3 days having a reduced hatchability while eggs stored longer than 7 days showed improved hatchability. Turning or not turning in this study resulted in no differences in hatchability (Stephenson, 1985).

Scientific investigations to determine if there is any benefit of improved hatchability to storing emu eggs before placing them into the incubator were absent among the available literature. The physiological zero for emu embryos as well as optimum storage conditions (temperatures, humidity levels, turning, position, and duration) for emu eggs have not been documented in research literature. Minnaar and

Minnaar (1992) have surmised emu eggs may be held in storage up to 3 days without negatively affecting hatchability. Literature is lacking as to the benefit of storing emu eggs prior to artificial incubation as reported for other species by Admundson and MacIlraith (1948), Funk *et al.* (1950), Kosin (1954), and Chahil and Johnson (1974). According to Shane (1996) emu eggs may be stored as long as 6 days before a decline in hatchability is evident. If emu eggs are stored prior to incubation, Minnaar and Minnaar (1992) recommend storing emu eggs at 4.5°C - 15.5°C (13.0°C optimum) in a horizontal position and turning two times per day. They propose a physiological zero temperature for emu embryos to be 21.1°C, however there are no controlled studies to substantiate these statements at the present time.

Minnaar and Minnaar (1992) stated the high humidities required for preincubation of domestic fowl were not necessary during storage of emu eggs due to the thicker shell of the emu egg and therefore negligible water loss during storage. This lack of weight loss during storage may be due to an additional unique layer covering the cuticle of the emu egg shell, however the function of this layer and its possible affect on water loss are unknown (Freeman and Vince, 1974). In regard to emu egg storage conditions, Shane's (1996) opinion again differed from Minnaar and Minnaar (1992). He proposed the recommendations of Stewart (1996 and 1992) for storage of ostrich eggs are applicable to storage of emu eggs with the exception of storage duration. Stewart suggested a temperature range of 12.8°C - 18.3°C and a high relative humidity of 75% with the eggs positioned vertically, air cell end up. Also, the eggs do not need to

be turned unless stored longer than 7 days. Again there were no research studies found to support these recommendations.

Incubation

Temperature

The optimum temperature range in forced air incubators to artificially incubate chicken eggs is 37.0°C to 38.0°C for best hatchability. As temperatures successively rise and fall from this range, hatchability decreases linearly arriving at 0% for temperatures of 35.0°C and 40.5°C (Romanoff, 1936). The temperature of the egg increases due to an increase in metabolic rate of the embryo by the last quarter of incubation (Romijn and Lokhorst, 1955). Meir and Ar (1990) found evidence of this for ostrich eggs when they measured a temperature difference of 2.0°C between the air temperature of the incubator and the surface temperature of the egg. These studies support Romanoff's (1936) recommendation for lowering the incubator temperature at the beginning of the last quarter of incubation as a safeguard to prevent possible heat injury to the embryo. This practice increases the likelihood of greater chick hatch weight to initial egg weight ratio. Romanoff concluded that temperatures resulting in maximum hatchability have the highest ratios, therefore these ratios may represent a measurement of incubation efficiency. According to Minnaar and Minnaar (1992) there is an optimum chick hatch weight to initial egg weight ratio of 0.67 for emu chicks. They relate this ratio to egg

weight loss during incubation, lower ratios having excess water loss and higher ratios indicating insufficient water loss.

Evidence has been presented by Romanoff (1936) that duration and quality of the hatch can be influenced by temperature. Lower temperatures delay hatch time and produce higher quality chicks, whereas higher temperatures shorten the hatch time, but at the expense of decreased hatchability. Temperatures above 39.5°C however, were noted to seemingly delay the hatch of chicken eggs. More recent evidence of chick quality related to incubation temperature has been presented by French (1992) with turkeys. Using three incubation temperatures, 36.5°C, 37.5°C, and 38.5°C, he found that poult weights decreased as temperatures increased and down color was observed as being paler in color, an indication of poor quality. Hatchability was lowest for eggs incubated at 38.5°C with high embryo mortality during the last half of incubation. Mortalities at this higher temperature had a high occurrence of hemorrhages and abnormalities. Unfortunately French did not investigate the effect of incubation temperature on hatch time.

Stewart (1992) recommended, based on personal observation, an incubation temperature range for emu eggs of 36.0°C-36.7°C. The average body temperatures of two male emus incubating eggs were measured by Butterman and Dawson (1989) to be 37.7°C and 37.9°C with little variation over the course of incubation. They reported both temperatures successful in hatching chicks. However, Buttemer *et al.* (1988) reported increasing temperatures in naturally incubated emu eggs. They documented gradually increasing temperatures from set to day 10 from 32.0°C to 34.0°C.

Temperatures remained constant until day 15, then gradually increased from 34.0°C to 36.0°C up to day 35 and remained constant at 36.0°C from day 35 through to hatch.

Humidity and Egg Weight Loss

The relationship between relative humidity in the incubator and the percent egg weight loss during incubation involves the interaction of many factors. Relative humidity and temperature have an inverse relationship, as temperature increases, the optimum relative humidity decreases and vice versa (Lundy, 1969). Relative humidity in the incubator along with egg shell porosity influence egg weight loss during incubation and subsequent hatchability (Ar *et al.*, 1974; Lundy, 1969). Egg shell porosity increases during incubation when there is an increase in incubation temperature along with a decrease in relative humidity. Higher hatchability is evident in eggs with low porosity compared to eggs having high porosity (Lundy, 1969). Lundy reported ideal water loss to obtain highest hatchability ranges from 10%-12% of the initial egg weight and that relative humidity levels from 40%-70% are best for reaching weight losses in this range.

During incubation, metabolic water is generated from increasing metabolic activity of the embryo, thus increasing the water content of the egg (Rahn *et al.*, 1979). The increase in metabolic water causes an increase of water vapor pressure inside the egg (Meir and Ar, 1990; Rahn *et al.*, 1979) The additional amount of relative water must be lost for successful hatching to occur. This is done during incubation by diffusion of water through microscopic pores in the shell. The total amount of water transferred out

of the shell is determined by the water vapor conductance (porosity) and the relative humidity in the incubator (Rahn *et al.*, 1979; Rahn and Ar, 1974; Booth and Rahn, 1990; Tullett 1990).

Rahn *et al.* (1979), determined that there was a consistency in water loss totaling approximately 15% of the initial egg weight regardless of incubation length or initial egg mass. This conclusion indicated that the rate of water loss remains uniform during incubation for all species until external pipping occurs. However, more recent studies by Davis *et al.* (1988) and Hulet *et al.* (1987) have disagreed with Rahn *et al.* (1979) reporting an increase in the rate of water loss during incubation and percentages of weight loss similar to the 10%-12% weight loss reported by Lundy (1969).

Davis *et al.* (1988) reported highest hatchability for chicken eggs occurred when weight loss was 12%, while Hulet *et al.* (1987) found hatchability best for turkeys when egg weight loss was 11.5%. Reports of optimum water loss rates for ratites were also less than that predicted by Rahn *et al.* (1979): ostrich eggs averaged from 11-13.5% loss (Bertram and Burger 1981; Jarvis *et al.* 1985; Brown *et al.* 1996), a 10.0% loss for both emu eggs (Buttemer *et al.*, 1988) and rhea eggs (Cannon *et al.*, 1986). Ar *et al.* (1994) received best hatch for ostrich with weight loss between 12%-15%, having highest at a 13% loss.

Booth and Rahn (1990) studied the rate of water loss for chicken, turkey, and quail eggs. They found an increase in the rate of water loss during incubation beginning on day 10, 15, and 11 of incubation, respectively. They attributed the increase in rate of loss as a result of increased metabolic activity of the embryo, and subsequent increased

water vapor pressure inside the egg. Similar results have been reported by Buttemer *et al.* (1988) and Meir and Ar (1990) for emu and ostrich eggs respectively.

Buttemer *et al.* (1988) and Vleck *et al.* (1980) reported emu eggs to have very low water vapor conductance relative to their long incubation period and large egg mass. Vleck *et al.* suggested this may be an adaption to conserve water in an arid environment. However, Buttemer *et al.* proposed the low water vapor conductance may be to achieve a target optimum weight loss percentage comparable to the 11%-12% loss in chicken eggs. Stewart (1992) recommended a relativity humidity range of 25%-40% for incubation of emu eggs.

Hen Age

There are no studies on the affect of hen age on hatchability for emu. Due to the extreme differences in age of maturity, life span, and intensity of egg production it is difficult to make comparisons between emu hens and commercial poultry hens. However, some general principles may be relevant. Fasenko *et al.* (1992a) reported lower fertility and hatchability in older chicken hens (31-54 weeks of age (WOA)). Landauer (1961) compiled a comprehensive review of studies on the relationship of egg size to hatchability. There was general agreement among investigators that egg size increases with age of the hen. Large eggs were reported to have low hatching potential, while medium sized eggs have the highest hatchability.

The literature includes conflicting studies with regard to the hatchability of small ostrich eggs. In a recent study, Deeming (1995a) reported ostrich eggs had a significant difference in the size of hatched versus unhatched eggs with a greater number of large eggs not hatching. Both small and large ostrich eggs were found by Ar *et al.* (1994) to have a somewhat lower hatchability than medium sized eggs.

Tserveni-gousi and Yannakopoulos (1995) reported chicken hens reaching sexual maturity at a later age (140 days of age) laid larger eggs during the first laying period than hens that laid their first egg at an earlier age. As egg weight and age of hen increased, these investigators found that egg shape index (width/length) decreased. Earlier studies found egg shape had no effect on hatchability, except eggs with low egg index (extremely long, narrow eggs) seldom hatch (Landauer, 1961). Hoyt (1976) reported that shape is not related to surface-volume relationship and concluded the shape of an egg has insignificant or no effect on gas exchange compared to other incubation parameters.

Embryo Development and Mortality

As mentioned previously, temperature, humidity, and egg rotation are three important factors influencing successful artificial incubation. However, the eggs must have the potential for hatching to permit these factors to work efficiently. First the eggs must be fertile and free of infection. Infertility can be influenced by breeder behavior, health, age, and genetics (Landauer, 1961 and Lundy, 1969). Breeder nutrition and

subsequent nutrients in the egg must be sufficient for proper embryo development. Parent and embryo genetics can also influence embryo development.

Many nutrients are essential for the normal development of the avian embryo. The embryo will not survive if quantities present are insufficient or absent in the egg (Landauer, 1961). The causes of embryo mortalities resulting from nutritional deficiencies have been well reviewed and their effects on embryo development described by Beer (1969) and Landauer (1961 and 1973). In their reviews, Beer and Landauer concentrate on the indirect nutrient deficiencies that could cause hatchability problems, although there are both direct and indirect deficiencies. Direct deficiencies are resultant from insufficiency of one or more essential nutrients in the feed. Inaccurate formulation or mixing of feed, or improper storage resulting in deterioration could cause direct deficiencies. The indirect deficiencies are those arising from parasites and insufficient feed consumption (Beer, 1969), and maternal hereditary influence for utilization of nutrients (Landauer, 1961). As a result of the investigations reviewed by Beer and Landauer, many of the requirements for hen breeder diets were established for high hatchability in poultry. Beer noted that hatchability problems due to direct deficiencies in the diet are rare in commercial poultry since requirements have been established for breeder diets.

The nutrient requirements of emu hens have not been established (Angel, 1993). Also, low hatchability reported for emus (Ford, 1994) in relation to breeder diet or heredity has not been investigated. Subsequently it is not known if breeder nutrition (directly or indirectly) for emu may be a possible cause for poor hatch and/or chick

viability. Angel (1993) investigating nutrient levels of emu and ostrich eggs as indicators of hen diet found low levels of vitamin A and folic acid in emu eggs relative to levels found in ostrich and chicken eggs. Also protein levels were slightly lower in the emu eggs. Angel (1993) suggested the lower levels may be due to the reduced feed intake of emu hens during breeding, thus indicating a possible indirect deficiency. Beer (1969) and Landauer (1961) described deficiencies of vitamin A and folic acid as being detrimental to embryo viability. Raines (1994) stated that in her opinion vitamin deficiencies are less of a problem than vitamin overdose in the ratite industry.

There are two distinct embryo mortality peaks during incubation, one early within the first week for chickens, and a more pronounced late peak just prior to hatching (Landauer, 1961). Mortalities occurring between these two peaks usually signify a nutritional deficiency. High embryo mortality seen from day three to five of incubation in chickens may indicate severe nutritional deficiencies (Abbott, 1975). Landauer (1961 and 1973) submitted that heredity and environmental factors influencing hatchability stated earlier are probable causes for the early and late mortality peaks.

Minnaar and Minnaar (1992) witnessed the occurrence of dead embryos having clubbed down and parrot beak, characteristics of riboflavin and biotin deficiencies, respectively (Beer, 1969 and Landauer, 1961). Parrot beak can also be a sign of folic acid or nicotinic acid deficiency (Beer, 1969). Other mortalities observed by Minnaar and Minnaar and known to be caused by nutritional deficiencies in chickens include dwarfed embryos, which can occur from a deficiency of riboflavin, folic acid, or vitamin B_{12} (Beer, 1969). However, dwarfed embryo has also been identified as a genetic lethal

mutation in poultry by Asmundson (1945). A descriptive review of harmful genetic effects on hatchability in poultry has been presented by Landauer (1961 and 1973) and Crawford (1990). No scientific literature relating to genetic factors negatively affecting hatchability of emu eggs is available at this time.

Management and Growth

Spatial Conditions

Spatial requirements for raising meat type chickens have been specified by the Nutrition Council of American Feed Manufacturers Association. Birds are considered stressed at levels below the recommended allowances for population density and feeding space. To test the theory that stress is reduced by increasing feeding space when bird density is higher than recommended levels, Hansen and Becker (1960) analyzed the relationship between population density, feeding space, and growth in young chickens. Results showed a decrease in growth rate as population densities increased and no effect of feeding space on growth rate, however reduced growth would occur if levels approached the minimum level recommended. One trial of the study showed a strong indication that feather picking was aggravated by limited feeding space. These researchers found reduced growth from high density was not compensated by increasing feeding space. They explained that the lack of compensation was probably due to increased difficulty for a bird to obtain feed.

Controlled studies determining the spatial requirements for ratites are absent in the scientific literature. However, several publications are available in which the authors have made recommendations not based on scientific research pertaining to space requirements of ratites. For ratite chicks less than 9.0 kg, up to eight weeks of age, Raines (1994) asserted the minimum recommendation is 40 sq. ft. per chick and 225 sq. ft. per chick over 9.0 kg. If impaction (a build up of food in the crop or stomach causing an inability of food to move through the digestive tract) becomes a problem with management conditions remaining constant, Raines suggested inadequate space may be the problem. Wade (1992) stressed that adequate space permitting sufficient exercise as the primary requirement for ratite chicks up to 12 weeks of age. He recommended outside oblong pens providing 200-400 sq. ft. per chick plus an enclosed shelter having 3-15 sq. ft. per chick. Wade prescribed exercise to minimize the occurrence of impaction and leg rotation in chicks. He indicated that although ample exercise may reduce incidence of leg rotation, the cause is probably multifactorial. Parsons (1994) and Stewart (1994) also advocate ratite chicks to be raised in large outside pens providing plenty of room for exercise.

The previous three authors recommending large space requirements for ratite chicks are in agreement that restricted space increases the probability of leg problems in growing ratite chicks. Again, there were no controlled studies found in the literature to support these recommendations. Stewart (1994) also suggested that sand and straw provoke leg problems and therefore, advocates flooring with good traction such as brushed concrete, pine shavings, or natural dirt. Even though Raines (1994) appeared to

be in agreement with these authors that the birds need large areas for exercise, she maintains that feed management rather than spatial conditions may be a more probable cause of leg problems in growing chicks. After observing problems of leg rotations, yolk sac retention, and stunted growth in ratite chicks on farms adding vitamin supplements to the feed of both emu and ostrich chicks, Raines witnessed a cessation in the occurrence of these problems when the only difference in management was not using the supplements.

Kestin *et al.* (1992) studied the relationship between occurrence of leg weakness and genotype in broiler chickens. When birds of different genotype were raised under conditions conducive to minimize leg problems, i.e. free range and low protein diet, evidence of leg weakness was still apparent. Kestin *et al.* concluded genetics was the probable factor for observed leg weakness and suggested that to reduce the occurrence of leg weakness, management practices should be investigated including feed, population density, and opportunity for exercise. Genetic factors should be considered in the long term as the best chance of eliminating leg weakness.

Diet and Growth

Nutrient requirements, feed efficiency, and growth data for young ratite chicks is limited in the scientific literature. However, the lysine requirement for emu chicks has been documented. O'Malley (1995) and Angel (1996) noted the lysine requirement for emu chicks had been determined by Mannion *et al.* (1995) to be .90 and .825 g lysine/MJ

ME for maximum growth and feed conversion, respectively. However, O'Malley recommended .80g lysine/MJ ME for chicks 0-8 weeks of age and lowering to .65g lysine/MJ ME from 8-20 weeks of age.

O'Malley (1995) also reported brief results of an unpublished study which tested diet preference of emu for energy versus protein. Energy was suggested to be preferred over protein. He recommended protein levels of 16.5% for emu chicks from 0-20 weeks of age.

In a nutrition study by Gandini *et al.* (1986), the effects of diet on growth were investigated for ostrich chicks eight and 10 days to eight weeks of age. The chicks were kept in 3.0 m X 2.5 m outside pens during the day and in shelters at night. Twenty chicks were fed separate mash diets providing 14%, 16%, 18%, and 20% protein, all having a ME of 2700 kcal/kg and 1.4% calcium. Results for weight gain among the diets showed that chicks fed the 20% protein diet had the greatest weight gain (9.134kg) and chicks fed the 14% protein diet had the lowest gain (5.438), but these differences were not significant. Feed conversion was best (1.65) for the 18% diet, slightly better than 20% diet (1.69), while the 14% diet was less efficient at 2.19, but again the differences were not significant.

During week six and seven of the study three chicks fed 20% diet and one chick from both the 16% and 14% diet groups developed leg deformities. Gandini *et al.* (1986) suggested the leg deformities were aggravated by insufficient dietary calcium. Ullrey and Allen (1996) concluded high energy-low fiber diets, along with lack of adequate exercise increase occurrence of leg problems. Also, rapid growth due to high

dietary energy levels especially in combination with the higher 20% protein diet were suggested to increase the incidence of leg deformities (Ullrey and Allen, 1996 and Vohra, 1992). Vohra after emphasizing this point made a contradictory suggestion for target values of 18-20% protein and 2600-2700 kcal/kg ME in formulating ratite chick diets.

Blue-McLendon and Bailey (1994) reported no leg problems during their study of ostrich chicks up to 14 months old. Through eight weeks of age, chicks in this study were fed *ad libitum* a mixture of a ratite starter diet and a ratite grower diet. The investigators reported the diet fed to chicks up to eight weeks included 20% protein, 2200 kcal/kg ME, and 12% fiber. An average eight week weight gain of 14.4 kg was much higher than that reported by Gandini (1986). The results of this study showed an opposite effect of the 20% protein level in the previous study, but is more in agreement with the proposed level by Vohra (1992). However, this study yielded better results with lower energy levels than suggested by Vohra. Blue-McLendon and Bailey did not begin measuring feed consumption until 12 weeks of age. Moreover, neither feed consumption nor feed conversion was given.

Ullrey and Allen (1996) reported that Ullrey (unpublished, 1982) conducted a study with 39 emu chicks similar to the study with ostrich chicks by Gandini *et al.* (1986). Four groups of emu chicks from one day to three weeks of age were kept in 2 m X 6 m pens having indoor-outdoor carpeting, then moved to 4 m X 4 m pens with sand floors where they were housed through 12 weeks of age. Each pen was assigned one of four diets. Diets 1 and 3 contained 18.5% protein, 2349 kcal/kg ME and 8% crude fiber

compared to diets 2 and 4 having 22% protein, 2249 kcal/kg ME, and 10% crude fiber. Calcium was higher in diets 3 and 4 (1.6%) than in diets 1 and 2 (1.2%). Abnormal legs occurred in three chicks fed the lower protein-higher energy diets with different calcium amounts, one from pen 1 (1.2% calcium) and two from pen 3 (1.6% calcium). No leg problems were observed in chicks on diets 2 and 4 with higher protein-lower energy. From this study, diet 4 (22% protein, 2249 kcal/kg ME, and 1.6% calcium) became the basis for a generic ratite feed used at the San Diego Zoo and San Diego Wild Animal Park for 12 years.

Ullrey and Allen (1996) support the concept of one diet for all ages for two reasons. The first being that there is not sufficient data available to determine what the requirements are for any age group. Second, most farmers and zoos don't have enough different aged birds for it to be economical to purchase separate starter, grower, and breeder feeds. Growth data was not reported by Ullrey and Allen for this 1982 study.

A nutritional study of emu chicks from approximately 4 to 17 weeks of age conducted by Waterhouse and Haley (1997) found a significant difference in feed conversion among four diets of 20%, 22% 23.5%, and 25.5% protein levels with 2460-2526 kcal ME/kg. The 22% protein diet had the best feed conversion at 3.4 and the 25.5% protein diet had the poorest ratio at 3.9. Chicks fed the 25.5% protein diet were reported to have a significant lower weight gain than the diets providing 20%, 22%, and 23.5%. Although there was no significant difference for gain among the three later diets, the 20% protein diet had the highest average gain. These investigators did not report

dimensions of the pens, floor type, or any occurrence/absence of leg problems during their study.

MATERIALS AND METHODS

Artificial Incubation

Experiment 1

Eighty-nine emu eggs were donated by the Oregon and Washington Emu Associations. Members collected the eggs produced at their individual farms. Information accompanied each egg identifying the donor, the date the egg was laid, the breeder pair, ages of breeder pair, and the breeder diet. The eggs were either transported directly to the OSU Department of Animal Sciences' egg storage unit or to central collection points where the eggs were gathered, then collectively transported to the department's egg storage unit. The eggs were received in two groups one week apart.

The first group of eggs arrived at the storage unit over a three day period. Upon arrival, all eggs were stored at approximately 12.8°C until all eggs were received. Excessive dirt was removed by brushing the egg with a coarse, dry paper towel. A felt tipped marker was used to number the individual eggs. The egg numbers were recorded along with the information from the accompanying data sheets supplied by the producer. All data collected for each egg and resulting chick were recorded throughout the study with reference to the respective egg number. The eggs were weighed individually to the nearest gram on a Toledo Honest Weight™ gram scale and the weights were recorded

before setting the eggs into one of two Jamesway 252 incubators. The egg racks of the Jamesway 252 incubators were modified to hold the larger emu eggs.

Seventeen eggs randomly selected from the first group of 25 were set horizontally in the incubator trays. The incubator was kept at an average temperature of 36.5°C and an average wet bulb temperature of 26.1°C, yielding an average calculated relative humidity (RH) of 43%. The wet bulb temperature was increased to an average temperature of 28.0°C (53% RH) on day 47 of incubation when eggs were transferred into hatching baskets modified to reduce the possibility of chicks jumping out. The number of days from lay to set ranged from 7-13 days for this group.

Eight of the original 25 eggs remained in storage for an additional eight days to be set with the second group. Once the delivery of the second group of eggs (64) was complete, the 64 eggs, plus the 8 eggs from the first group, were cleaned, numbered, weighed, and recorded in the same manner as the first group. The eggs were placed in a second Jamesway 252 incubator with 15 of the smaller sized eggs set in a vertical position. The remainder of the eggs were placed in the incubator horizontally. The incubator was maintained at an average incubation temperature of 36.7°C with an average wet bulb temperature of 25.4°C (41% RH). On day 47 of incubation, the eggs were transferred into individual hatching baskets and the wet bulb temperature was increased to an average of 27.7°C (50% RH). The number of days from lay to set ranged from 6 to 29 days for the second group and 11 to 19 days for the eight eggs held back from the first group.

The eggs in both incubators were automatically turned every 3 hours through transfer into the hatcher at 47 days of incubation. All eggs were weighed and candled weekly through day 47 to determine developmental status and to calculate weight loss. An infrared egg candler (Ratek Industries) was used for candling the eggs. A video recorder was connected to the candler to record the candling of selected eggs. Those eggs appearing infertile or abnormal were broken open and the contents examined. Breakout results were recorded for each egg and photos were taken to document infertile blastodiscs and various stages of the contents of selected eggs for abnormal and apparent normal development in dead embryos. Embryo development stages were based on chicken embryo development stages described by Hamburger and Hamiltion (1951).

Eggs appearing viable on day 47 of incubation were transferred into individual hatching baskets to allow for pedigree of the chicks. After the eggs were transferred, they were no longer turned. Hatching baskets were lined with excelsior to prevent hatched chicks from spraddling. The Jamesway 252 incubator used to incubate the first group of eggs was also used as a hatcher for both groups of eggs.

Hatching was monitored approximately every four hours over a period of 10 days after transfer into the hatcher. The hatched chicks were identified by placement of numbered metal Tab End Style #898 wing band (National Wing Band & Tag Company) bent in a circular shape and closed around the distal end of the tarsometatarsus of each chick. After banding, each chick was weighed on the Toledo gram scale and the hatch weights recorded. The chicks were returned to the hatching baskets in the hatcher until they were completely dry, then transferred into brooder pens. All eggs not hatched by

day 57 of incubation were broken out and contents examined to determine stage of development and/or abnormalities. The results of breakouts were recorded and selected embryos were photographed.

Experiment 2

Twenty-three emu eggs were acquired from a single farm. The date the egg was laid and the identity of the breeder pair were provided. The eggs were collectively transported directly from the farm to the university. All eggs were numbered and weighed as described for Experiment 1 and set horizontally into a Jamesway AVN incubator, designed for incubation of ratite eggs. The incubator was maintained at an average temperature of 36.0°C with an average wet bulb temperature of 24.7°C (41.0% RH) up to day 49 of incubation. The number of days from lay to set ranged from 3 to 62 days.

All eggs were candled and weighed weekly through day 49 and the weights recorded. The eggs were automatically turned every 3 hours up to the day of transfer. On day 49 the eggs were transferred into group hatching baskets with excelsior floors and placed into an adjacent Jamesway AVN hatcher. The hatcher was maintained at an average temperature of 35.3°C with an average wet bulb temperature of 26.1°C (48.0% RH). After hatching, chicks were banded, weighed and placed back into the hatcher as described in Experiment 1. All eggs appearing clear or dead at candling and all eggs not hatched by day 56 of incubation were broken open and contents examined to determine fertility, stage of development, and abnormalities.

Experiment 3

One-hundred and twenty-nine eggs were donated by the Oregon and Washington Emu Associations and collected and stored in a similar manner as described for Experiment 1. Eggs were received over a period of 3 days and placed in a storage room maintained at an approximate temperature of 12.8°C until set. All eggs were cleaned, numbered, weighed, and recorded as described for Experiment 1. An AB 0.001 inch shock-proof caliper scale was used to measure the length and width of each egg. The egg index of each egg was calculated by dividing the width of the egg by the length. The eggs were randomly selected and placed horizontally into one of two Jamesway 252 incubators or a Jamesway AVN incubator. The incubators were maintained at an average temperature of 36.6°C, 36.0°C, and 35.8°C, respectively. The average wet bulb temperatures of the three incubators were 25.2°C (41.0% RH), 24.7°C (40.0% RH), and 25.0°C (41.0% RH), respectively. The number of days from lay to set ranged from 2 to 22 days.

Candling and weighing of selected eggs was done on days 10, 20, 30, and 40 of incubation. Selected eggs were recorded at each candling via video tape. Two eggs from each temperature group appearing to have normal development at candling on days 10, 20, 30, and 40 of incubation were broken out and examined to determined the stage of development. Photos were taken to document stage of development in relation to the day of incubation. Eggs appearing clear or having dead embryos were broken open and examined. Photo documentation was made of selected contents (Appendix A).

On day 46 of incubation, the incubation temperatures for the three incubators were lowered 0.5°C and the wet bulb temperatures were increased approximately 1.0°C. Methods following transfer of the eggs to the hatcher through post hatch weighing, banding, and return to the hatcher were the same as described previously for Experiment 1.

Egg shell thickness was determined by averaging measurements taken at three separate locations along the length of the hatched or broken shells using a Fowler & NSK Max-Cal electronic digital caliper scale.

Analyses of Incubation

Due to unbalanced sample size of eggs, GLM Proc (SAS, 1994) simple linear regression analysis was performed to establish a relationship between hen age and preincubation parameters of set weight, egg shell thickness, and egg index. Eggs from the three experiments were grouped for analysis of binomial results using logistic regression (Proc Logistic, SAS, 1995) of the effects of hen age on fertility. The binomial results of hatchability for temperature and incubator type were analyszed using logistic regression (Proc Logistic, SAS, 1995). Binomial incubation results were analyzed by logistic regression (Proc Logistic, SAS, 1995).

Chick Growth

Detailed records of management activities and observations were recorded daily. Feed and water was provided *ad libitum* throughout these experiments.

Experiment 1

Chicks were placed in one of six experimental pens measuring 3.04 m long X 0.91 m wide X 0.91 m tall inside a non-insulated metal sided building. The front and side panels of the pens were constructed of chicken wire stretched and stapled over a wood frame. One of the 3.0 m sides was secured to an interior plywood wall. The cement floor of the pens was insulated with approximately a 7.5-10.0 cm layer of coarse sand. The sand was covered by Mighty Gripper Nonskid material to provide foot traction for young chicks and to minimize their consumption of sand.

At approximately one week of age, chicks from group one in Incubation Experiment 1 were moved from temporary brooders, randomly selected, and placed into pens A and B. Chicks from group two were transferred from the hatcher, randomly selected, and put in pens C-F. Pens A, C, and F were fed a chick starter mash diet having a protein level of 20.1% and 2961 kcal/kg metabolizable energy. The three remaining pens, B, D, and E, were given a broiler starter mash feed containing 21.4% protein and 3016 kcal/kg metabolizable energy.

Each pen was provided with a hanging heat lamp, a waterer, and a feeder. The heat lamps were hung approximately 1.5m from the front end of the pens. Chick

waterers with volume of 3.8 liters were used to provide water for the first 2 weeks. Round metal pans measuring 30.5cm in diameter by 5.1cm deep were used as feeders for the same period. The chick waterers and metal feeders were replaced at 3 weeks with buckets 24.1 cm square by 33.0 cm deep. Holes approximately 14.0cm square were cut in the front side of the buckets to give chicks easy access to the feed and water. The water buckets and feeders in each pen were hung by the handles at opposite ends of the pens to encourage the chicks to exercise and to avoid crowding at the feeders. The feeders and water buckets were raised as the chicks grew to prevent the chicks from defecating into the feed or water. The water buckets were replaced with buckets without the holes as the chicks grew tall enough to drink from the top of the buckets. The heat lamps were raised as needed to maintain an appropriate environmental temperature for the chicks based on their behavior. Heat lamps were on 24 hours a day up to 3 weeks of age, then only at night thereafter through 6 weeks of age when the heat lamps were removed. All pens were cleaned daily and chicks were checked twice per day on a routine schedule.

Chicks and feed were weighed every two weeks through 8 weeks of age to determine the average weight gain and feed intake per pen. Chicks were weighed by placing them individually in a 113.6 liter plastic container used to confine the birds for weighing on an O-Haus kg/lb floor scale. The container was lined with Mighty Gripper Nonskid™ material to minimize the potential for leg injuries due to the smooth plastic surface of the container.

The metal leg bands were replaced two separate times as the chicks grew. The first replacements were metal turkey wing bands, similar to the original bands, but larger. The second replacements were Velcro bands individually numbered with a marker. These were adjusted as needed throughout the remainder of the experiment.

The individual pen was considered the experimental unit. Due to an unbalanced sample size in the experimental units, statistical analyses were completed using a one-way analysis of variance GLM Proc (SAS, 1995). If a significance was found, mean data separation was revealed by t-test (GLM Proc, SAS, 1994) and least significant difference (GLM Proc SAS, 1995). Alpha levels were set to be 0.05 to determine significance.

Experiment 2

The pens for this experiment were pens E and F used in Experiment 1 (Figure 1). The pen construction was slightly modified for Experiment 2 by attaching a 1.27 cm X 30.5 cm board at the bottom of adjacent sides of the pens along the length sandwiching the chicken wire between the boards. This modification was to minimize the possibility of chicks toes becoming entangled in the chicken wire. Eleven chicks hatched in incubation Experiment 2 were randomly selected and alternately placed in the two adjacent pens, 6 chicks in the first pen, 5 chicks in the second pen. Chicks in both pens were fed a commercial gamebird starter crumble diet having 30% protein and 2578 kcal/kg. Wood shavings bedding was placed in the first at a depth of 7.5-10.0cm, while a 7.5 - 10.0 cm layer of chopped annual rye grass straw served as litter for the second

pen. Chick waterers, 3.8 liter capacity, were used in both pens up to the chicks being 1.5 weeks of age. For the same duration, flat plastic trays 16.5 cm wide X 58.4 cm long X 3.8 cm deep lined with rubber matting were used for feeders. The rubber lining was to prevent the chicks from slipping if they stepped in the feeder. After 1.5 weeks the feeders and waters were replaced with the buckets having holes used during chick growth Experiment 1. A second hole was cut in the feeder bucket to allow more chicks access to feed. Feeders and water buckets were placed at opposite ends of the pens and raised as the chicks grew. When the chicks were 3 weeks of age, automatic waterers replaced the bucket waterers and an additional feeder was added to each pen. Pens were spot cleaned twice daily and litter replaced once a week during the first week and as needed there after. The original metal leg bands were replaced two separate times as the chicks out grew the diameter of the bands. The first replacements were ½" plastic circular turkey leg bands. The second replacements were similar to the plastic turkey leg bands, but expandable, self adjusting as the chick's leg increased in diameter.

Methods used for weighing chicks and feed were similar to those described for chick growth Experiment 1. Growth measurements of the tarsometatarsus, middle toe, and beak were taken weekly from one to ten weeks of age. A Fowler & NSK Max-Cal™ electronic digital caliper was used to measure the length of the middle toe from the top of the nail to the distal end of the last (proximal) scale on the middle toe, and the beak from the proximal end of the nostril to the tip of the beak. An AB 0.001 inch shock-proof caliper was used to measure the tarsometatarsus from the bend of the hock to the distal end of the tarsometatarsus. Observational analyses were used to

determine effectiveness of substrates to absorb droppings and provide secure footing for chicks.

Analysis of feed samples to determine actual levels of protein, NDF, ADF, calcium, phosphorus, sodium, potassium, iron, zinc, copper, and manganese was done by DHI Forage Testing Laboratory, Ithaca, New York. Analysis of amino acid levels was done by AAA Laboratory, Mercer Island, Washington. Net energy level was provided by the feed manufacturer.

Experiment 3

This study was a 2 X 3 factorial design using two pen sizes, 6 large and 6 small, and three commercial crumble diets with different protein levels of 18%, 23%, and 28% protein donated by feed mills in western Oregon. Metabolizable energy levels were 2640, 2513, and 2650 kcal/kg, respectively. The small pens were the same 6 pens used in the Experiment 1, but having the same construction modification described in Experiment 2. The 6 large pens measured 11 m long X 1.83 m wide X 1.52 m tall. These pens were constructed with hog panels supported by a steel frame. The hog panels were lined with chicken wire to prevent chicks from putting a leg through large holes in the hog panels and injuring or breaking a leg. All pens were covered with a 7.5-10.0 cm layer of wood shavings as litter. Forty-eight chicks were grouped by post-hatch age for 1-2 day, 3-4 day, 5-6 day, 7-9 day. Then one chick was randomly selected from each age group in sequence and assigned to one pen to achieve an even age

distribution among the pens. Feed and water containers, leg bands, and replacements were the same as described for Experiment 2. Growth measurements of the tarsometatarsus, middle toe, and beak were done weekly from one to ten weeks of age using the same methods described for chick growth in Experiment 2.

All chicks were raised in the small pens and fed the same 30% protein commercial gamebird diet fed to chicks in Experiment 2 until three weeks of age. At three weeks of age, the chicks were given the study diets. The chicks assigned to the large pens were placed in their respective pens and all pens were given the assigned study diet. There were two replicate pens for each diet in both pen size groups. The chicks were allowed one week of transition to adjust to the new diet and pen size. From four to ten weeks of age the chicks and feed were weighed weekly.

Laboratory analysis of samples taken from each of the diets to determine actual levels of protein, NDF, ADF, calcium, phosphorus, sodium, potassium, iron, zinc, copper, and manganese was done by DHI Forage Testing Laboratory, Ithaca, New York. Analysis of amino acid levels was done by AAA Laboratory, Mercer Island, Washington. Metabolizable energy levels were provided by the respective feed mills.

The individual pen was considered the experimental unit with diet and pen size serving as the main effects. Statistical analyses were completed on the main effects using two-way analysis of variance GLM Proc (SAS, 1995). If a significance was found, a GLM one-way analysis of variance was performed for each component of diet and pen size. Mean data separation was revealed by t-test (GLM Proc, SAS, 1994) and least

significant difference (GLM Proc, SAS, 1995). Alpha levels were set to be 0.05 to determine significance.

RESULTS AND DISCUSSION

Artificial Incubation

Landauer (1961 and 1973), Lundy (1969), and Beer (1969) have provided extensive reviews of research pertaining to the numerous factors affecting the hatchability of artificially incubated avian eggs. Many of these factors occur prior to incubation such as breeder age, breeder diet, genetics, method of collecting and handling of eggs, egg storage conditions, and pre-incubation egg storage duration.

The present study had no control over any of the variables occurring prior to incubation which influence hatchability. Due to unbalanced sample size of eggs, potential outliers, and the variability among the preincubation characteristics of hen age, breeder diet, genetics, egg handling, and egg storage any significant statistical associations among the variables tested were not considered conclusive. The following analyses of incubation data were reported as observational and used to indicate patterns which can be utilized as base-lines for future investigations.

Hatchability has been defined as the percentage of eggs that hatch from the total number of eggs set regardless of fertility or condition of the chicks (Landauer, 1961; Lundy, 1969). However, Lundy suggested a more applicable definition is the percentage of fertile eggs that hatch (those having the ability to hatch), but only if a researcher differentiates between true infertile eggs and early dead in shell events by careful examination of the content of all eggs by the end of incubation. Hatchability in the

following discussion is defined as the percentage of fertile eggs that hatched since eggs were closely examined for fertility.

Pre-incubation Storage

Hatchability results indicated an increase in the duration of pre-incubation egg storage to be associated with a decrease in hatchability of emu eggs. The highest percent hatch (63.8%) (Table 1) occurred for eggs held seven days or less and decreased sequentially for each additional week of storage. This pattern is similar to results reported by Funk *el al.* (1950) for chickens, Kosin (1954) for turkeys, and Chahil and Johnson (1974) for quail. Funk and Kosin reported a progressive decline in eggs held longer than seven days. Chahil and Johnson found a decrease in hatchability of Japanese quail eggs after only one day of storage. All emu eggs contributed to the current study were stored at least two days prior to incubation, thus it was not determined if there is an advantage to setting emu eggs immediately after lay.

Funk (1934) and Chahil and Johnson (1974) concluded that a longer incubation period was required for eggs stored for extended periods prior to incubation. However, results in the present study showed no indication that the length of incubation was affected by the duration of pre-incubation storage of emu eggs (Table 1). Eggs stored longer than 14 days required approximately the same or less time for incubation as eggs stored 14 days or less.

Table 1. Influence of egg storage duration from lay to set on length of incubation and hatchability of emu eggs.

	Number of Days Stored			
	≤7	8-14	15-21	≥21
Total fertile eggs	47	97	23	8
%Hatchability	63.8 (30)[+]	52.6 (51)	47.8 (11)	42.8 (3)
Incubation length (days)	51.9±1.6 SD	51.7±2.0 SD	51.4±1.0 SD	52.0±1.0 SD

(5)[+] Total hatched eggs

Incubation

Temperature and Incubator Type

No standard parameters were established for artificial incubation of emu eggs at the time of this study, therefore several strategies were used to test incubator temperature, humidity, and type to achieve maximum hatch. Temperature and humidity levels were based on those used by farmers asserting high hatch rates. Individual trials of the three experiments were compared to test affects of different temperature or incubator type on hatch.

Romanoff (1936) reported that higher temperatures shorten incubation duration and decrease hatchability, while lower temperatures increase incubation time and delay hatch time. Data in the present study showed similar results that higher incubation temperatures were associated with a decrease in hatchability. Incubation temperatures of $\leq$36.0°C had an average hatchability of 64.3%, while incubation temperatures >36.0°C had an average hatchability of only 47.2% (Table 2). Individually the three trials with temperatures $\leq$36.0°C resulted in higher percent hatchability than any of the three trials averaging temperatures >36.0°C (Table 2). However, there was no evidence of a difference in hatch spread (Table 2). The AVN incubator run at 36.0°C resulted in the lowest hatch spread of three days in contrast to the nine day hatch spread of eggs in the Jamesway 252 incubator run at the same temperature. The difference was probably not due to incubator type, but due to the fact that the AVN eggs were placed into multiple hatching baskets. This incubator was the only incubator that had group hatching baskets

Table 2. Influence of incubator type and temperature on hatchability of emu eggs and viability of hatched chicks.

	AVN Incubator		252 Incubator			
Mean Temperature	36.0°C*	35.8°C*	36.0°C*	36.6°C*	36.7°C**	36.5°C**
% RH[#]	41/48	41/48	40/47	41/48	41/50	42/53
Total eggs set [@]	22	24	43	45	72	17
Set eggs, %hatch	54.5%	58.3%	55.8%	46.7%	38.9%	41.2%
Fertility %	59.1%(13)[+]	91.7% (22)	81.4% (35)	82.2% (37)	83.3% (60)	76.5% (13)
Hatchability %	92.3%(12)[++]	63.6% (14)	54.3% (19)	45.9% (17)	46.7% (28)	53.8% (7)
Incubation length (days)	52-54	50-54	49-57	50-55	48-57	50-53
Hatch spread (days)	3	5	9	6	10	4
Chick weight/set egg weight ratio	65 ±.022	65 ±.030	65 ±.022	66 ±.027	66 ±.036	66 ±.022
Chick mortality 0-8WOA	1	5	2	2	6	3
Aver. Hatchability%		64.3%			47.2%	

[#] Incubator relative humidity / hatcher relative humidity.
*Temperature decreased 1°C at transfer.
**Temperature constant throughout incubation.

[@] Totals do not include eggs broken open to stage embryo development
(5)[+] Total fertile eggs
(5)[++]Total hatched eggs

rather than individual hatching baskets. These results support those reported by Cannon *et al.* (1986) and Freeman and Vince (1974) who found eggs in contact or close proximity with other eggs stimulate synchronous hatching.

The highest incubation temperature (36.7°C) had the only occurrence of hemorrhage in the chorioallantois membrane (CAM) in late incubation just prior to hatch. French (1992) found high temperatures during artificial incubation of turkey eggs resulted in high embryo mortality during the last half of incubation, which included a high occurrence of hemorrhages. The Jamesway 252 incubator at 36.0°C had only a slightly better percent hatch (54.3%) than the Jamesway 252 incubator with the highest temperature at 36.7°C (46.7%) (Table 2), however the lower temperature had no hemorrhaging in late stage mortality.

Percent hatch results for the Jamesway 252 incubators indicated no advantage to lowering the temperature 0.5°C at transfer as reported by Romanoff (1936). Percent hatch for the incubators having temperature lowered at transfer were similar to the percent hatch results of the Jamesway 252 incubators temperatures kept constant at transfer (Table 2).

Romanoff (1936) reported that temperatures producing the maximum hatch resulted in the greatest hatch weight to initial egg weight ratio. He theorized that these ratios may serve as a measurement of incubation efficiency. The current study results with emu eggs do not support Romanoff's theory. Although incubation temperatures >36.0°C for emu eggs had a higher hatch weight to set egg weight ratio than those ≤36.0°C, the higher temperatures were not those resulting in maximum hatchability

among the emu incubation trials (Table 2). Trials having lower incubation temperatures (≤36.0°C) resulting in best hatchability had chick hatch weight to set egg weight ratios of 0.65. Temperatures >36.0°C resulting in lower hatchability were calculated to have ratios of 0.66. Both of the ratios calculated in the present study are below the ratio of 0.67 that Minnaar and Minnaar (1992) have suggested as being the optimum ratio for emu hatchlings, however, their ratio was not determined by controlled studies. Further studies are needed to determine the optimum ratio for emus.

Hatchability was higher in the AVN incubators resulting in an average hatch of 74.2% than in the Jamesway 252 incubators which had an average hatch of 49.0%. The highest hatchability of 92.3% occurred in the AVN incubator having a temperature of 36.0°C (Table 2). This result may have been due to the difference in incubators, since both tests using AVN incubators resulted in higher hatchability than any of the four tests run in the Jamesway 252 incubators (Table 2). However, as noted previously, the higher success at 92.3% hatchability of the AVN at 36.0°C possibly resulted from a difference in the number of eggs per hatching basket. Again, these results support those reported by Cannon *et al.* (1986) and Freeman and Vince (1974) who found eggs in contact or close proximity with other eggs stimulate the hatching process, thus producing a higher hatch than eggs not next to other eggs. Since all the eggs in the AVN run at 36.0°C were from a single farm, other factors possibly attributing to higher percent hatch include genetics, hen diet, hen age, handling practice, and storage conditions.

Romanoff (1936) reported lower temperatures produce higher quality chicks. Supporting Romanoff's conclusion French (1992) found hatched poult quality decreased

with an increase in incubation temperatures. Data in the present study showed similar results that lower temperatures increased chick quality. Higher chick quality was observed at lower incubation temperatures. Chicks hatched in Experiment 2 in the AVN incubator run at 36.0°C were visibly stronger and more active than those hatched in the other experiments. Again, all eggs in this trial were from a single farm and only four different emu breeder pairs. Therefore, it was impossible to conclude if the higher quality chicks were due to incubator type, temperature, or any of the other possible factors affecting artificial incubation of eggs noted previously.

Chicks hatched during Experiment 3 at slightly lower temperature (35.8°C), but in the same AVN incubator used in Experiment 2 and chicks hatched at the same temperature of 36.0°C in a Jamesway 252 incubator in Experiment 3 were not as strong or active as the chicks hatched in Experiment 2. However, all three trials individually produced better quality chicks than the chicks hatched in the trials at temperatures >36.0°C. Unexpectedly chick mortality to eight weeks of age among the three higher temperatures was only slightly greater than chicks hatched at lower temperatures (Table 2).

Although it was reported earlier in this discussion that lowering the incubation temperature 0.5°C at transfer did not affect the number of chicks actually hatched, it was observed that the lowering of the temperature did improve chick quality. The largest group of weak chicks resulted in the two Jamesway 252 incubators during Experiment 1. These incubators were maintained at constant temperatures of 36.7°C and 36.6°C throughout incubation. Also, the occurrence of deaths from hemorrhagic CAM just

prior to hatch occurred in the incubator with the highest temperature (37.7°C). The higher temperature may have functioned as an external stimulus to cause premature hatching resulting in the increased incidence of weaker chicks at hatch and death from hemorrhaging just prior to hatch.

Meir and Ar (1990) recorded a higher temperature of 2.0°C in egg content surface when measuring the difference between the temperature of egg content surface and the temperature in the incubator. Butterman *et al.* (1988) reported temperatures in naturally incubated emu eggs increased to 36.0°C by day 35 and remained constant through the remainder of incubation. Incubation trials in the current study in which temperatures were lowered at transfer had higher quality chicks and lower incidence of pre-hatch mortality due to CAM hemorrhaging than those trials with constant temperatures throughout incubation. The findings of the two previous studies indicate that incubator temperatures of 36.0°C in the present study were possibly as much as two degrees centigrade higher than optimum for emu eggs.

Further investigations are needed to establish an optimum incubation temperature and protocol for artificial incubation of emu eggs. These investigations should include control of factors occurring prior to incubation that can affect hatch to provide uniform and balanced trial samples.

Egg Weight Loss

The majority of hatch occurred in eggs having a weight loss of <15% during incubation from set through transfer (Table 3). Weight losses were lower than the 15% loss predicted by Rahn (1979) for all avian species. However, average mean hatched egg weight loss of 13.6% ±2.7 SD (Table 3) was higher than the optimum 10% loss for emu eggs reported by Buttemer *et al.* (1988). Although the majority hatch occurred in eggs showing weight loss within the range of 11.0%-14.9% (Table 3), a few eggs successfully hatched outside this range having losses as low as 8.8% and as high as 18.1%. Ar *et al.* (1994) reported a similar egg weight loss range of 12%-15% for ostrich eggs, having the best hatch at 13% loss.

Until further studies are conducted, results of the present study suggest a target weight loss of less than 15% to be less detrimental than losses ≥ 15%. Future studies for incubation weight loss should encompass determination of egg shell porosity and water vapor conductance.

Hen Age

Emu hens ≤2 years of age had lower percent fertility than older hens. Analysis using SAS logistic regression revealed significant evidence ($p<0.05$) that as hen age increases, egg fertility increases (Table 4). Highest fertility (95.8%) occurred in eggs laid by hens ≥ 7 years of age. The best hatchability (86.9%) also occurred within this age

Table 3. Influence of temperature on egg weight loss and hatchability of emu eggs.

	Percent Weight Loss					
	≤8.0	9.0-10.9	11.0-12.9	13.0-14.9	15.0-16.9	≥17
36.0°C (13 eggs)[+]						
% hatched		15.4%	30.8%	30.8%	15.4%	
% non-hatched		7.4%				
36.5°C (10 eggs)						
% hatched		10.0%	10.0%	20.0%		30.0%
% non-hatched				10.0%	10.0%	10.0%
36.7°C (52 eggs)						
% hatched	1.9%	7.7%	11.5%	25.0%	5.8%	1.9%
% non-hatched			13.5%	9.6%	13.5%	9,6%
All Temps. (75 eggs)						
% hatched	1.3%	9.3%	14.7%	25.3%	6.7%	5.3%
% non-hatched		1.3%	9.3%	8.0%	10.8%	8.0%

(5)[+] Total fertile eggs up to transfer.

Table 4. Influence of hen age on fertility and hatchability of emu eggs.

	Hen Age (yrs)			
	≤2	3	4-6	≥7
Total eggs set	98	61	44	24
%Fertility	74.4[a] (73)[+]	90.2[c] (55)	88.6[b] (39)	95.8[d] (23)
Total fertile eggs expected to hatch*	65	48	37	23
%Hatchability	52.3%(34)[++]	45.8% (22)	45.9% (17)	86.9% (20)

a,b,c,d Different letters indicate significant difference ($p<.05$) for each column
$(5)^{+}$ Total fertile eggs
$(5)^{++}$Total hatched eggs
* Totals do not include eggs broken open to stage embryo development

group. These results are contradictory to the lower fertility and hatchability in older chicken hens reported by Fasenko *et al.* (1992a).

There are no studies reporting the maximum peak reproductive age for emu hens or at what age a hen is to be considered reproductively old compared to old chicken hens. However, emu farmers assert a higher reproductive success for older hens on their farms.

Emu hens ≤ 2 years of age averaged the lowest average mean set egg weight at 509 ±60g compared to older hens (Table 5). A pattern appearing in Table 6 indicates that as hens age there is less probability ($p<0.05$) of producing a small egg. The highest percentage of eggs produced by hens ≥ 4 years of age weighed over 550g compared to the highest percentage for hens ≤ 3 years of age weighing ≤ 550g. This is in agreement with the investigations reviewed by Landauer (1961) reporting that as hen age increases there is an increase in egg size. In contrast, hens ≥ 7 years of age laid eggs averaging slightly less (591±59g) than eggs from hens 4-6 years of age (606±73g). Future studies should research if there is a peak in egg weight as hens age and at what hen age might egg weight begin to decline.

Analysis of shell thickness measurements showed no evidence ($p>0.05$) that hen age influenced shell thickness (Table 5). Due to the imbalance of the sample size for the different hen age groups, results reported here should be considered with caution. Breeder diet and genetics are also attributing factors in set egg weight and egg shell thickness and should be considered in future studies.

Table 5. Influence of hen age on set weight, shell thickness, and egg index.

	Hen Age (yrs)			
	≤2	3	4-6	≥7
Average set weight (g)	509 ±60[a] (98)[+]	558 ±59[b] (61)	606 ±73[d] (44)	591 ±59[c] (24)
Average shell thickness (mm)	.77 ±.08[a] (43)	.77 ±.10[a] (42)	.76 ±.07[a] (30)	.78 ±.04[a] (4)
Average egg index (width ÷ length)	.667 ±.049[a] (39)	.654 ±.029[a] (42)	.660 ±.026[a] (31)	.665 ±.039[a] (4)

a,b,c,d Different letters indicate significant difference ($p<.05$) for each column.
$(5)^{+}$ Total eggs measured

Table 6. Influence of hen age on egg set weight as a percentage of eggs set for each age group.

	Egg weight (g)						
	≤400	401-450	451-500	501-550	551-600	601-650	>650
Hen age (yrs)							
≤2 (98)[+]	3.1%	14.3%	24.5%	37.8%	14.3%	4.1%	2.0%
3 (61)	--	4.9%	9.8%	39.3%	18.0%	24.6%	3.3%
4-6 (44)	--	--	11.4%	9.1%	29.5%	27.3%	22.7%
≥7 (24)	--	--	4.2%	25.0%	16.7%	45.8%	8.3%

(5)[+] Number of eggs within age group

Tserveni-gousi and Yannakopoulos (1995) found as egg weight and hen age increase, the egg shape index decreases. In the present study, age of the emu hens revealed no indication ($p>0.05$) of influencing egg index (Table 5).

Set Egg Weight

Egg weights ≤500g had the lowest hatch at 48.6%, while eggs >650g had the highest hatch at 75% (Table 7). Medium sized eggs 501g to 650g had a hatchability of 54%. Results showed as set weight increased, there was an increase in hatch. These figures contradict the findings reported by Landauer (1961) that medium sized eggs have the highest hatchability. Ar *et al.* (1994) also found medium sized eggs to produce the highest hatchability for ostrich eggs. Deeming (1995a) reported a significant drop in hatchability for large ostrich eggs. In relation to maximum hatchability, optimum emu egg size has not been defined.

Shell Thickness

Hatchability values in Table 7 indicate a possible inverse relationship between hatchability and egg shell thickness. Hatchability decreased as egg shell thickness increased. Mean shell thickness for emu eggs was 0.77 ±0.08 mm and the mean emu egg weight for this group of eggs was 560 ±80 g.

Brown *et al.* (1996) reported a significant correlation between egg shell thickness and egg weight in ostrich eggs with heavier eggs having thicker shells. Comparing egg

Table 7. General pattern for influence of egg weight, egg shell thickness, and egg index (shape) on hatchability of emu eggs.

	Set Egg Weight (g)				
	≤500	501-550	551-600	601-650	>650
Total eggs set	37	72	40	41	16
% Fertility	64.9	79.2	75.5	90.2	100.0
% Hatchability	48.6 (18)[+]	56.1 (32)	48.4 (15)	54.1 (20)	75.0 (12)

	Egg Shell Thickness (mm)				
	≤.71	.71-.75	.76-.80	.81-.85	.86
Total eggs measured	26	23	25	16	21
% Fertility	73.0	92.0	95.6	87.5	73.7
% Hatchability	68.0 (13)	65.2 (15)	54.5 (12)	50.0 (7)	21.4 (3)

	Egg Index (width ÷ length)									
	≤.62	.63	.64	.65	.66	.67	.68	.69	.70	≥.71
Total eggs measured	9	9	9	23	12	13	13	9	5	10
% Fertility	88.8	100.0	100.0	91.3	91.7	69.2	84.6	66.7	60.0	80.0
% Hatchability	25.2(2)	55.6(5)	66.7(6)	61.9(13)	63.6(7)	44.4(4)	36.4(4)	83.3(5)	33.3(1)	25.0(2)

(5)[+] Total of fertile eggs hatched

weight and shell thickness of emu eggs no evidence ($p > 0.05$) was found to support a correlation between shell thickness and egg weight. A larger sample size is needed to determine if a relationship exists between emu egg shell thickness, egg weight and hatchability.

Egg Index

Although there was no pattern showing that egg index influenced hatch of the emu eggs (Table 7), eggs having the lowest egg index (very long narrow eggs) and eggs with the highest egg index (short round eggs) had the lowest percent hatch (Table 7). These observations are consistent with results reported by Landauer (1961) that very long, narrow eggs, and short, round eggs seldom hatch.

Embryo Development and Egg Content Morphology

Results below for each of the three incubation experiments are summarized in Table 8. Eggs incubated in a vertical position during Experiment 1 are reviewed separately in the results below. Embryo developmental stages were proportionately based on the development stages for the chicken embryo described by Hamburger and Hamilton (1951). Selected photos of candling and egg breakout content are described in Appendix A. Photo documentation and description of normal 10, 20, 30, and 40 day embryo development from Experiment 3 are found in Appendix A.

Table 8. Summary of artificial incubation of emu eggs as a percentage of total eggs set* and total fertile eggs set.

	TES	TH*	FT*	FH	LD	MD	ED	BWE	PD	FND	HC	MP	INF*	MY*	UN*
EXP. 1															
36.7°C v	17	11.8	64.7	18.2	45.5	27.3	9.1				18.2	45.5	23.5	5.9	11.8
36.7°C	55	47.3	83.6	56.5	32.6	4.3	2.2	2.2	2.2		8.7	6.5	10.9	3.6	5.5
36.5°C	17	41.2	76.5	53.8	15.4		15.4		15.4			15.4	23.5	5.9	
EXP. 2															
36.0°C	22	54.5	54.1	92.3					7.7				31.8	13.6	9.1
EXP. 3															
36.6°C	45	37.8	82.2	45.9	24.3	10.8	10.8	2.7	5.4			5.4	17.8		
36.0°C	43	44.2	81.4	54.3	25.7	5.7	5.7		8.6			2.9	16.3		2.3
35.8°C	24	58.3	91.7	63.6	4.5	18.1	4.5		4.5	4.5		4.5	8.3		

v = Vertically positioned eggs
TES = Total number of eggs set
TH* = Percent eggs hatched of total eggs set
FT* = Percent fertile of total eggs set
FH = Percent hatched of total fertile eggs set
LD = Percent late stage embryo death of total fertile eggs set
MD = Middle stage embryo death of total fertile eggs set
ED = Early stage embryo death of total fertile eggs set

Table 8 continued.

BWE = Percent blastoderm without embryo of total fertile eggs set
PD = Percent positive development of total fertile eggs set
FND = Percent fertile with no development of total fertile eggs set
HC = Percent hemorrhagic CAM of total fertile eggs set
MP = Percent malpositions of total fertile eggs set
INF* = Percent infertile of total eggs set
MY* = Percent of mottled yolks of total eggs set
UN* = Percent fertility unknown of total eggs set

In Experiment 1, eggs were incubated in two separate Jamesway 252 incubators, maintained at either 36.7°C or 36.5°C throughout incubation. Eggs set at 36.7°C included 17 eggs set in a vertical position, air cell up. During incubation several of the vertically positioned eggs had rolled over into a horizontal position and were repositioned to vertical. Of the seventeen eggs, two chicks hatched, nine embryos died in the shell, four eggs were infertile, and fertility was not determined for two eggs, one appeared infected and the other had a mottled yolk. The first chick to hatch had a clubbed foot. The second chick had a hemorrhagic CAM and was stuck to the shell. It required assistance in hatching. However, the chick was very weak and died at four days of age.

As mentioned previously , Landauer (1961) and Ar *et al.* (1994) reported medium sized eggs have the highest probability of hatching. The majority of the seventeen eggs in this trial were small size averaging 455g, therefore biasing any conclusions for affect of incubation position on hatch that might be offered.

Of the nine dead in shell embryos, five were fully developed (late dead, ≥46 days of incubation). One of the five had pipped the air cell and had a partially absorbed yolk. Three were malpositioned (Hamburger and Oppenheim, 1967) upside-down and had yolks not absorbed and one of the three had a hemorrhagic CAM. Two embryos were malpositioned upside-down with beak in the yolk. Upside-down malposition was also observed for the chick with the clubbed foot that successfully hatched.

An early dead (<17days incubated) embryo was documented at Stage 20± and appeared normal. Abnormal mid-dead (18-45 days incubated) embryos at approximately

Stage 40-43 (34-42 days of incubation) were observed in three of the nine unhatched eggs. All three were dwarfed and two of these had reduced legs and deformed beaks. The deformed beaks had maxilla curled upward, a reduced mandible curled down. This deformity is similar to that reported as Donald Duck beak (Abbott and Lantz, 1975). One of the eggs containing a Donald Duck dwarfed embryo, an egg with the dwarfed embryo, and an infected egg had been stored 16, 19, and 15 days, respectively. All other eggs including the two hatched eggs were stored ≤ 14 days.

The other 55 eggs incubated at 36.7°C with the 17 vertically positioned eggs above resulted in 26 hatched chicks, 18 embryos dead in the shell, one blastoderm without embryo (BWE), one positive development (PD), and six infertile. Fertility was not determined for three eggs, including one infected and two that had mottled yolks. Eight of the hatched eggs had been stored >14 days prior to incubation with the longest storage at 27 days. The remaining 18 hatched eggs varied in storage length from 6-14 days. The two eggs containing mottled yolks were stored 10 and 15 days. Pre-incubation storage for the other eggs in this trial varied from 14 to 29 days.

As the last tray of eggs for this group was placed into the incubator at set, the tray tipped over and four eggs dropped approximately one foot to the bottom of the incubator. Since none cracked, the eggs were replaced into the tray and set in the incubator. All but one of the four dropped eggs hatched, and of the three hatched, one had been originally collected from a cold mud puddle. All three chicks were healthy. These results may indicate that emu eggs are not as fragile as one might expect. Four other chicks that had been malpositioned upside-down pipped the bottom of the shell.

Two of these chicks successfully hatched and two (one having a curved toe) were assisted in hatching and survived.

Fifteen late dead embryos included two with yolks approximately 80% absorbed; one malpositioned upside-down with yolk out and a hemorrhagic CAM; two malpositioned with beak between toes including one with a hemorrhagic CAM; five pipped internally including two normal, one with yolk out, two with hemorrhagic CAM and one of these had a ruptured yolk; one dwarfed with yolk out; two normal with ruptured yolk; and two infected. Mid incubation deaths revealed a dwarfed embryo with subcutaneous hemorrhage and an embryo with a brain hernia and edema. Only one embryo died early in incubation. It was estimated to have died at about Stage ±23.

Seventeen eggs in a second Experiment 1 trial were incubated at 36.5°C in a second Jamesway 252 incubator resulting in seven hatched chicks, four dead in shell embryos, two PD, and four infertile. One chick successfully hatched even though it had been malpositioned upside-down in the shell. Another hatched from an egg that was very light in color. Abbott (1975) stated a light colored egg from a hen that normally lays eggs with dark pigment may be evidence that the egg was laid too early. Under these conditions, light colored eggs seldom hatch. It was unknown if light pigment was typical in eggs from the hen producing the light colored egg in the present study or if the light colored egg was prematurely laid.

Two late dead embryos had pipped the air cell and yolks were not absorbed, the one malpositioned with head between its legs. An early dead embryo was found similar to Stage 30 and was ceolosomic. Another embryo at approximately the same stage had

subcutaneous hemorrhage. The four infertile eggs included one with a mottled yolk and one with uric acid plaques. Pre-incubation storage for all eggs in this trial was ranged from 6-13 days.

Eggs in Experiment 2 were incubated at a temperature of 36.0°C in an AVN ratite incubator and transferred at day 46 into an AVN hatcher with the temperature decreased 0.5°C. Out of 22 eggs set, there were 12 hatched, one PD, seven infertile, and two unknowns, possibly infected. One chick was stuck in the shell after pipping and was assisted in hatching. It was very weak and died the next day. All other hatched chicks were strong and active. Seven chicks hatched from eggs stored <14 days. Five chicks hatched from eggs stored 14 to 22 days. Mottled yolks occurred in three of the infertile eggs. These eggs had been stored 30, 42, and 49 days. The remaining four infertile had normal yolks and had been stored >49 days prior to incubation.

Experiment 3 involved three trials having three different temperatures and two incubator types to incubate eggs. The first trial involved 53 eggs incubated at 36.6°C in a Jamesway 252 incubator and transferred at 46 days with the temperature decreased 0.5°C. Results revealed 17 hatched chicks, 17 dead in shell embryos, two PD, one BWE, and eight infertile. Two of the hatched eggs had been stored 15 and 16 days. One of the PD eggs was stored 22 days. All other eggs were stored <14 days. Eight eggs appearing normal during candling on days 10, 20, 30, and 40 of incubation were broken open and selected photos are described in Appendix A.

Examination of nine late dead embryos revealed one normal with internal pip, three with yolks partially absorbed, two upside-down malpositions including one with

yolk partially absorbed, one edema with yolk partially absorbed, and two full term infected. At approximately Stage 40-44, two mid-dead embryos had subcutaneous hemorrhage, a third was infected, and a fourth had a crossed beak and was missing the left eye. Four early-dead embryos described at approximately Stage 23-28 of development included one coelosomic, one extremely dwarfed, and two normal in appearance. A double yolk was discovered in one of the infertile eggs.

In the second trial, 52 eggs were incubated in a second Jamesway 252 incubator run at 36.0°C and transferred at day 46 with the temperature decreased 0.5°C. Nine of the 52 eggs were broken open to document embryo development on days 10, 20, 30, and 40 of incubation. Selected photos are described in Appendix A. Incubation results revealed 19 hatched chicks, 13 dead in shell embryos, three PD, seven infertile, and one unknown.

Nine late-dead embryos included three which had pipped the air cell, but the one had the yolk slightly out (5%). Two had yolks approximately 75% absorbed with one having attempted pip of the air cell. Three embryos were dried out and had yolks partially absorbed, and one had no absorption of the yolk. Developmental stage was approximately Stage 36-43 for two dwarfed mid-dead embryos. One early-dead was estimated to be at Stage 13 with the heart and head formed. A second early-dead was considered at Stage 23 of development. Possible infection was noted for one of the infertile eggs. Egg storage time prior to incubation was 16 days for one hatched egg. The remainder of the eggs in this trial were stored ≤ 11 days.

The third trial of eggs in Experiment 3 was incubated at 35.8°C in an AVN incubator and transferred into an AVN hatcher with the temperature maintained at 0.5°C lower. Out of 24 eggs set, 14 hatched, and there was one late-dead, 4 mid-dead, one early-dead, one PD, one fertile with no development (FND), and two infertile. One chick was assisted in hatching after it became stuck in the shell for two days. This chick had difficulty keeping its neck straight. The neck curled down and caused the head to roll under the chest. The chick was culled at 6 days of age when the condition became more severe.

The late-dead embryo was malpositioned with its head between the legs and yolk partially absorbed. Mid-dead embryos included a Donald Duck beak and a dwarfed embryo with a crooked neck. Both embryos were hemorrhagic and about Stage 38-40. A second embryo was discovered having a Donald Duck beak and was estimated to be at Stage 41-43. Another embryo about the same stage was found to have a crooked lower beak. An early-dead embryo about Stage 25 was found to be hemorrhagic. Pre-incubation storage time was <14 days for all eggs except one. The egg containing the Stage 38-40 Donald Duck beak had been stored for 16 days.

The one FND egg was described as having the blastodisc lifted up from the surface of the yolk similar to the shape of a volcano. Incidence of FND is rare and occur in eggs held in storage too long or exposed to very low temperatures over a period of several days prior to incubation (Abbott, 1975).

Although chicks successfully hatch when upside-down in the shell, their chance for successful hatch is reduced. Malpositions are affected by egg shape, position of the

egg during incubation, low incubation temperatures, improper turning, and some gene mutations (Abbott, 1975). Hen diets deficient in vitamins A and D and in linoleic acid have been reported to increase the number of malpositions (Landauer, 1973). Cause of the incidence of malpositions in present study was difficult to determine. An improper turning effect may have caused malposition in the eggs set vertically in Experiment 1 which toppled over and were reset to a vertical position. A larger sample size is needed to determine if egg incubation position has an effect on hatch or the occurrence of malpositions.

Deeming (1995b) has cautioned that malposition described for domestic fowl may not be applicable to ostriches because normal positions the ostrich assumes before hatching were found to be different than in the domestic fowl. Emu farmers have claimed emu embryos to have a different hatching process than domestic fowl. It is possible that emu embryos also adopt a different position than described for domestic fowl. Detailed examination of emu embryo hatching process is lacking in the scientific literature and until full descriptions of the process are documented, reported malpositions of emu embryos should be carefully weighed.

Abbott and Lantz (1975) described Donald Duck beak as an autosomal recessive mutant. The embryo described in trial 3 of Experiment 3 at Stage 41-43 as having a crooked lower beak may have been a less severe expression of a Donald Duck beak mutant. The Donald Duck beak has been categorized as a lethal embryonic mutation, however Landauer (1961) reported a chick with this mutation and assisted at hatch survived to $7^1/_2$ weeks of age.

Dwarfed embryos have been identified as genetic lethal mutation in poultry by Asmundson (1945). However, dwarfed embryos have been reported by Beer (1969) to occur from nutritional deficiency of riboflavin, biotin, B_{12}, or manganese. Beer also reported a deficiency in pantothenic acid to cause subcutaneous hemorrhage. Investigations involving heredity and nutrition are needed to determine the cause of the abnormalities found in emu embryos. It was difficult to make any conclusions as to the causal effect of abnormalities seen in the present study since breeder diet varied for the eggs incubated and genetic history was unknown.

The occurrence of PD or BWE can have several possible causes. These include rough handling of eggs, improper storage temperature (>70 F), disease in breeders, inbreeding, and parthenogenesis (Abbott, 1975). It was impossible to determine the causal effect of PDs and BWEs in the present study.

Fasenko *et al.* (1992b) demonstrated an increase in mottled yolks in eggs stored ≥ 14 days before incubation compared to eggs stored ≤ 7 days. Results in the current study found no conclusive evidence to support Fasenko *et al.*'s analysis. Four mottled yolks were discovered during breakout in Experiment 1, however only one had been stored longer than 14 days. The other three were stored for 7, 9, and 10 days, respectively. No mottling was observed in 18 additional eggs stored ≥ 14 days nor in 23 eggs stored <14 days. Experiment 2 produced three mottled yolks in eggs stored 30, 42, and 49 days. Although seven additional eggs had been stored longer than 14 days ranging from 24 to 62 days, no mottled yolks were observed. Experiment 3 revealed no

mottled yolks during breakout of 62 eggs, however only two unhatched eggs had been stored longer than 14 days.

Abbott (1975) and Landauer (1961) described two mortality peaks during incubation. The first occurring early in incubation caused by the failure of one or more organ systems to form. The second and larger peak occurs in late incubation. This peak is related to the embryo's change in position to prepare for hatching, utilization of albumen, absorption of the yolk, and the shift from allantoic to pulmonary respiration. Mortality falling between the two peaks is normally low and due to nutritional deficiencies or genetic mutation. Incubation results reported as late dead (LD), middle dead (MD), early dead (ED) in Table 8 display a pattern similar to the two mortality peaks described by Abbott (1975). However, there was a higher than expected mortality occurring between the two peaks. The results reported here confirm the need for investigations to determine the nutritional requirements of emu.

The percent hatches of the total eggs set (Table 8) were similar to the hatchability results reported by Ford (1994). This hatchability rate is unacceptable for a successful production operation and another strong indicator of the important need for more investigation embracing incubation parameters and management, nutritional requirements, and genetic influences.

Chick Growth and Management

Diet and Growth

The first seven chicks hatched in Exp. 1 were excluded in the diet and growth analysis due to the one week difference in age from the second group hatched. Experiments 1 and 3 each had four chicks removed at less than one week of age (WOA). Exp. 2 had one chick removed at less than one WOA. Chicks removed included one with an injury from a fall out of the hatcher tray, three weak at hatch, two with deformities, two with spraddle legs, and one with an infected yolk sac. These chicks were not included in the growth analysis. Analysis of the experimental diets are listed in Table 9.

In Exp. 1, 13 chicks fed a 21.4% protein broiler starter (BS) diet with 3016 kcal ME/kg had higher weight gain ($p<0.05$) than nine chicks fed a 20.4 % protein chick starter (CS) diet having 2961 kcal ME/kg (Table 10). At eight WOA, chicks on the BS diet had a mean weight gain of 5.36 ±0.14 kg compared to a mean of 4.87 ±0.16 kg for chicks fed the CS diet. Feed conversion ratio through eight WOA was significantly better at 1.98 ($p<0.05$) among chicks fed the BS diet than the ratio of 2.13 for chicks fed the CS diet (Table 10). The was no difference in feed intake between the two diet groups.

Eleven chicks given a commercial 30% protein gamebird starter (GB) diet with 2578 kcal ME/kg during Exp. 2 gained an average of 5.17 ±0.74 kg per chick up to eight WOA. Feed conversion was calculated to be 2.54. Average tarsometatarsus length at

Table 9. Laboratory analysis of ingredients and amino acids in diets.

Ingredient as Sampled	Broiler Starter *	Chick Starter *	Gamebird	All in One	Emu	Turkey / Gamebird
% Crude Protein	21.36	20.43	30.30	19.20	23.70	29.20
ME kcal/kg **	3016.00	2961.00	2578.00	2640.00	2513.00	2650.00
%ADF	2.40	2.72	4.20	2.80	5.10	1.80
%NDF			10.00	9.90	15.90	6.00
%Calcium	0.81	1.29	1.40	4.03	8.20	1.28
% Phosphorus	0.71	0.88	1.03	0.81	0.93	0.85
% Magnesium	1564.48 mg/kg	1690.13 mg/kg	0.29	0.19	0.22	0.19
% Potassium	0.84	0.82	1.44	0.74	1.20	1.18
%Sodium	0.16	0.27	0.234	0.173	0.423	0.149
ppm Iron	35.88 mg/kg	112.60 mg/kg	296	267	420	341
ppm Zinc	51.46 mg/kg	51.30 mg/kg	152	94	290	201
ppm Copper	4.23 mg/kg	13.30 mg/kg	24	15	37	21
ppm Manganese	77.34 mg/kg	78.56 mg/kg	164	102	236	175
ppm Molybdenum			3.60	1.9	2.10	1.70

(*ingredient amounts calculated) **values provided by feed mill

Table 9 continued.

Amino Acid as Sampled	Broiler Starter *	Chick Starter *	Gamebird	All in One	Emu	Turkey / Gamebird
% Alanine			1.526	1.162	1.203	1.485
% Arginine	1.440	1.360	2.245	1.228	1.398	1.795
% Aspartine			2.995	1.479	1.963	2.253
% Cystine	0.360	0.350				
% Glucamine			5.355	2.940	3.626	4.231
% Glycine	1.420	1.360	1.574	1.354	1.355	1.878
% Histidine	0.490	0.470	0.783	0.438	0.516	0.627
% Isoleucine	1.030	0.980	1.314	0.644	0.878	1.001
% Leucine	1.860	1.800	2.325	1.414	1.564	1.878
% Lysine	1.130	1.070	1.740	0.921	1.297	1.622
% Methionine	0.440	0.380	0.435	0.258	0.345	0.566
% Phenylalanine	1.120	1.070	1.462	0.798	1.014	1.171
% Pro			1.992	1.466	1.639	1.945

*ingredient amounts calculated

Table 9 continued.

Amino Acid as Sampled	Broiler Starter *	Chick Starter *	Gamebird	All in One	Emu	Turkey / Gamebird
% Serine	1.140	1.090	1.583	0.903	1.068	1.269
% Threonine	0.870	0.840	1.225	0.693	0.874	0.991
% Tyrosine	0.790	0.750	1.022	0.551	0.670	0.807
% Valine	1.090	1.030	1.480	0.834	1.073	1.224

(*ingredient amounts calculated)

Table 10. Effect of diet on feed consumption, weight gain, and feed conversion in emu chicks raised on sand up to 8 WOA.

	Feed consumption (kg)	Weight gain (kg)	Feed conversion
EXP 1 (diet)			
Broiler Starter	$10.49_a \pm 0.15$	$5.35_a \pm 0.14$	1.98^a
Chick Starter	$10.35_a \pm 0.17$	$4.87_b \pm 0.16$	2.13^b

a,b Different letters indicate significant difference ($p<.05$) for each row

eight WOA was 21.6 cm with a mean growth of 13.63 ±0.71 cm per chick from one to eight weeks. Final middle toe length was measured at 5.98 ±0.40 cm with mean growth of 3.13±0.40 cm. Mean beak growth was 0.90 ±0.09 cm to a length of 2.29 ±0.09 cm at eight WOA (Table 11).

In Exp. 3, 44 chicks were raised in two different pen sizes and fed three separate commercial diets; an 18% protein all-in-one poultry (AIO) 2640 kcal ME/kg diet, a 23% protein emu (EMU) 2513 kcal ME/kg diet, and a 28% protein turkey-gamebird starter (TG) 2650 kcal ME/kg diet. Results revealed no interaction between pen size and diet among the variables of weight gain, feed conversion (Table 12), or growth of tarsometatarsus, middle toe, and beak (Table 13). However, there was a significant difference in the main effect of diet between the EMU diet and the TG diet for feed intake (Table 12). Chicks fed the EMU diet consumed significantly more feed ($p<0.05$) than chicks fed the TG diet. The results showed that chicks gained no advantage in weight gain ($p>0.05$) (Table 12) or growth ($p>0.05$) (Table 13) by consuming higher amounts of the EMU diet.

Litter in pens fed the EMU diet for the present study was observed to become wet litter in half the time required by litter in pens fed the AIO and TG protein diets. Laboratory analysis of the diets (Table 9) indicated a higher content of fiber and Na in the emu diet than the other two diets. High levels of either fiber or Na may have been the possible cause of the higher moisture content in the litter (Morrison, 1961) of pens fed the emu diet.

Table 11. Effect of diet on feed consumption, weight gain, feed conversion, and growth of beak, middle toe, and tarsometatarsus in emu chicks raised up to 8 WOA.

	Feed consumption (kg)	Weight gain (kg)	Feed conversion	Beak (cm)	Middle Toe (cm)	Tarsometatarsus (cm)
EXP (diet)						
Gamebird	13.15 ±0.44	5.17 ±0.74	2.54	0.90 ±0.09	3.13 ±0.40	13.63 ±0.71

Table 12. Effect of diet on feed consumption, weight gain, and feed conversion in emu chicks from 4-10 WOA.

	Feed consumption (kg)	Weight gain (kg)	Feed conversion
EXP 3 (diet)			
All In One	15.53_{ab} ±0.55	5.30_{a} ±0.26	2.95^{a}
Emu	16.78_{a} ±0.55	5.39_{a} ±0.26	3.12^{a}
Turkey / Gambird	14.37_{b} ±0.55	5.03_{a} ±0.26	2.87^{a}
EXP 3 (pen size)			
Small	15.97_{ab} ±0.45	5.23_{a} ±0.21	3.06^{a}
Large	15.15_{ab} ±0.45	5.25_{a} ±0.21	2.89^{a}

a,b Different letters indicate significant difference ($p<.05$) for each row

Table 13. Effect of diet on growth in emu chicks from 1-10 WOA.

	Beak (cm)	Middle Toe (cm)	Tarsometatarsus (cm)
EXP 3 (diet)			
All In One	1.24$_a$ ±0.04	3.98$_a$ ±0.13	16.78^a ±0.29
Emu	1.16$_a$ ±0.04	3.93$_a$ ±0.13	17.03^a ±0.29
Turkey / Gamebird	1.16$_a$ ±0.04	3.74$_a$ ±0.13	16.88^a ±0.29
EXP 3 (pen size)			
Small	1.21$_a$ ±0.03	3.84$_a$ ±0.10	16.69^a ±0.24
Large	1.15$_a$ ±0.03	3.94$_a$ ±0.10	17.10^a ±0.24

a,b Different letters indicate significant difference ($p<.05$) for each row

The birds fed EMU diet had the highest overall average weight gain while the TG diet had the lowest among the three diets, however neither was significant (Table 12). Although no significant difference was found for feed efficiency among the diets, average overall feed conversion was best for the TG diet at 2.87, and poorest for the EMU diet at 3.12 (Table 12).

In a nutrition study by Gandini *et al.* (1986), twenty ostrich chicks were raised in 3 m X 2.5 m pens and fed four separate diets providing 14%, 16%, 18%. and 20% levels of protein. Each of the diets contained 2700 kcal ME/kg and 1.4% calcium. Results showed chicks fed the 18% protein diet had the best feed conversion of 1.65 at 8 WOA. Highest weight gain was in the chicks fed the 20% protein diet and lowest for those fed the 14% protein diet. During weeks six and seven of the study leg deformities occurred in five chicks, one chick each from the 14% and 16% protein diet groups and three chicks from the 20% protein diet group. These researchers suggested the leg deformities had been generated by insufficient dietary calcium. Ullrey and Allen (1996) and Vohra (1992) summarizing this study suggested the leg deformities to be caused by rapid growth due to high energy level (2700 kcal ME/kg) in combination with high protein (20%) in the diet. Ullrey and Allen were in agreement with Vohra, but also included low fiber level along with the high levels of energy and protein.

Ullrey (unpublished, 1982) conducted an earlier study with 39 emu chicks from one day old through 12 WOA. The chicks were randomly placed in groups given one of four diets. Two diets had 2350 kcal ME/kg providing 18.5% protein and 8.0% fiber each with different calcium levels of 1.2% and 1.6%. The other two diets had 2250 kcal

ME/kg providing 22% protein and 10.0% fiber each with different calcium levels of 1.2% and 1.6%. Ullrey reported abnormal legs developed in chicks fed the two diets with 18.5% protein, one was from the group with 1.2% calcium in the diet and two were from the group with 1.6% calcium in the diet. The two diets providing higher protein (22.0%), lower energy (2250 kcal ME/kg) and higher fiber (10.0%) resulted in no leg problems. Therefore, calcium was not considered as the causal factor for the leg problems, and it was suggested that the occurrence of abnormal legs was related to lower protein in combination with higher energy and lower fiber.

Cause of chick mortality including culled chicks with leg problems in the present study was inconclusive, however mortality did not appear to be induced by diet. There was no evidence in the present study to support the suggested dietary effects reported by Gandini *et al.* (1986) and Ullrey (unpublished, !982). The 13 chicks fed the BS diet in Exp. 1 with the lowest level of calcium at 0.81% and the highest ME levels of 3016 kcal ME/kg among the experimental diets had one chick develop a leg problem at 5 WOA (Table 14). The CS diet had four mortalities between one and three weeks including two sudden death, one impaction, and one chick with both legs locked close to the body. No mortalities or leg problems occurred from one to ten WOA among the 16 chicks fed the EMU diet in Exp 3. There were no mortalities or leg problems one to eight WOA among the 11 chicks in Exp. 2 raised on a high 30.3% protein GB diet, the highest level among the study diets, with high energy of 2578 kcal ME/kg and 1.4% calcium. However, three (6.81%) of the 44 chicks in Exp. 3 fed the same GB diet as in Exp. 2 up to three WOA, developed leg problems (Table 14). One of the three chicks was noted to

Table 14. Effects of diet on mortality of emu chicks.

	1-2	2-3 WOA	3-4 WOA	4-5 WOA	5-6 WOA	>6 WOA
EXP. 1 (diet)						
Chick Starter	2	2	0	0	0	0
Broiler Starter	0	0	0	1	0	0
EXP 2 (diet)						
Gamebird	0	0	0	0	0	0
EXP 3 (diet)						
Gamebird	1	2	0	0	0	0
All In One	0	0	1	0	0	0
Emu	0	0	0	0	0	0
Turkey / Gamebird	0	0	1	0	0	0

have a leg problem prior to the second week of measurements and the other two chicks developed a leg problem within one and two days after the chicks had been handled. One of the 14 chicks fed the TG diet for less than one week developed a leg problem between 3-4 WOA. The leg problem developed within one day after the chicks were handled. An additional mortality in Exp. 3 resulted from a leg problem in a chick between 3-4 WOA fed the AIO diet for less than one week. This chick also had a pronounced crooked spine observed at an earlier age. After the fifth week measurements were taken, two chicks on the TG diet were observed to be limping, however the problems corrected within a week. No mortalities occurred beyond five WOA among the three diets.

Management

Spatial

Raines (1994) recommended chicks be raised with minimum square footage per bird of 40 sq. ft. per chick up to 9 kg or eight WOA, and 200 sq. ft. per chick over 9 kg. Wade (1992) stressed adequate space permitting sufficient exercise up to 12 WOA for ratite chicks. He recommended large pens with 200-400 sq. ft. per chick to minimize the occurrence of impaction and leg problems. These recommendations were not based on results of controlled studies. Ullrey and Allen (1996) suggested the probability of limited exercise as a plausible explanation of leg problems. However, both Raines and Wade suggested the cause of leg problems is probably multifactorial. Kestin *et al.* (1992) also

indicated leg problems to be a multifactorial problem and concluded genetics as a contributing component for observed leg weakness in broilers. Both Kestin *et al.* (1992) and Raines (1994) suggested feed management as a probable cause of leg problems.

Hansen and Becker (1960) showed decreased growth rate in chickens as population densities increased. Feather picking behavior was observed in birds stressed by limited feeding space due to high density.

Chicks in the present study were raised up to approximately 7 kg body weight at eight WOA and 9 kg at ten WOA in pens with ≤10 sq. ft. per chick and 54-72 sq. ft. per chick in small and large pens, respectively. In Exp. 3, pen size had no effect on emu chick growth. Chicks raised in small pens 0.91 m X 3.04 m showed no difference in weight gain ($p>0.05$), feed conversion ($p>0.05$) (Table 12), or growth of the beak, middle toe, and tarsometatarsus ($p>0.05$) (Table 13) than chicks raised in large pens 1.83 m X 11.0 m. Although chicks raised in the large pens had better gain and feed conversion than those in the small pens, neither was significantly better (Table 12).

Density did not appear to affect mortality rates. All mortalities including chicks culled due to leg problems occurred up to four WOA and in the small pens having four or less chicks. The five chick mortalities in Exp. 3 occurred within two small pens, three chicks in one pen and two in a second pen, with each pen having only 4 chicks per pen. Chicks in Experiments 1 and 2 raised in groups of five to seven chicks per pen, 4.29 sq. ft. to 6.00 sq. ft. per bird, had no mortality through 8 WOA. In Exp. 3, four of the six of the small pens with eight chicks per pen, <4.00sq. ft. per chick, had no mortalities for the first three weeks before half the birds were moved to larger pens. Chick behavior in high

density pens was observed to be similar to behavior of chicks raised in lower density pens and showed no indication that the birds were stressed.

One chick each in Experiments 2 and 3 that were observed to have strange behavior. Both birds when excited would walk or run backwards. The chick in Exp. 3 also displayed a behavior of bending its neck with the head down in a slight swaying motion between the legs. This behavior was similar to the "Bobber" behavior in turkeys described by Harper and Bernier (1972), and Harper *et al.* (1988). These researchers defined the behavior as a genetic disorder caused by an inherited recessive sex-linked gene. The chick displayed the behavior when excited and it would last only for a short period after the disturbance influencing the response ceased. However, the chick would also display the behavior when attempting to eat. The chick approached the feed tray with the neck held out straight parallel to the ground, with the head looking down rather than forward. As the chick reached the food tray and began to lower the head to eat, the neck curved under and the head went between the legs, simultaneously the chick would begin walking backwards. Several subsequent attempts would be made before the chick would either be successful or give up. This behavior faded completely by 10 WOA. The condition in turkeys was reported by Harper and Beinier (1972) to remain throughout the life of the bird. Although, Harper *et al.* (1988) found that the bobber condition was corrected within in two to four weeks after affected poults were exposed to natural sunlight 15-16 hours daily. The chick in Exp. 2, however, continued to walk backwards when excited and the condition did not correct even after the chick was moved to an outdoor pen at the end of the experiment. The cause of the abnormal behaviors in the

two chicks and what precipitated the correction of the condition was not determined in the present study. Future studies should include genetics affects.

Gandini *et al.* (1986) raised twenty ostrich chicks in 3 m X 2.5 m pens and fed four separate diets in the nutritional study discussed above. These authors did not indicate the probability of pen size as a possible cause for the development of leg problems during six and seven of the study. Ullrey and Allen (1996) made reference to lack of exercise as a possible cause of the leg problems in their summary of Gandini *et al.* 's nutritional study.

Substrate

In the unpublished study by Ullrey (1982) mentioned previously, emu chicks were raised in small pens, 2 m X 6 m, with indoor-outdoor carpeting for 3 weeks, then moved to 4 m X 4 m pens with sand substrate for the rest of the study period. Three chicks developed leg problems, however it was not noted at what age the chicks developed leg problems. Sand substrate may have been a possible cause for the leg problems.

Stewart (1994) suggested sand and straw provoke leg problems in ratite chicks and recommended flooring with good traction such as brushed concrete, pine shavings, or natural dirt. In the present study, it was observed that pine shavings provided the best footing for emu chicks raised in indoor pens. Droppings easily mixed from the surface of the litter to underneath as shavings were tossed by the movement of the chicks, thus

keeping the chicks relatively clean. In pens with sand or chopped grass as substrate, droppings became compacted on the surface of both substrates. The compaction was most apparent in the pen with grass clippings. A combination of chopped grass and droppings became packed on the bottom of the feet, causing chicks difficulty in walking. Thus, the chopped rye grass substrate was replaced with pine shavings. Pens having pine shaving as substrate were determined much less labor intensive, easier to clean, and required less frequent cleaning, than sand or grass clippings substrates.

SUMMARY

The success of today's poultry industry is the result of intensive research over many decades investigating artificial incubation, nutrition, genetics, and management techniques. Maximum hatchability is dependent on multiple factors occurring before and during artificial incubation. The effect of these parameters can have a direct or indirect effect on the success of the hatch and the quality of subsequent hatched chicks. Also, proper nutrition and management are essential for hatched chick development and growth. For a productive operation, it is imperative for the farmer to be aware of the effects of all the parameters and effectively control and manipulate these parameters to achieve maximum hatch and viability of chicks.

The consensus among emu farmers is the need to use incubators designed specifically for ratite eggs to achieve maximum hatchability of emu eggs. Differences in incubator type would mainly affect humidity level which is inversely related to temperature in the incubator Landauer (1961). Other differences in incubator design that affect humidity levels during incubation include air flow and the rate of air exchange between inside and outside the incubator. Research has determined the most important requirements for high hatchability of artificially incubated eggs are proper temperature, humidity, and turning of the eggs during incubation (Landauer, 1961: Lundy, 1969; Tullett, 1990). Results of the present study revealed temperatures between 35.8°C-36.0°C produced the higher hatch of fertile eggs and the better quality chicks compared to temperatures ≥36.0°C. The AVN incubator, designed for ratite eggs, produced a

higher hatch and better quality chicks than the Jamesway 252 incubators. It was difficult to conclude a difference in performance of the incubators due to the fact that one of the trials run in the AVN incubator had all eggs from one farm, therefore breeder birds were fed the same diet, and the eggs were handled, stored and transported in a similar manner. Also, the eggs in this trial were incubated with multiple eggs per hatching basket promoting synchronous hatching. All of these factors could have contributed to the higher success of this incubator. However, it was observed that temperatures 35.8°C-36.0°C regardless of incubator type resulted in better hatch and chick quality than the incubators run at higher temperatures.

Consensus also states that chicks require large pens for maximum excercise and a special emu diet containing high quantities of vitamins and minerals for adequate growth and prevention of leg problems. The high amounts of vitamins and minerals increase feed costs thereby increasing overall production costs. Analyses in the present study revealed no advantage to feeding a specialized emu diet or raising chicks in large pens up to 10 weeks of age.

It is a complex task to experimentally isolate any one parameter as to its effect, direct or indirect, on hatchability of an egg or viability of the hatched chick. The reader should be cautioned that this complexity suggests the results in the present study to be inconclusive. However, the results presented in this report can be used as a baseline for further research.

For the most efficient production of emu, the requirements for artificial incubation, nutrition, and spatial needs of the emu must be determined. In the short

term, future artificial emu egg incubation investigations should emphasize direct and indirect effects of breeder age and nutrition, preincubation egg storage, incubation temperature and humidity, and incubator type on hatchability and embryo development. For long term investigations, researchers should concentrate on genetic effects related to emu embryo viability, chick leg problems, and chick behavior.

The present study was limited by the fact that the donated emu eggs used in the incubation experiments were not from a uniform population sample, therefore yielding uncertain results. Until the requirements for emu production are established by further controlled research, emu farmers are encouraged to use the information presented in this report as a resource and as a basis in making choices for least cost production.

The following statements are based on the results of the present study and may be useful to emu farmers:

1. Breeders ≥4 years of age for maximum fertility and hatchability.
2. Store emu eggs no longer than seven days prior to incubation.
3. Incubate emu eggs at a temperature between 35.8 C - 36.0 C and decrease temperature 1.0 C at transfer.
4. Strive for a 13% (ranging 11%-14%) egg weight loss during incubation.
5. Candle eggs at two to three weeks using an infrared candler and remove any non-viable eggs.
6. Break open and examine removed nondeveloping eggs and unhatched eggs to determine true fertility and identify any abnormalities.

7. Emu chicks can be raised successfully on pine shavings substrate, indoors, in pens as small as 3.0 m X 0.9 m to 10 weeks of age.

8. Chicks can be grown successfully using a commercial poultry or gamebird starter diet *ad libitum* up to 10 weeks of age.

BIBLIOGRAPHY

Abbott, U.K., 1975. Causes of and corrections for poor hatchability. *Southeastern's hatchery-breeder flock management clinic*. July:1-13.

Abbott, U.K. and F.H. Lantz, 1967. A second occurrence of Donald Duck beak in chickens. *Journal of Heredity* **58**:240-242.

Angel, C. R., 1993. Research update: Age changes in digestibility of nutrients in ostriches and nutrient profiles of ostrich and emu eggs as indicators of nutrient status of the hen and chick. *Proceedings of the 1993 Association of Avian Veterinarians Annual Conference* 275-281.

Angel, C.R., 1996. A review of ratite nutrition. *Animal Feed Science and Technology* **60**(3-4):241-246.

Asmundson, V.S., 1945. Crooked neck dwarf in the domestic fowl. *Journal of Heredity* **36**:173-176.

Asmundson, V.S. and J.J. MacIlraith, 1948. Preincubation tests with turkey eggs. *Poultry Science* **27**:394-401.

Ar, A., Aizik, M.N. Campi, D., 1994. Standard Values of Ostrich Eggs. *International Hatchery Practice* **9**(2):25.

Ar, A., C.V. Paganelli, R.B. Reeves, D.G.Greene, and H. Rahn, 1974. The avian egg: water vapor conductance, shell thickness, and functional pore area. *Condor* **76**:153-158.

Beer, A.E., 1969. A review of the effects of nutritional deficiences on hatchability 93-108. In: *The Fertility and Hatchability of the Hen's Egg.* T.C. Carter and B.M. Freeman eds., Oliver and Boyd, Edinburgh, 199p.

Bertram, B. and Burger, A., 1981. Aspects of incubation in ostriches. *Ostrich* **52**:36-43

Blakers, M., S.J.J.F. Davies, and P.N. Reilly, 1984. *The Atlas of Australian Birds / Royal Ornithologists' Union.* Melbourne University Press, 738p.

Blue-McLendon, A. And C.A. Bailey, 1994. Research on the growth rates of ostrich chicks. *Main Conference Proceedings, Association of Avian Veterinarians* 131-132.

Booth D.T. and H. Rahn, 1990. Factors modifying rate of water loss from birds eggs during incubation. *Physiological Zoology* **63**:697-709.

Brown, C.R., D. Peinke, and A Loveridge, 1996. Mortality in near-term ostrich embryos during artifical incubation. *British Poultry Science* **37**(1): 73.

Burton, M. And R. Burton, eds., 1969. *The International Wildlife Encyclopedia* Volume 2. Marshall Cavendish Corporation, New York, 2800p.

Buttemer, W.A., L.B. Astheimer, B. Lee, and T.J. Dawson, 1988. Thermal and water relations of emu eggs during natural incubation. *Physiological Zoology* **61**(6): 483-494.

Buttemer, W. and T.Dawson, 1989. Body temperature, water flux and estimated energy expenditure of incubating emus (*Dromaius novaehollandiae*). *Comparative Biochemistry and Physiology, A Comparative Physiology* **94**(1):21-24.

Campbell, B. And E. Lack, eds., 1985. *A Dictionary of Birds.* The Brithish Ornithologist's Union. Town Head House, Calton, Waterhouse, Staffordshire, England, 670p.

Cannon. M.E., R.E. Carpenter, and R.A. Ackerman, 1986. Synchronous hatching and oxygen consumption of Darwin's Rhea eggs (*Pterocnemia pennata*). *Physiological Zoology* **59**(1):95-108.

Chahil, P.S. and W.A. Johnson, 1974. Effect of preincubation storage, parental age, and rate of lay on hatchability in *Coturnix coturnix japonica*. *Poultry Science* **53**(2):529-534.

Cho P., R. Brown, and M. Anderson, 1984. Comparative gross anatomy of ratites. *Zoo Biology* **3**:133-144.

Cracraft, J., 1974. Phylogeny and evolution of the ratite birds. *Ibis* **116**(4):494-521.

Crawford, R.D., ed., 1990. *Poultry Breeding and Genetics.* Elsevier, Amsterdam, 1123p.

Davies, S.J.J.F., 1978. The food of emus. *Australian Journal of Ecology* **3**(4): 411-422.

Davis, T.A., S.S. Shey, and R.A. Ackerman, 1988. Embryonic Osmoregulation: Consequences of high and low water loss during incubationof the chicken egg. *Journal of Experimental Zoology* **245**:144-156.

Dawson, T.J., R.M. Herd, and E. Skadhauge, 1985. Osmotic and ionic regulation during dehydration in a large bird, the emu (Dromaius novaehollandia): an important role for the cloaca-rectum. *Quarterly Journal of Experimental Physiology* **70**(3): 423-436.

Dawson, T., F.J. McNaughton, A.R. Goldsmith, and A.A. Degen, 1994. Ratite-like neoteny induced by neonatal thyroidectomy of European starlings, Sturnus vulgasis. *Journal of Zoology (London)* **232**(4): 633-639.

deBeer, G., 1956. The evolution of ratites. *Bulletin of the British Museum (Natural History) Zoology* **4**(2):59-70.

Deeming, D.C., 1995a. Factors affecting hatchability during commerical incubation of Ostrich (*Struthio camelus*) eggs. *British Poultry Science* **36**:51-65.

Deeming, D.C., 1995b. The hatching sequence of Ostrich (Strutiocamelus) embryos with notes on development as observed by candling. *British Poultry Science* **36**:67-78.

Duke, G.E., A.A. Degen, and J.K. Reynhout, 1995. Movement of urine in the lower colon and cloaca of ostriches. *Condor* **97**(1): 165-173.

Fasenko, G.M., R.T. Hardin, and F.E. Robinson, 1992a. Relationship of hen age and egg sequence position with fertility, hatchability, viability, and preincubation embryonic development in broiler breeders. *Poultry Science* **71**:1374-1383.

Fasenko, G.M., F.E. Robinson, and R.T. Harden, 1992b. Research note. Variability in preincubation embryonic development in domestic fowl. 2. Effects of duration of egg storage period. *Poultry Science* **71**:2129-2132.

Feduccia, A., 1996. The evolution of flightlessness, 231-289. In: *The Origin and Evolution of Birds.* Yale University Press, New Haven and London, 420p.

Ford, C.R., 1994. U.S. emu population census and growth projection study. *AEA News* September, 1994.

Fowler, M.E., 1991. Comparative clinical anatomy of ratites. *Journal of Zoo an Wildlife Medicine* **22**(2): 204-227.

Freeman, B. and M. Vince, 1974. *Development of the avian embryo.* Wiley & Sons, 362p.

French, N., 1992. Incubation temperature and pathology. *International Hatchery Practice* 7(2):31.

Funk, E.M., 1934. Factors influencing hatchability in the domestic fowl. *Missouri Agricultural Experiment Station Bulletin* **341**.

Funk, E.M., and H.V. Biellier 1944. The minimum temperature for embryonic development in the domestic fowl (*Gallus domesticus*). *Poultry Science* **23**:538-540.

Funk, E.M. and J. Forward, 1951. The effect of humidity and turning of eggs before incubation on hatching results. *Missouri Agricultural Experiment Station Bulletin* **544**.

Funk, E.M., J. Forward, and H.C. Kempster, 1950. Effect of holding temperatures on hatchability of eggs. *Missouri Agricultural Experiment Station Bulletin* **539**:

Gandini, G.C.M., R.E.J. Burroughs, and H. Ebedes, 1986. Preliminary investigation into the nutrition of ostrich chicks (*Struthio camelus*) under intensive conditions. *Journal of South Africa Veterinary Association* **57**:39-42.

Garnett, S., 1992. The action plan for Australian Birds, Australian Nature Conservation Agency (ANCA) at http://www.anca.gov.au.

Grzimek, B., ed., 1973. Ratites, 89-109. In: *Animal Life Encyclopedia* vol. 7 Birds I. Van Nostrand Reinhold Company, New York, Cincinnati, Toronto, London, Melbourne, p.579.

Hamburger V. and H. L. Hamilton, 1951. A series of normal stages in the development of the chick embryo. *Journal of Morphology* **88**:49-92.

Hamburger V. and R. Oppenheim, 1967. Prehatching motility and hatching behavior in the chick. *Journal of Experimental Zoology* **166**(2):171-204.

Hansen, R.S. and W.A. Becker, 1960. Feeding space, population density and growthof young chickens. *Poultry Science* **39**:654-660.

Harper, J.A. and P.E. Bernier, 1972. Bobber: a cervical ataxia mutant in the domestic turkey. *Poultry Science* **51**:1815.

Harper, J.A., P.E. Bernier, T.F. Savage, and D.J. West, 1988. Bobber: A cervical ataxic mutant in the domestic turkey responsive to sunlight. *Journal of Heredity* **79**:155-159.

Herd, R.M., 1985. Anatomy and histology of the gut of the emu *Dromaius novaehollandiae*. *Emu* **85**(1).

Hill, R., 1967. *Australian Birds.* Funk and Wagnalls, New York, 281p.

Houde, P., 1986. Ostrich ancestors found in the northern hemisphere suggest new hypothesis of ratite origins. *Nature* **324**:563-565.

Hoyt, D.F., 1976. The effect of shape on the surface volume relationships of bird's eggs. *Condor* **78**:343-349.

Hulet, R.M., V.L. Christensen, and L.G.Bagley, 1987. Controlled egg weight loss during incubation of turkey eggs. *Poultry Science* **66**:428-432.

Huxley, T.H., 1867. On the classification of birds and on the taxonomic value of the modifications of certain of the cranial bones observable in that class. *Proceedings of the Zoological Society of London* **1867**:415-472.

Jarvis, M., R. Keffen, And C. Jarvis, 1985. Some Physical Requirements for Ostrich Egg Incubation. *Ostrich* **56**:42-51.

Jenkins, F.A., Jr., K.P. Dial, and G.E. Goslow, Jr. 1988. A cineradiographic analysis of bird flight: wishbone in starlings is a spring. *Science* **241**:1495-1498.

Kestin, S.C., T.G. Knowles, and A.E. Tinch, 1992. Prevalence of leg weakness in broiler chickens and its relationship with genotype. *The Veterinary Record* **131**(9):190-194.

Kosin, I.L., 1954. Length of preincubation storage and heredity as factors affecting hatchability of turkey eggs. *Poultry Science* **33**: 24-28.

Landauer, W., 1961. *The Hatchability of Chicken Eggs as Influenced by Environment and Heredity.* Storrs Agricultural Experiment Station, College of Agriculture and Natural Resources, the University of Connecticut, Storrs, Connecticu, 278p.

Landauer W., 1973. *The Hatchability of Chicken Eggs as Influenced by Environment and Heredity.* (A Supplement to Monograph Number One and Number One Revised). Storrs Agricultural Experiment Station, College of Agriculture and Natural Resources, the University of Connecticut, Storrs, Connecticut, 54p.

Lundy, H., 1969. A review of the effects of temperature, humidity, turning and gaseous environment in the incubator on the hatchability of the hen's egg, 143-176. In:

The Fertility and Hatchability of the Hen's Egg. T.C. Carter and B.M. Freeman eds., Oliver and Boyd, Edinburgh, 199p.

Mannion, P.F., P.B. Kent, K.M. Trappet, G.W. Blight, 1995. Production and nutrition of emus Proceedings Australian Poultry Science Symposium 7:23-30. In: Angel, C.R. 1996. A review of ratite nutrition. *Animal Feed Science and Technology* **60** (3-4):241-246.

McDowell, S., 1948. The bony palate of birds. Part I. The Paleognathae. *Auk* **65**:520-549.

McGowan,C., 1984. Evolutionary relationships of ratites and carinates from the ontogeny of the tarsus. *Nature* **307**:733-735.

Meir, M. and A. Ar, 1990. Temperature at end of incubation for ostrich 2 C higher than surrounding incubator temperature. *Condor* **92**:556-563.

Minnaar, P., and M. Minnaar, 1992. *The Emu Farmer's Handbook.* Groveton, Texas: Induna Co.,178p.

Morrison, F.B., 1961. *Feeds and Feeding, Abridged. The Essentials of the Feeding, Care, and Management of Farm Animals, Including Poultry,* ninth edition. The Morrison Publishing Company, Clarement, Ontario, Canada, 696p.

Oliver, W.R.B., 1945. Avian evolution in New Zealand and Australia. *Emu* **45**:73

Olsen, M.W. and S.K. Haynes, 1948. The effect of different holding temperatures on the hatchability of hens' eggs. *Poultry Science* **27**:420-426.

O'Malley, P., 1995. Nutrition of ratites: Comparison of emu and ostrich requirements. Department of Agriculture, Western Australia.

Parsons, B., 1994. Emu farming in Flordia. *Main Conference Proceedings of the Association of Avian Veterinarians* 438-439.

Prager E., A. Wilson, D. Osuga, and R. Feeney, 1976. Evolution of flightless land birds on southern continent: transferrin comparison shows monophyletic origin of ratites. *Journal of Molecular Evolution* **8**(3): 283-294.

Proudfoot, F.G., 1964. The effects of plastic packaging and other treatments on hatching eggs. *Canadian Journal of Animal Science* **44**:87-95.

Proudfoot, F.G., 1966. Hatchability of stored chicken eggs as affected by daily turning during storage and pre-warming and vacuuming eggs enclosed in plastic with nitrogen. *Canadian Journal of Animal Science* **46**:47-50.

Proudfoot, F.G., 1969a. The handling and storage of hatching eggs, 127-141. In: *The Fertility and Hatchability of the Hen's Egg.* T.C. Carter and B.M. Freeman eds., Oliver and Boyd, Edinburgh, 199p.

Proudfoot, F.G., 1969b. Effect of packing orientation, daily positional change and vibration on the hatchability of chicken eggs stored up to four weeks. *Canadian Journal of Animal Science* **49**:29-35.

Rahn, H, and A.Ar, 1974. The avian egg; incubation time and water loss. *Condor* **76**:147-152.

Rahn, H., A. Ar, and C.V. Paganelli, 1979. How birds eggs breath. *Science American* **240**:46-55.

Raines, A.M., 1994. How to evaluate a ratite facility to aid in diagnosing chick mortality. *Main Conference Proceedings of the Association of Avian Veterinarians* 97-102.

Romanoff, A.L., 1936. Effects of different temperatures in the incubator on prenatal and postnatal development of the chick. *Poultry Science* **15**(4):311-315.

Romijn C. And W. Lokhorst, 1955. Chemical heat regulation in the chicken embryo. *Poultry Science* **34**:649-654.

Shane, S.M., 1996. Hatchery management in emu production. Chapter 8. In: Tully, T.N. and S.M. Shane, eds. 1996. *Ratite Management, Medicine, and Surgery.* Krieger Publishing Company, Malabar, Florida, 188p.

Silbey, C.G. and J.E. Ahlquist, 1990. *Phylogeny and Classification of Birds, a Study in Molecular Evolution.* Yale University Press, New Haven and London, 976p.

Stephenson, A.B., 1985. Position and turning of turkey eggs during storage prior to incubation. *Poultry Science* **64**:1279-1284.

Stewart, J.S., 1992. Ratite incubation. *Proceedings of the Association of Avian Veterinarians* 336-339.

Stewart, J.S., 1994. Ratite (ostrich and emu) industry management practices. *Proceedings of the Western Poultry Disease Conference*; Feb. 27 - Mar. 1; Sacramento, California **43**, 15-19.

Stewart, J.S., 1996. Hatching managementin ostrich production, Chapter 7. In: Tully, T.N. and S.M. Shane, eds. 1996. *Ratite Management, Medicine, and Surgery.* Krieger Publishing Company, Malabar, Florida, 188p.

Tserveni-gousi, A.S. and A.L. Yannakopoulos, 1995. Effect of hen age at sexual maturity on egg shape in pullets. *Poultry Science* **74**:279-284.

Tullett, S.G., 1990. Science and the art of incubation. *Poultry Science* **69**:1-15.

Ullrey, D.E., 1982. Unpublished. In: Ullrey, D.E. and M.E. Allen 1996. Nutrition and feeding of Ostriches. *Animal Feed Science Technology* **59**:27-36.

Ullrey, D.E. and M.E. Allen, 1996. Nutrition and feeding of Ostriches. *Animal Feed Science Technology* **59**:27-36.

Vleck D., C. Vleck, and D. Hoyt, 1980. Metabolism of avian embryos: ontogeny of oxygen consumption in the rhea and emu. *Physiological Zoology* **53**(2):125-135.

Vohra, P., 1992. Information on ostrich nutritional needs is still limited. *Feedstuffs* July 13:16.

Wade, John, 1992. Ratite Pediatric Medicine. *Proceedings of the Association of Avian Veterinarians* 340-353.

Waterhouse,H.N. and R. Haley, 1997. Emu guidelines can be made, more research needed. *Feedstuffs* **16**(18):13-14,16.

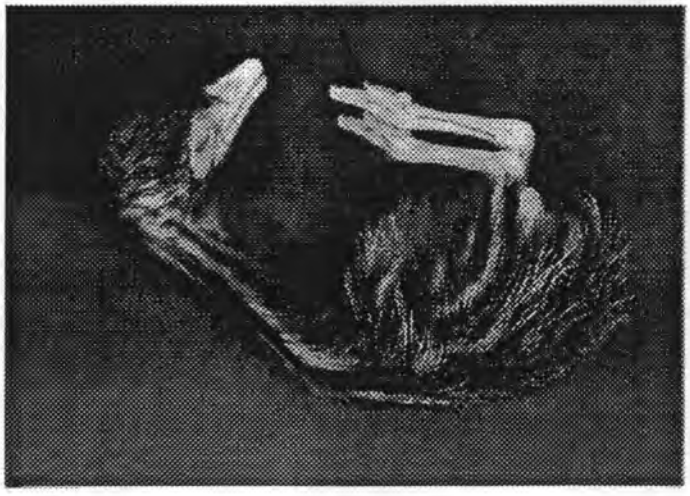
Withers, P., 1983. Energy, Water, and Solute Balance of the Ostrich, *Struthia Camelus. Physiological Zoology* **56** (4): 568-579.

APPENDIX

EMBRYONIC DEVELOPMENT OF THE EMU
(Dromaius novaehollandiae)

by
Carol Allen

Photos by James C. Hermes, PhD, Animal Sciences Department, Oregon State University



Learn to identify infertility and excessive mortality problems during artificial incubation of emu eggs.

Farming of emu has recently progressed from a breeder market to a slaughter market. For the emu industry to be competitive in marketing their products, it is important to have a maximum hatch of the total number of eggs laid. If you are an emu farmer, you must be able to distinguish fertile eggs and normal developing embryos from infertile eggs and early dead embryos during artificial incubation.

During incubation, development can be monitored using an infra-red candler. Candling should be done at one and two weeks of incubation and again at transfer of the eggs into a hatcher. After candling and at the end of incubation, all eggs not hatched

should be broken open to determine fertility, maximum stage of embryo development at time of death, or identify any abnormalities. These procedures will be helpful in identifying probable causes of consistent low numbers of hatched eggs, thus allowing for adjustments to obtain a maximum hatch.

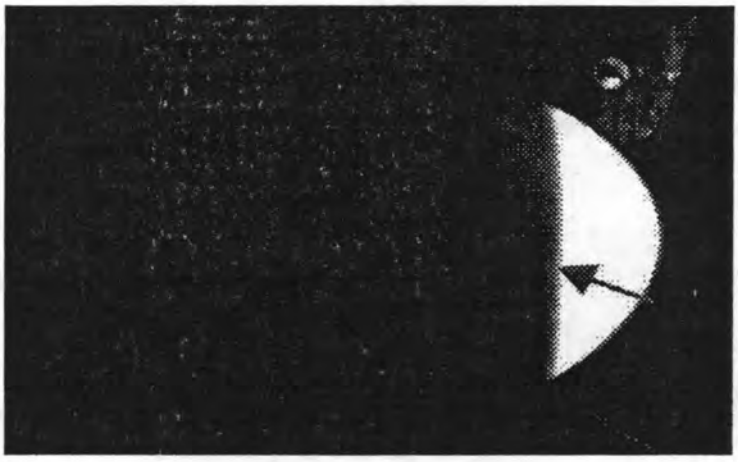
Possible results of candling and breakout of eggs

- Infertile (inside of egg appears clear when candling)
- Positive development (inside of egg appears clear when candling the egg)
- Blastoderm without embryo (inside of egg appears clear when candling the egg)
- Fertile / normal development (candling reveals a dark oval shadow during the first two weeks, and during the third candling the inside of the egg will be totally dark except for the air cell)
- Dead embryo: early or middle incubation death and dwarfed embryos (during the third candling the non-air cell part of the egg will not be totally dark)
- Dead embryo: late incubation death includes normal appearing embryos, dwarfed embryos, and deformed embryos (during the third candling the non-air cell part of the egg will be totally dark and no movement observed
- Live normal embryo (same as dead embryo - late incubation death except movement can usually be observed at the edge of the air cell

Samples of observations made while candling emu eggs during artificial incubation (all eggs pictured here were incubated horizontally)

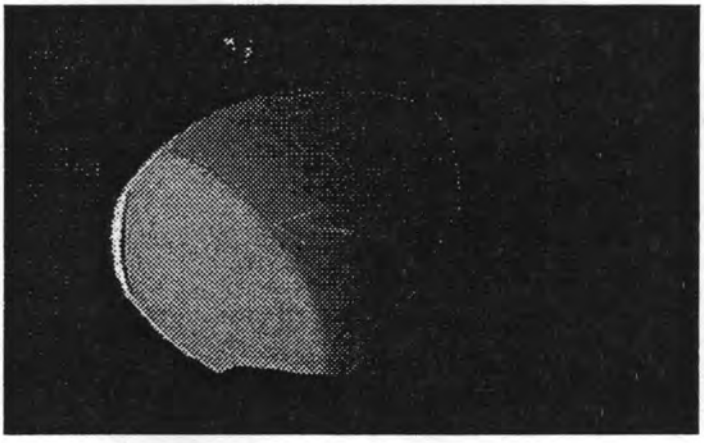
Day 10 of incubation

A horizontal line appears along the midline of the egg dividing the egg into a dark side and a grey side. At this stage of incubation, this appearance indicates a live embryo. The dark shadowed area will roll as the egg is turned. This type of phenomenon is not seen when candling other avian species. The air cell is located on the right. It is dark in appearance because the candling light is at the center of the egg on the back side.



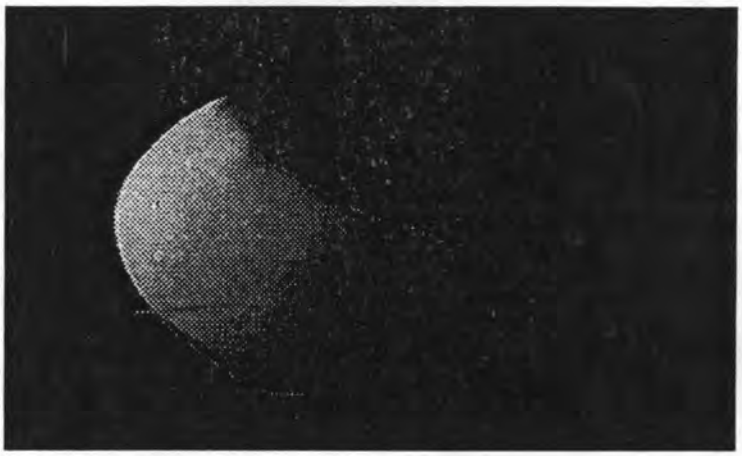
Day 20 of incubation

The air cell is clear with a sharp defined edge (arrow). The remainder of the egg is dark indicating the embryo is alive.



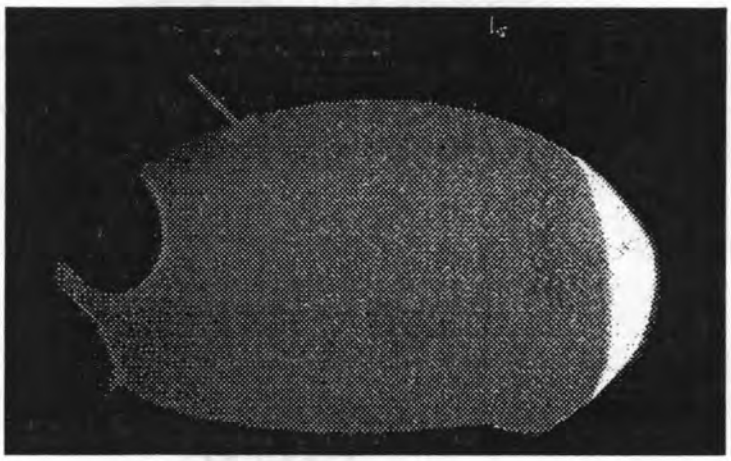
Day 20 of incubation

The air cell is tilted and the edge is not as sharp as in the previous photo. A dark uneven mass appeared to be loosely floating within the grey area as the egg was turned (circled in red). The embryo is probably dead, however the abnormal air cell position could be caused by the horizontal incubation position of the egg.



Day 30 of incubation

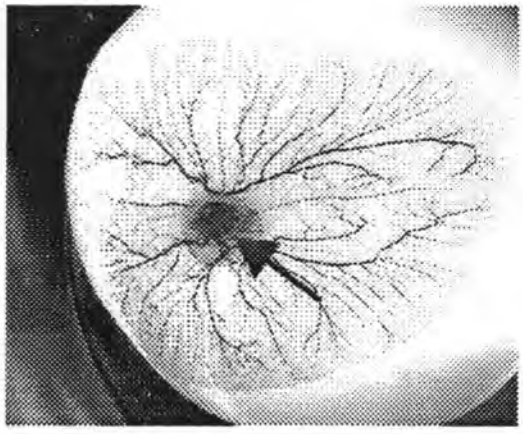
The air cell has a "U" shaped dip to one side. The embryo is alive. All eggs in this study having an air cell with this shape had embryos alive at transfer to the hatcher. The abnormal air cell shape may be due to the horizontal incubation position of the egg.



Any day throughout incubation

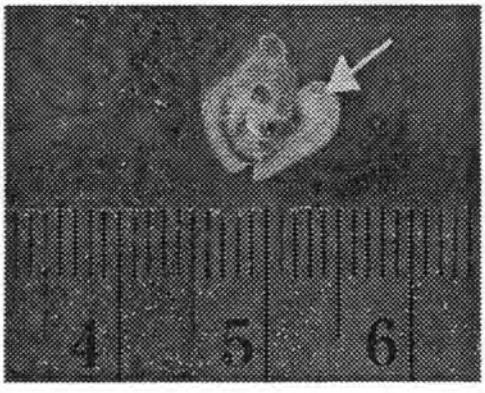
The inside of the shell appears light in color and clear other than the shadow of the yolk. After ten days of incubation, the clear egg can be removed with confidence that it will not hatch. Problems that can result in the clear appearance in the egg interior include infertile, pre-oviposital death, positive development of membranes, and blastoderm without embryo.

Emu embryos and other observations made after breakout at ten day intervals during incubation. Photos include normal and abnormal development of embryos, and contents of eggs that contain no embryos.



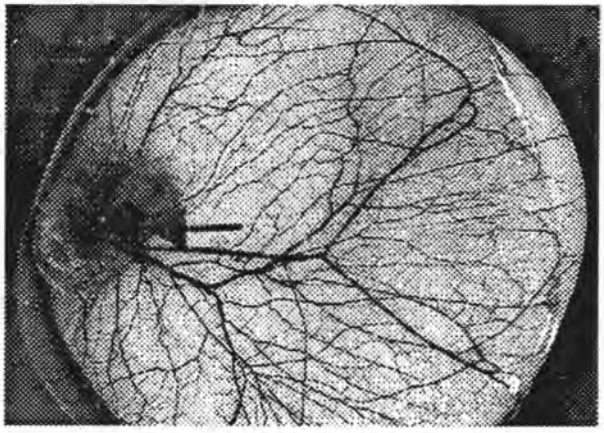
Day 10 of incubation

This is a picture of a normal 10 day embryo. It appears to be at Stage 21 (3.5-4 days) in the chicken embryo development classification (Hamburger and Hamilton, 1951). The eye of the embryo is the small dark spot indicated by the arrow. The point of the arrow ends at the top of the embryo's head. The eye pigmentation at this stage is faint or grayish. The external circulatory system is well developed in the CAM membrane and will continue to grow encompassing the exterior of the yolk through incubation.



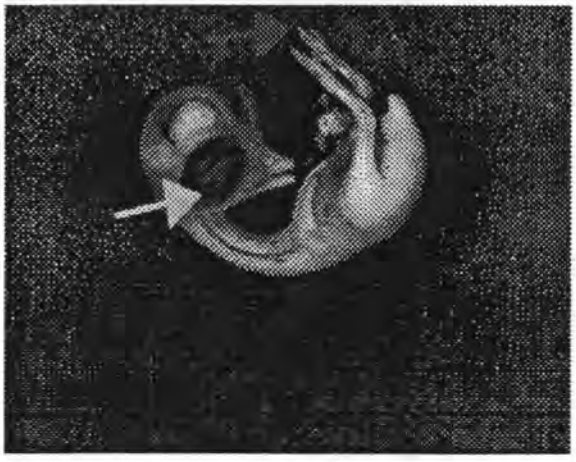
Day 10 of incubation

This is a picture of the previous embryo, but removed from the yolk. The embryo at ten days appears mostly translucent. Wing buds and leg buds (arrows) are present and slightly asymmetrical. The dorsal contour of the trunk is still straight.



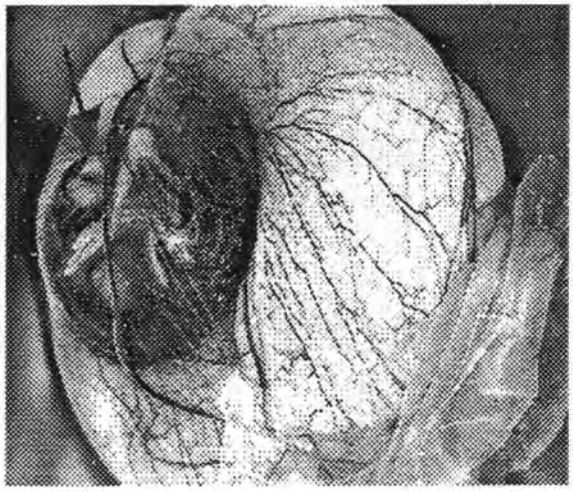
Day 20 of incubation

This photo of a normal 20 day emu embryo shows the extended development of the CAM with its well developed circulatory system surrounding the yolk. The eye is darker and more prominent than on day ten.



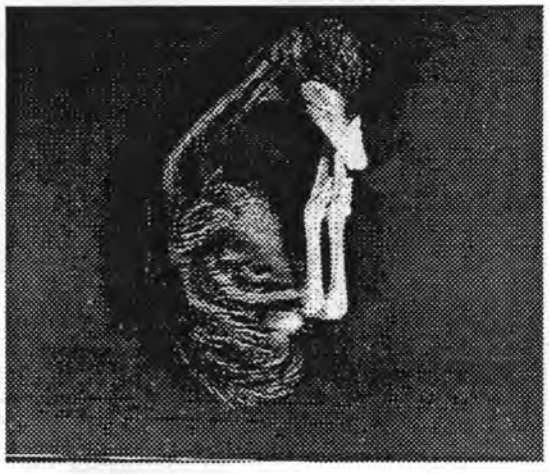
Day 20 of incubation

At day 20 of incubation, the normal emu embryo is approximately at Stage 35 (8-9 days) in the chicken embryo. The beak has become long and narrow. The wings and legs also have become long and narrow. The toes have lengthened and become separated (arrow). Feather tracks are beginning to appear. The eye appears ellipsoid, the growing nictitating membrane is approaching the scleral papillae, the small light dots arranged in a circle on the eye (arrow). At this stage, there are 18 of these dots on the eye.



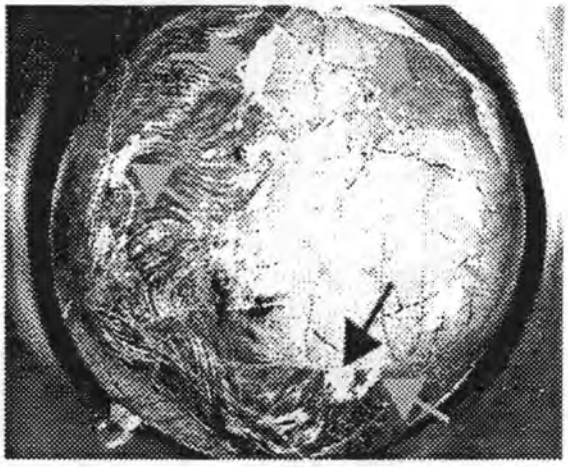
Day 30 of incubation

The 30 day emu embryo is at about the same stage development as the 12 day chicken embryo. The eye opening is ellipse and the lids have covered approximately 3/4 of the eye (arrow). Feathers are well developed and cover the entire body.



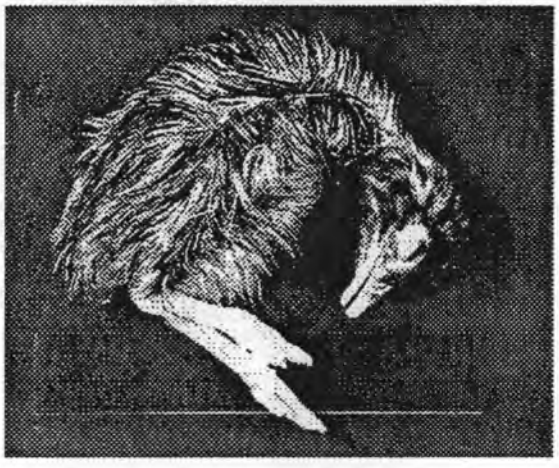
Day 30 of incubation

The eye of this embryo was still opened at breakout (refer to the top photo), but closed after the embryo was removed from the yolk. The eye is normally still opened at this stage. The extent of the feather development can be easily seen. The beak and the claws on the toes (arrow) and wing have become calcified.



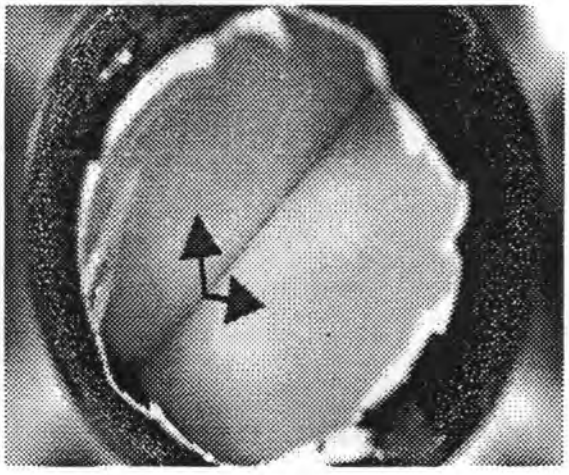
Day 40 of incubation

By day 40, the emu embryo is approximately at Stage 40-43 (14-18 days) in the chicken embryo. The eyes are now closed (red arrow). The hemorrhagic appearance in this photo is due to blood vessels breaking when removing the yolk from the shell.



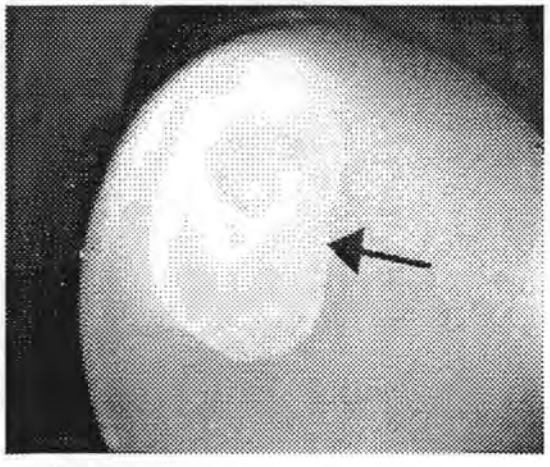
Day 40 of incubation

The 40 day emu embryo physically appears fully developed. The feathering is dense and scales are very apparent on the legs and toes. The eyes are completely closed. From this stage to hatch, not much change is visible. Change in development of the chicken embryo from this stage is based on the length of the toe and beak. Reliability of this measurement for recording change in the emu embryo is still unknown.



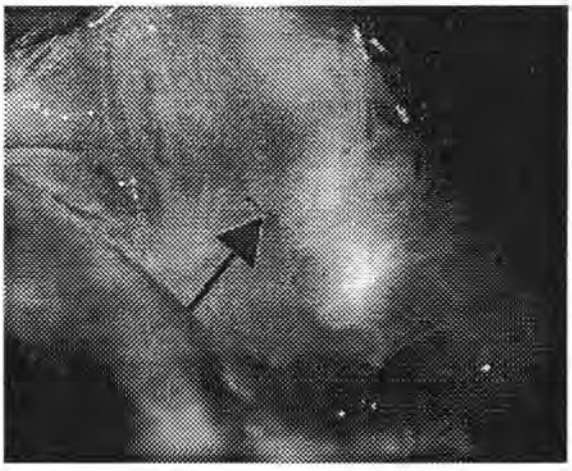
Double yolked egg

This egg contained a double yolk. Both yolks were determined infertile. Although rare, double yolk eggs have been found to be fertile. Twin embryos have been found as early deads in the eggshell of chickens and have been reported by farmers in emu eggs.



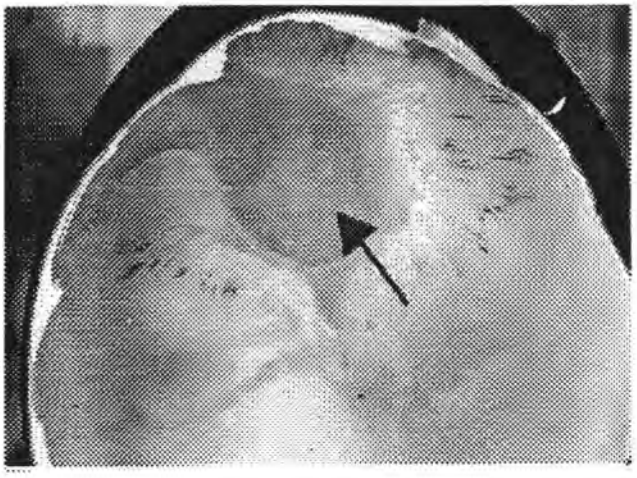
Positive development

This is a fertile egg with some membrane development (arrow) occurring after very early death of the embryo. There is no obvious embryo present. This condition can appear if eggs are roughly handled or held too long before incubating.



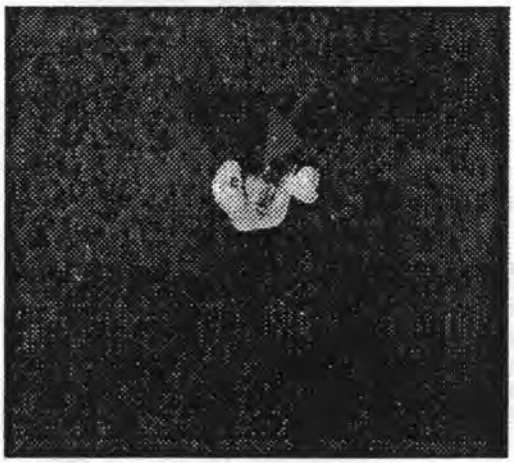
Blastoderm without embryo (BWE)

Characteristics of a BWE are some membrane development similar to a positive development, but having a blood ring present (arrow). Some blood rings are more pronounced than others. The egg is fertile, but there is no embryo present. These are rare, but can occur due to eggs being held too long or at improper temperatures before incubation. Rough handling is another probable cause.



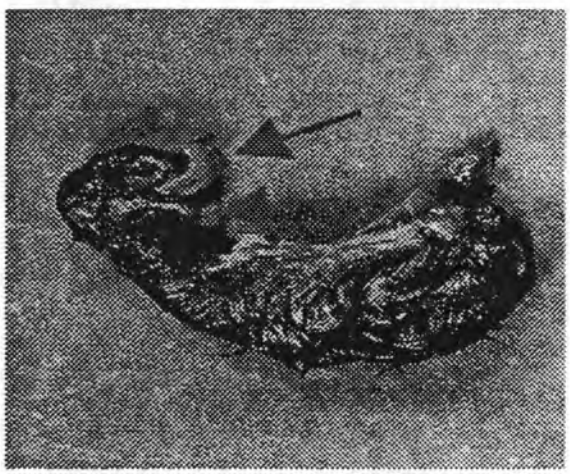
Cystic embryo

The embryo indicated by the arrow had died very early in development. Membrane development has occurred with some formation of blood. Causes for this condition can be similar to those previously mentioned. This is a rare condition.



An early dead embryo

This is a very small embryo and can be easily missed if eggs are blown out rather than broken open and examined. Comparing this embryo to the ten day embryo pictured previously, this embryo is more advanced, but extremely dwarfed in size relative to its developmental stage. The heart is exterior (arrow).



Embryo with a Donald Duck beak

Embryos having a Donald Duck beak are rare. The upper beak is curled upward and back while the lower beak is reduced and curled under as indicated by the arrows. These embryos are usually dwarfed in size. Donald Duck beak is a lethal characteristic, however there has been at least one incidence of a hatched chicken showing this abnormality (Landauer, 1961). The cause can be either genetic or nutritional.