



UF/IFAS Range Cattle Research and Education Center

– FIELD DAY –

April 9, 2015



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Schedule

- 8:00 a.m.** **Check-in / Registration & Sponsor's Booths Open**
- 9:00** **Welcome (inside the Grazinglands Education Building)**
Dr. John Arthington, Professor and Center Director
- 9:05** **Opening Remarks**
*Dr. Jack Payne
UF-IFAS Senior Vice President of Agriculture and Natural Resources*
- 9:10** **Break (5 minutes)**
Attendees split into two groups, with half boarding wagons to travel to field sites. Both groups will see all the presentations, just at different times.
- 9:15** **Fetal Programming in Livestock**
Dr. Phillip Lancaster, Assistant Professor
- Replacement Heifer Economics**
Chris Prevatt, Regional Specialized Agent II
- Pasture Selenium Application - Impacts on Selenium Status of Forage-fed Cattle**
Dr. John Arthington, Professor and Center Director
- Control of Perennial Grasses**
Dr. Brent Sellers, Associate Professor and Associate Center Director
- The Environmental and Economic Cost of Wild Hogs**
Dr. Raoul Boughton, Assistant Professor
- 12:15 p.m.** **Sponsor's Booths Open (12:15 – 3:00 p.m.)**
Lunch - under the tent
- 1:15** **Graduate Student Program (inside the Grazinglands Education Building)**
- | | | |
|-----------------------|------------------------|----------------------|
| <i>Wes Anderson</i> | <i>Julie Burford</i> | <i>Connor Crank</i> |
| <i>Cody Lastinger</i> | <i>Juliana Ranches</i> | <i>JK Yarborough</i> |
| <i>Paul Vining</i> | <i>Ke Zhang</i> | |
- 3:00** **Adjourn**

Welcome to Ona!

Established in 1941, the UF/IFAS Range Cattle Research and Education Center has a long history of service to Florida's cattle and land managers and a promising future ahead. Our mission is to provide science-based information to address the challenges affecting owners and managers of grazinglands. Through efforts centered on the enhancement of livestock, forages, and natural resources, our faculty programs, together with support staff, are dedicated to conducting beneficial research, offering engaging extension programs, and educating graduate students – tomorrow's science leaders. Situated on 2,840 acres in SW Hardee County, our faculty programs focus on beef cattle nutrition and management, economics, forages, soil fertility, pasture and rangeland weed management, and rangeland ecosystems and wildlife.

As you will see today, a great deal has happened at the Center since our last field day in October 2013. During some of our presentations today you will enjoy a first at the Center, air-conditioning, as we utilize our newly constructed Mosaic Grazinglands Classroom inside the Grazinglands Education Building. We are very grateful for the generous gifts from The Mosaic Company and The Florida Cattleman's Foundation that made this building possible. In many ways 2014 was busy and fruitful year. In addition to adding two new faculty programs, Dr. Raoul Boughton and Chris Prevatt, RCREC faculty generated 23 refereed publications, 36 extension documents (EDIS), graduated 4 students, and hosted 11 international scholars and interns.

We value your support as our clients and partners. We realize that you face new challenges every day in cattle and forage management. It is our goal to continue to earn your trust as we work together to address your challenges and create a bright future for Florida cattlemen.

We thank you for coming and hope you enjoy your visit. We invite you to participate in other activities involving faculty from the Range Cattle Research and Education Center. You can find more information on our website, <http://rcrec-ona.ifas.ufl.edu/> or like us on Facebook. You may also feel free to contact us anytime at ona@ifas.ufl.edu or 863-735-1314.

The RCREC Faculty

John Arthington
Raoul Boughton
Phillip Lancaster
Chris Prevatt
Brent Sellers
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Fetal Programming in Livestock

Phillip Lancaster

In the 1990s, Dr. David Barker studied the incidence of chronic disease in people conceived or born in the Netherlands during the Hunger Winter of 1944-1945. During this period, a German blockade cut off food shipments from farm areas resulting in food consumption of 500-1000 calories per day. Because of the reduced nutrient intake, the fetuses of gestating women were exposed to severe malnutrition in-utero. Dr. Barker observed that people conceived or born in the blockade region had a higher incidence of chronic disease such as impaired glucose tolerance, high cholesterol, high blood pressure, cardiovascular disease, and obesity as adults than people born in other regions of the country. This led to the hypothesis that nutritional stress, and we now know other stressors, during gestation can cause long-term changes in offspring resulting in altered metabolic function later in life. This is called fetal programming, and it has been widely studied in both humans and animals during recent years.

To understand fetal programming, it is important to understand the difference between genotype and phenotype. The genotype describes the DNA an animal inherits from its parents and the phenotype results from the expression of the genes encoded in the DNA. In fetal programming, the intrauterine environment experienced by the fetus alters the phenotype, but the genotype does not change.

In addition to DNA, which is the primary structure of a gene, gene expression is also influenced by the secondary structure of the gene. Changes in secondary structure are called epigenetic changes. Epigenetic changes affect the amount of the enzyme or hormone that is made from the gene, which in turn affects performance of the animal. These changes allow an animal to adapt to its environment even after the genotype has been determined. Recent research indicates that epigenetic changes potentially play a large role in traits that are controlled by multiple genes like feed intake, average daily gain, ribeye area, and marbling score to name a few.

Several different stressors can impact fetal development and programming of performance later in life; malnutrition, heat stress, and immune challenge. It has been known for some time that severe nutrient restriction of the dam during late gestation can result in low birth weight calves and lambs, but only recently has research looked at more long term effects on the performance of those offspring. Calves and lambs with low birth weight have slower pre-weaning growth rate and lower weaning weights than offspring with normal birth weight. This is a consequence of changes in muscle metabolism that result in slower rates of muscle growth and more nutrients being directed toward fat deposition, which ultimately impacts carcass quality even at the same body weight. Lambs with low birth weight have been shown to use feed energy less efficiently for muscle growth and fat deposition, and have altered reproductive performance such as lower blood progesterone concentrations during estrous and reduced formation of a corpus luteum, which could negatively impact fertility. Overall low birth weight

resulting from severe nutrient restriction of the dam during late gestation negatively affects several aspects of livestock production.

In normal beef cattle production systems, nutrient restriction of the dam severe enough to adversely affect birth weight of the offspring is uncommon. However, recent research in livestock species has found that moderate maternal nutrient restriction during gestation can negatively impact growth, carcass quality, reproductive performance, milk production, and feed efficiency in offspring. The effect of maternal nutrient restriction on offspring performance depends upon stage of gestation, because the fetal tissues developing during the affected stage of gestation determine which traits may be impacted. In early gestation, fetal growth consists primarily of vital organ development, which could affect basal metabolism and function of many metabolic processes. In mid-gestation, primary muscle fibers are developing, which could affect muscling, composition of gain, average daily gain and feed efficiency. Fat tissue and reproductive organs develop in late gestation, thus there is an effect on marbling score and heifer fertility.

In the Great Plains and Western US, beef cows in late gestation may graze dormant winter native prairie that is low in protein, which does not negatively impact birth weight of calves. Until recently it was thought that beef cows could adapt to the lower plane of nutrition due to lower nutrient requirements during this time of the production cycle. However, recent studies at the University of Nebraska and University of Wyoming have changed that theory. Protein supplementation of dams grazing dormant native prairie increased weaning weight of offspring even though birth weight was not affected. Additionally, heifer offspring from protein-supplemented dams had greater pregnancy rates than those from non-supplemented dams, but there was no difference in pregnancy rates when rebred as first-calf cows. In the feedlot, fewer steers from protein supplemented dams were treated for respiratory and gastrointestinal disease, but rate of gain and feed efficiency were not affected. Protein supplementation of dams increased carcass weight, back fat thickness, marbling score, and tenderness of rib eye muscle of steer offspring compared with steers from non-supplemented dams. These reports demonstrate that even moderate nutrient restriction of pregnant cows can negatively impact performance of their offspring.

Other research indicates that energy and protein restriction of dams during mid-gestation result in slower rate of gain of steer offspring in the feedlot, less rib fat thickness, and tougher steaks. Additionally, energy and protein restriction of dams during mid-gestation resulted in increased internal fat and less muscle weight in lamb offspring. In early gestation, energy and protein restriction of dams caused reduced milk production of ewe offspring during their first lactation. Also, dairy cows fed diets deficient in methionine during the pre-conception period had altered embryo development; however, no data on offspring performance was collected in this study.

In South Florida, the critical nutritional period is during the pre-conception and early gestation periods for cow herds calving in the fall. There is very little data on the effect of maternal nutrition during the pre-conception and early gestation periods on fetal development and

programming of offspring performance, and no data in beef cows in South Florida. Thus, a current focus of my research program is to evaluate maternal nutrition during this critical time period. Currently, a study is underway to evaluate the effect of maternal protein or methionine addition to molasses supplements on growth, and energy and protein metabolism of offspring.

Over feeding pregnant dams may also have negative effects on performance of offspring. Feeding pregnant ewes at 140% of nutrient requirements resulted in lower birth weight of lambs compared with lambs from dams fed at 100% of nutrient requirements. Ewe offspring from overfed dams had lower milk production during their first lactation than those from dams fed 100% of nutrient requirements; the milk production of ewes from overfed dams was the same as ewes from nutrient-restricted dams. Lambs from overfed dams had similar growth rate as lambs from dams fed at 100% of nutrient requirements, but both had faster growth rates than lambs from nutrient-restricted dams. Overall, research results indicate that overfeeding dams during gestation does not increase performance of offspring compared with offspring from dams fed to meet nutrient requirements, and in some situations may have negative effects.

Heat stress has a significant impact on milk production of dairy cattle, particularly in the southeastern US, and thus has been studied extensively in dairy cattle. Several studies demonstrated that heat stress during mid to late gestation decreased calf birth weight in dairy cattle. These studies demonstrated that providing some type of cooling, even natural shade in open pasture, resulted in greater calf birth weight than calves from cows provided no cooling. Interestingly, more recent studies have found that calves from heat stressed cows can have reduced body weight up to one year of age. Also, calves from heat stressed cows have decreased ability to absorb antibodies from colostrum resulting in increased calf morbidity and mortality compared with calves from cows provided some type of cooling. Heifer calves from heat stressed cows have lower milk production during their first lactation as well. Research in dairy cattle suggests that heat stress in pregnant beef cows may have negative effects on offspring performance, but no studies have been conducted in beef cows. Also, there is no information on the effect of *Bos indicus*-influenced breeds with regard to the effects of heat stress on offspring performance. Adaptation of *Bos indicus* breeds to high ambient temperatures is through dissipation of heat by increasing blood flow to the skin suggesting that blood flow and nutrients to the fetus may be reduced possibly having a negative impact on fetal development in *Bos indicus* breeds relative to *Bos taurus* breeds. Research is needed to evaluate the effects of breed and heat stress on offspring performance in beef cows in South Florida.

An induced immune challenge of the pregnant dam has been shown to improve the immune response of offspring in other species. However, no studies have been conducted in cattle or any livestock species. Therefore, a study was conducted at the Range Cattle REC to evaluate the effect of maternal immune challenge of beef cows during late gestation on fetal programming of the immune response in subsequent offspring. Cows received either an injection of bacterial toxin or saline at approximately 230 days of gestation. Cows receiving the bacterial toxin had an increase in body temperature of about 1°F for 6 hours post injection compared with the cows

given saline indicating that the cows did mount an immune response to the bacterial toxin. There was no difference in birth weight of calves from cows given bacterial toxin or saline, but calves from cows given bacterial toxin weighed 30 lb more at weaning when adjusted for age of calf. The heifer calves from these cows were evaluated for their immune response to the same bacterial toxin. The heifer calves from cows given bacterial toxin had a more subtle increase in body temperature and markers of immune response when challenged with the toxin compared with heifer calves from cows given saline, indicating that heifers previously exposed to the toxin were able to fight off the infection with a less severe reaction. Previous research indicates that a severe immune response redirects nutrients from muscle growth to the immune system. Therefore, we believe that the calves from cows given bacterial toxin had increased weaning weight because of a greater ability to fight off infection without redirecting as many nutrients away from muscle growth. A second study is being conducted to further evaluate the effect of maternal immune challenge on growth and immune response of the offspring.

In conclusion, research indicates that several stressors during gestation could impact performance of the offspring, with the impact of nutritional stress being among the best understood. Improper maternal nutrition can negatively impact several aspects of offspring performance. Therefore, the best recommendation at the present time is to manage the nutrition of the cow herd to meet nutrient requirements throughout the beef cow production cycle. Please refer to "*Basic Nutrient Requirements of Beef Cows*" (<http://edis.ifas.ufl.edu/an190>) for more information on managing the nutrition of the cow herd. Regarding heat stress, it is recommended that pregnant beef cows in South Florida be provided shade to help alleviate any possible negative effects of heat stress on performance of subsequent offspring. Pastures should be designed to provide natural shade from large trees, and there should be enough space available for all cows to comfortably rest in the shade during the heat of the day. If no natural shade is available, then man-made shade structures may be used. Management practices regarding maternal nutrition and heat stress focus on preventing a negative outcome. Maternal immune challenge is thus far the only management practice focused on increasing the performance of offspring, but there is still much to learn about this response as there has been only one study to date. Additionally, bacterial infection in pregnant beef cows can cause abortion. Thus, it is not recommended that producers induce a bacterial infection in pregnant beef cows at this time.

Replacement Heifer Economics: The Cost of Raising Beef Replacement Heifers

Chris Prevatt

The cost analysis presented in this paper focuses on the cost of raising beef replacement heifers. Calculating the cost of a raised beef replacement heifer seems simple. Most producers include the production costs of heifer development and the value of the weaned heifer calf. However, the situation is a little more complicated than it seems. There are an infinite number of replacement heifer development programs and levels of management which all have different levels of success and associated costs of getting a non-pregnant heifer pregnant. In addition, there are at least two adjustments that should be included in the analysis; 1) the gain or loss on open replacement heifers that are culled and the loss of those that die and 2) an adjustment for the reduced inventory of brood cows when raising beef replacement heifers since this decision decreases the total number of brood cows that the ranch can support. It is important to include these two adjustments to correctly calculate the total cost of raising beef replacement heifers.

The costs associated with raising a replacement heifer can be large. The example in Table 1 provides an estimated cost of a raised beef replacement heifer in Florida during 2015. The estimates are expressed on a per-heifer basis as well as the cost of raising 100 beef replacement heifers.

The example presented in Table 1 represents only one of an infinite number of development strategies and levels of management to raise beef replacement heifers. The budget in Table 1 is based on developing and breeding 100 weaned heifer calves. It was assumed that 84 of the 100 heifers (84%) became pregnant, 15 animals (15%) were culled, and 1 heifer died (1%). The variable and fixed costs for the beef replacement heifer program were \$1,874.13 and \$318.67 per heifer, respectively. The total variable and fixed costs were \$2,193 per heifer assuming all heifers became pregnant and no death loss occurred. The adjustment for non-breeders and death loss was -\$148 per heifer and the adjustment for the reduced inventory of brood cows was -\$133 per heifer. The resulting total cost of a raised beef replacement heifer in this budget was \$2,474 per heifer.

Please note that the cost to raise beef replacement heifers varies considerably between producers. Estimated costs for a given ranch may be higher or lower than those presented in the example budget because of location, resources, heifer development strategy, level of management, inputs, and conception rate. Whenever possible, producers should make adjustments and use their own production and financial information to determine their cost of raising replacement heifers. However, the example budget will provide a template to follow when estimating the cost of developing a raised beef replacement heifer.

Table 1. Estimated Costs of Raised Beef Replacement Heifers, Florida 2015

Item	Description	Unit	Quantity	\$/Unit	\$/Heifer	\$/100 Heifers
Estimated Variable Costs						
Heifer Calf	Value of Weaned Heifer	lbs.	525.00	\$2.45	\$1,286.25	\$128,625.00
Land Rent	Improved Perennial Pasture	acre	2.00	\$25.00	\$50.00	\$5,000.00
Pasture Costs	Fertilizer, Misc. Costs	acre	2.00	\$50.00	\$100.00	\$10,000.00
Mixed Hay	53% TDN, 7% CP	tons	0.90	\$80.00	\$72.00	\$7,200.00
Energy Supplement	72% TDN, 16% CP	tons	0.26	\$225.00	\$59.06	\$5,906.25
Protein Supplement	75% TDN, 46% CP	tons	0.11	\$175.00	\$19.69	\$1,968.75
Mineral and Salt		tons	0.04	\$650.00	\$23.73	\$2,372.50
Breeding Cost	Cost of Bull Service	head	1.00	\$60.00	\$60.00	\$6,000.00
Mach. & Equip.	Fuel, Oil, Lube, etc.	head	1.00	\$25.50	\$25.50	\$2,550.00
Vet. and Med.	Preg Check, Vac., Supplies	head	1.00	\$23.50	\$23.50	\$2,350.00
Labor	(Wages & Fringe)	hours	3.00	\$12.50	\$37.50	\$3,750.00
Misc. Expenses		head	1.00	\$15.00	\$15.00	\$1,500.00
Interest on ½ of Operating Costs		dollars	\$485.98	5.75%	\$27.94	\$2,794.36
Interest on Heifer Calf		dollars	\$1,286.25	5.75%	\$73.96	\$7,395.94
Total Variable Costs					\$1,874.13	\$187,412.79
Estimated Fixed Costs						
General Overhead		dollars	\$1,874.13	9%	\$168.67	\$16,867.15
Management Fee		head	1.00	\$150.00	\$150.00	\$15,000.00
Total Fixed Costs					\$318.67	\$31,867.15
Total Variable and Fixed Costs					\$2,193	\$219,280
Gain/Loss Adjustment For Non-Breeders*					(\$148)	(\$14,832)
Adjustment for Reduced Brood Cow Inventory**					(\$133)	(\$2,667)
Total Cost of Raised Replacement Heifer					\$2,474	\$247,444.97

University of Florida, IFAS, Range Cattle Research and Education Center

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*The gain/loss adjustment for non-breeders is the market value of non-breeders minus their total variable and fixed costs.

**The adjustment for reduced brood cow inventory is the reduced profit resulting from a lower level of brood cow inventory associated with raising replacement heifers.

Sensitivity of Total Cost of a Raised Beef Replacement Heifer

The total cost of a raised beef replacement heifer is very sensitive to the production cost level (variable and fixed costs per heifer) and the percent of exposed heifers confirmed pregnant. Table 2 shows the estimated total cost of a raised replacement heifer based on various production cost levels and percent of exposed heifers confirmed pregnant. The production cost levels in Table 2 range from \$1,800 to \$2,600 per heifer and the percent of exposed heifers confirmed pregnant ranges from 60 to 90 percent. The estimated total cost of raising a pregnant replacement heifer ranged from \$1,850 (associated with a production cost level of \$1,800 and 90 percent of exposed heifers confirmed pregnant) to \$3,106 (associated with a production cost level of \$2,600 and 60 percent of exposed heifers confirmed pregnant).

Table 2. Estimated Total Cost of a Raised Replacement Heifer Based on Various Cost Levels and Percent of Heifers Confirmed as Bred

<i>Variable and Fixed Costs of Raised Repl. Hfr.</i>	Percent of Exposed Heifers Confirmed as Bred						
	90%	85%	80%	75%	70%	65%	60%
	(Estimated Total Cost of a Raised Replacement Heifer, \$/hd.)						
\$2,600	\$2,739	\$2,800	\$2,861	\$2,922	\$2,983	\$3,044	\$3,106
\$2,500	\$2,628	\$2,683	\$2,739	\$2,794	\$2,850	\$2,906	\$2,961
\$2,400	\$2,517	\$2,567	\$2,617	\$2,667	\$2,717	\$2,767	\$2,817
\$2,300	\$2,406	\$2,450	\$2,494	\$2,539	\$2,583	\$2,628	\$2,672
\$2,200	\$2,294	\$2,333	\$2,372	\$2,411	\$2,450	\$2,489	\$2,528
\$2,100	\$2,183	\$2,217	\$2,250	\$2,283	\$2,317	\$2,350	\$2,383
\$2,000	\$2,072	\$2,100	\$2,128	\$2,156	\$2,183	\$2,211	\$2,239
\$1,900	\$1,961	\$1,983	\$2,006	\$2,028	\$2,050	\$2,072	\$2,094
\$1,800	\$1,850	\$1,867	\$1,883	\$1,900	\$1,917	\$1,933	\$1,950

*Assumes 1% death loss and a market value of \$1,500 per head for culled open heifers.

In Table 2, a \$100 increase in the production cost level increases the total cost of a raised replacement heifer between \$111 (assumes 90% confirmed pregnant) and \$145 per heifer (assumes 60% pregnant). A 5% increase in the percent of exposed heifers confirmed pregnant decreases the total cost of a raised replacement heifer between \$17 (assumes a production cost level of \$1,800 per heifer) and \$61 per heifer (assumes a production cost level of \$2,600 per heifer). Thus, the greater the value of a weaned heifer the greater is the cost of open heifers.

Producers should use their own variable and fixed costs and their projected average conception rate when looking to calculate their total cost of a raised pregnant replacement heifer. These two variables have a large impact on the total cost of a raised pregnant replacement heifer. Additional attention to management has been shown to improve the percent of heifers confirmed pregnant as well as lower production costs which can result in a significantly lower cost of a raised beef replacement heifer.

Summary

Beef replacement heifers are a necessary, but costly part of every cow-calf operation. The cost of raising replacement heifer is highly influenced by the value of the heifer calf entering the replacement heifer program, development cost, and pregnancy rate. There are an infinite

number of replacement heifer development programs and levels of management which all have different associated costs and success of getting a non-pregnant heifer pregnant. Producers are encouraged to make adjustments and use their own production and financial information to determine their cost of raising replacement heifers.

Selenium Biofortification of Pasture Forage

Juliana Ranches and John Arthington

Introduction

Selenium (Se) is a trace element that is essential in small amount for humans and for animals; however the element is not essential for plants. Humans and animals require Se for the function of a large number of Se-dependent enzymes, called selenoproteins (Kryukov et al., 2003). The range of selenoproteins is between 25 and 38 and varies according species. For humans and cattle, there are 25 selenoproteins, but only half of them have their metabolic functions identified. The most widely understood selenoprotein is the antioxidant enzyme glutathione peroxidase (GPX). Glutathione peroxidase was the first mammalian protein shown to incorporate selenium in the form of selenocysteine into its catalytic site and was assumed to be associated with the antioxidant activity of selenium. The GPX enzyme is also well known for catalyzing the reduction of hydrogen peroxide and organic hydroperoxides, thus protecting cells from oxidative damage (Papp et al., 2007). The lack of Se can lead a large number of negative impacts with the most widely recognized being white muscle disease in calves. Other problems associated with Se deficiency include muscular weakness, reduced weight gain, diarrhea, stillbirths, abortions, retained placenta and diminished fertility.

The benefits promoted by Se supplementation have been shown in different fields of research. From human nutrition, researchers proposed that dietary Se is involved in cancer prevention, immune function, aging, and male reproduction (Kryukov et al., 2003). Studies with dairy calves have shown that feeding dams a supranutritional Se-yeast supplement or adding pharmacological dosages of Na selenite to colostrum both increase serum-IgG concentrations and total serum-IgG content in Se-supplemented calves (Hall et al., 2014). Another study with dairy cows, showed that Se supplementation 1 mo before calving increased blood GPx activity, slightly reduced the prevalence of intramammary infections at calving, and lowered SCC at the time of calving, regardless of Se source (Ceballos-Marquez et al., 2010). Researchers from Oregon State University, working with beef cattle, revealed that short-term exposure of cattle to Se-fertilized forage elevated whole blood Se concentrations and levels were sufficient to maintain adequate concentrations throughout grazing periods when there would be limited access to Se supplements (Hall et al., 2011).

In some regions of the World, such as parts of the western United States, Se toxicity can be a problem. There are two general types of toxicity, acute and chronic. Acute Se toxicity is caused by the consumption, usually in a single feeding, of a sufficient quantity of highly seleniferous plants. The indicator plants include certain species of Astragalus, prince's plume, and some woody asters. This kind of poisoning, produces severe symptoms and death occurs within a few hours. A second form of Se poisoning is the chronic toxicity, there are two different types of chronic poisoning dependent on the chemical form of the ingested selenium. "Blind staggers" occurs when animals ingest water-soluble Se compounds naturally found in accumulator plants. Toxicity from eating plants or grain with protein-bound, insoluble selenium is called "alkali disease." Selenium is the only trace mineral that is sometimes found in toxic concentrations in forages grown in specific regions of the US.

The range between adequate and toxic concentrations is narrower for Se compared to other essential trace minerals; however, in most regions of the country, Se-deficient forage is much more common than cases of Se excess. In a survey of 253 cow/calf operations in 18 US states, over 18% were classified as marginally or severely Se deficient by blood Se parameters (Dargatz adn Ross, 1996). Among the states analyzed, those located in the southeast region had the greatest percentage of operations classified as marginally or severely Se deficient (42.4%). Soils containing less than 0.5 mg/kg total are classified as Se deficient. According to NRC (1983) Se-deficient regions in the U.S. include New England, New York, New Jersey, Delaware, Pennsylvania, Maryland, West Virginia, Florida, Ohio, Indiana, Illinois, Michigan, Wisconsin, Washington State, Oregon, Montana, Arizona, and coastal regions of Virginia, the Carolinas and Georgia (Figures 1 and 2). Although Se is not an essential element in plant nutrition, the consumption of plants and plant products is the primary route by which animals and humans receive their dietary Se in the absence of any special supplementation. While higher plants do not require Se, they readily take it up from their environment and incorporate it into organic compounds using Se assimilation enzymes. The plants are responsible for the conversion of selenate-Se into organic Se compounds, this conversion is believed to occur in the chloroplasts.

Biofortification

One potential method for addressing Se nutrition in grazing cattle is the implementation of pasture Se applications with the intent of increasing plant Se content and thus the Se status of cattle grazing these forages. This strategy is called “biofortification”, and has been utilized in Finland since 1985 (Mäkelä et al., 1993). By definition biofortification is a strategy to increase the nutrient content of food.

Selenium from selenate sources appears to be more available for plant uptake compared to selenite sources (Archer, 1983). In Florida, spraying bermudagrass with sodium (Na) selenate at Se application ranges of 4 to 196 g Se/acre (via Na selenate) resulted in substantial increases in forage Se content by 2 weeks after application, decreasing rapidly by 12 weeks post-application (Table 1; Valle et al., 1993). Feeding forages grown on Se-fertilized hay fields impacts both Se status and performance of grazing cattle. In one study (Hall et al., 2011), weaned Angus-type calves were fed Se-fertilized alfalfa hay over a 7-week period. Alfalfa hay was grown on fields receiving applications of Na selenate in amounts providing 0, 9, 18, or 37 g Se/acre. These application rates resulted in a linear ($R^2 = 0.997$) response for Se application rate and subsequent Se content of alfalfa hay harvested 40 days after Se application (Figure 3; Inset A). In addition, calves consuming these hay treatments (approximately 2.5% body weight (BW) daily) experienced a linear ($R^2 = 0.979$) increase in whole blood Se concentrations as Se application rate (and Se content of hay) increased (Figure 3; Inset B).

In a recent study at the UF/IFAS, Range Cattle REC, we produced a high-Se hay crop by spraying a Jiggs bermudagrass hayfield with Na selenate at a rate of 105 g Se/acre. Selenium content of hay, harvested 8 weeks after Na selenate application, was greater for Se-treated vs. control pastures (7.73 ± 1.81 vs. 0.07 ± 0.04 mg/kg DM; $P < 0.001$). In a subsequent study, this hay crop was fed to weaned calves and Se status was evaluated over a 42-day study.

Calves were stratified by initial BW and randomly assigned to treatments including high-Se hay, low-Se hay + supplemental Na selenite, or No supplemental Se ($n = 14$, 14, and 4 calves, respectively). Calves were housed in drylot pens (2 calves/pen; 7, 7, and 2 pens per treatment). A pair-feeding design was utilized, whereas each pen of high-Se hay calves was paired to a pen of Na selenite - supplemented calves. Calves assigned to the high-Se hay treatment were provided ground, high-Se hay for a 4 hour period each morning. Pen dry matter intake was calculated and total daily Se intake/pen was estimated. Each Na selenite paired pen was then provided the same daily amount of Se via Na selenite hand-mixed into a limit-fed grain supplement. Therefore, each pen of calves receiving high-Se hay had a paired partner pen of calves receiving the same amount of Se via Na selenite. Liver Se concentrations remained unchanged for the negative control calves receiving no supplemental Se over the 42-day feeding period, but they were increased ($P < 0.001$) in calves receiving both high-Se hay and Na selenite treatments. Calves receiving high-Se hay had greater ($P < 0.05$) liver Se concentrations on day 21 and 42 than calves receiving Na selenite (Figure 4). Of notable interest, this difference was attributed only to the paired pens consuming < 3 mg Se daily (Figure 5). From these initial data, we hypothesize that there is a differential availability of Se in forage vs. inorganic sources dependent upon the total daily intake with a critical point of approximately 3 mg/day in beef calves. We are currently examining these data further in both periparturient cows and calves.

Summary

Selenium is the essential trace mineral most commonly found to be deficient among beef cows and calves in Florida. Biofortification of pasture forage with Se appears to be effective in increasing calf Se status. Further investigation is warranted to understand the impacts of this management system on cow and calf productivity and overall system economics.

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Table 1. Average forage Se concentrations (mg/kg; DM basis) at different weeks after spraying with Na selenate.¹

Se application rate, g/acre	Weeks after spraying Na selenate				
	2	4	6	12	18
0	1.4 ^a	0.9 ^a	1.5 ^a	0.5 ^a	0.4 ^a
10	2.9 ^a	2.7 ^a	2.3 ^a	0.7 ^b	0.8 ^b
49	12.8 ^a	6.3 ^b	4.8 ^{bc}	0.5 ^c	0.6 ^b
98	26.1 ^a	15.5 ^b	11.9 ^b	0.8 ^c	1.0 ^b
196	51.5 ^a	28.2 ^b	25.7 ^b	0.7 ^c	0.7 ^b

¹Data adapted from Valle et al. (1993). Means are based on 4 replicates per treatment.

²Means with unlike superscripts within each row differ ($P < 0.05$).

Figure 1: Selenium Soil Content, by county across United States. Data from: U.S Geological Survey.

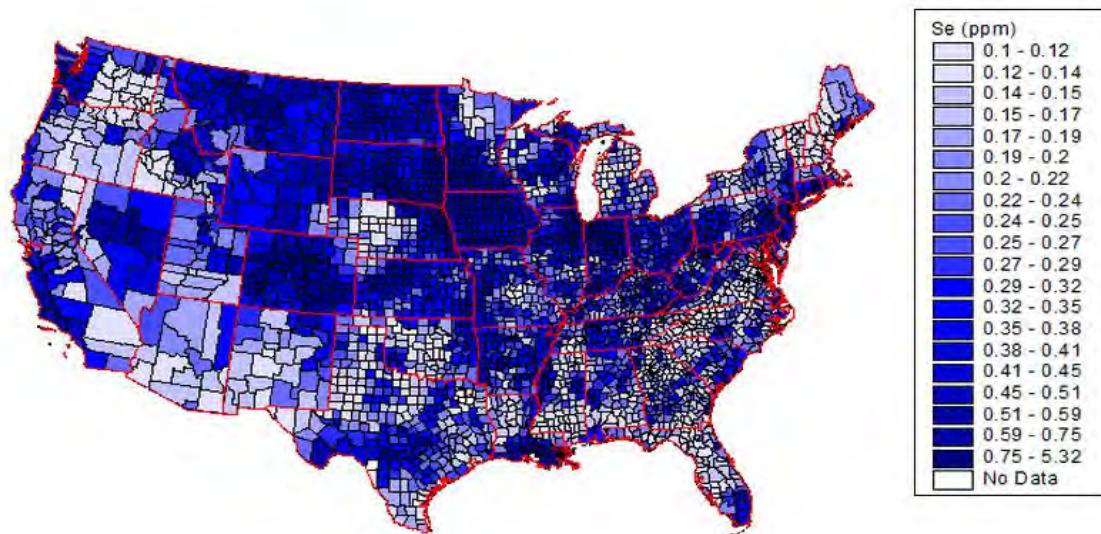
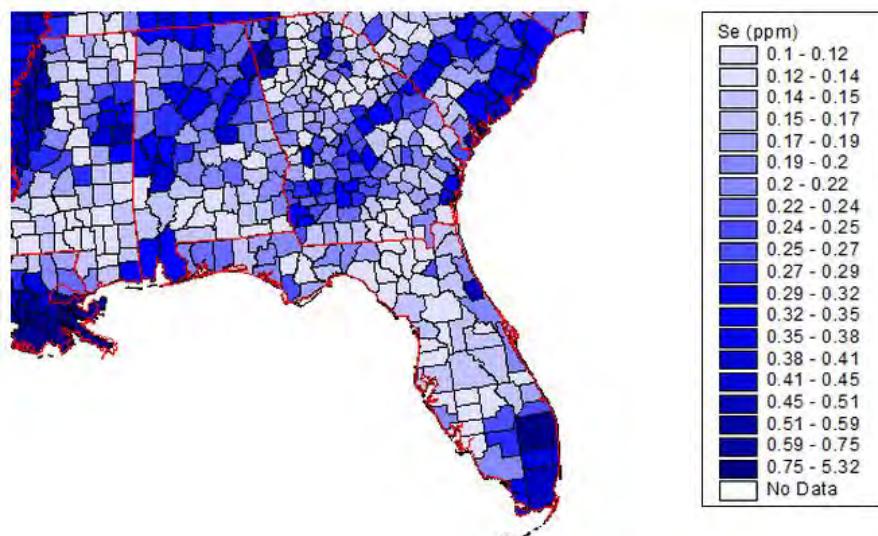


Figure 2: Selenium Soil Content, by county at Florida State. Data from: U.S Geological Survey.



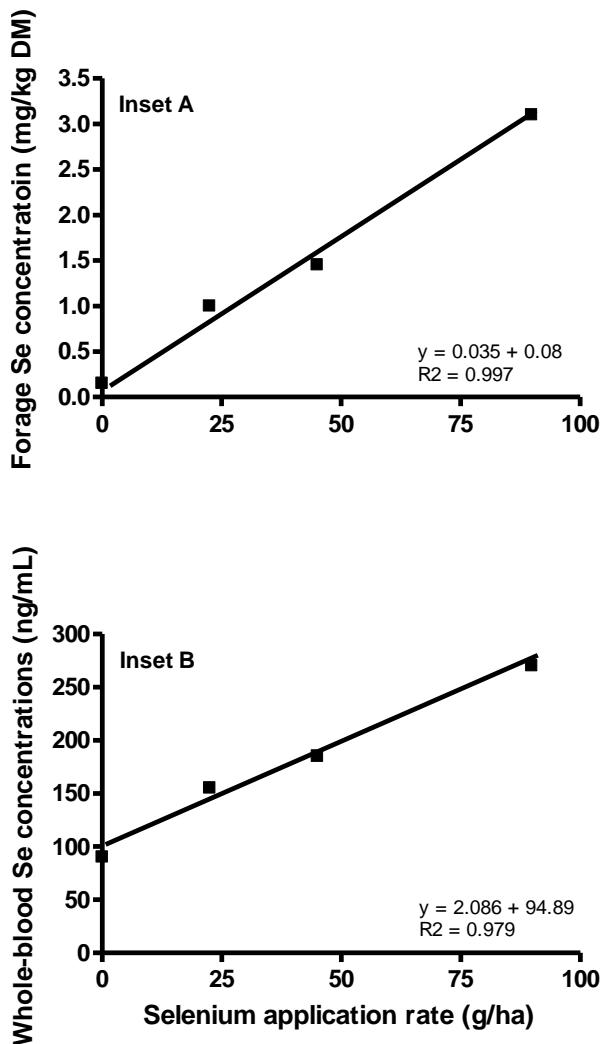


Figure 3. Effects of Na selenate application to alfalfa hay fields on subsequent forage Se content (A) and Se status (B) of calves consuming the hay. Note: g/ha $\div 2.45$ = g/acre.
Data adapted from Hall et al. (2013).

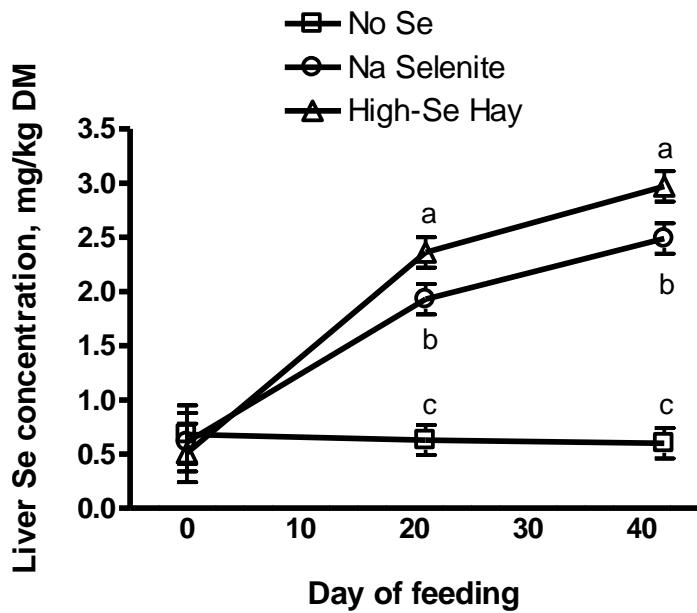


Figure 4. Liver Se concentrations among calves offered high-Se hay or a Na selenite supplement. Basal diet contained 0.6 mg Se daily (No Se treatment). Calves fed high-Se hay and the Na selenite supplement were pair-fed to control overall daily Se intake (ave. 2.8 mg Se/day).
a,b,c Means differ within day; P < 0.05.

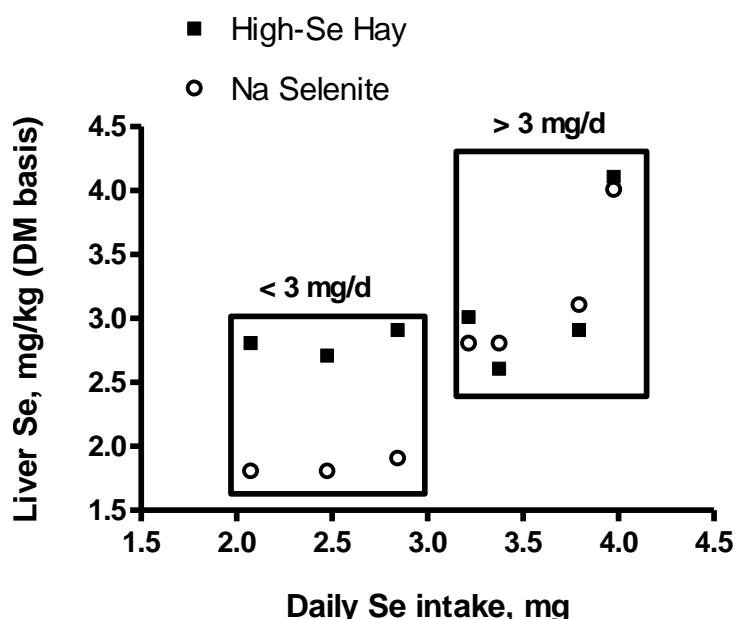


Figure 5. Liver Se concentrations (d 42) among pair-fed calves. X-axis denotes average daily Se intake (mg/day) among each pair-fed calf group.

Control of Perennial Grasses in Pastures

Brent Sellers

Managing perennial grass weeds in improved forage grasses is a difficult task on most cow-calf operations in Florida, and also for those wanting to produce high quality hay. The most challenging perennial grasses in Florida include smutgrass, cogongrass, broomsedge, vaseygrass, guineagrass, and most recently, Indian cupscale. While not every control option is available for each improved grass forage species, there are some options for us to consider.

Smutgrass

This species has been troublesome over the past 60-70 years and about the only things that have changed is the transition from small smutgrass to giant smutgrass as well as the loss of dalapon and the introduction of hexazinone (Velpar, Velossa, etc.) for control. In bahiagrass and bermudagrass pastures, Velpar at a rate of 2 qt/acre should be applied during the rainy season, which is near the maximum allowable rate for use in pastures. Recent research suggests that reduced rates (1 to 1.5 qt/acre) applied annually for 2 years provides similar levels as a single 2 qt/acre application. While these reduced rates work under optimal conditions, a bit more risk is involved in this application strategy. However, even with the 2 qt/acre rate, there is also some risk. For example, rainfall exceeding 3 inches within 2 weeks of hexazinone application often fails to provide optimum control. Limpograss and stargrass are much less tolerant than bahiagrass and bermudagrass, and the label does not indicate that hexazinone can be used in pure stands of these forage species. The research we've conducted in limpograss pastures has indicated that limpograss is not tolerant to the 2 qt/acre application rate, and approximately 30-50% yield loss should be expected from an application of 1 qt/acre.

Pasture renovation should be considered when greater than 80% of the pasture is infested with smutgrass. Spray the entire pasture with 4 qt/acre glyphosate and begin tillage practices no earlier than three weeks after application. Repeated tillage will destroy newly emerged smutgrass and will aid in depleting the soil seedbank. The final seedbed should be a smooth, flat surface devoid of vegetation. For additional information on bahiagrass varieties and seeding rates, see EDIS publications AG342/SS-AGR-332, *Bahiagrass (Paspalum notatum): Overview and Management* (<http://edis.ifas.ufl.edu/ag342>) and AG107/SS-AGR-161, *Forage Planting and Establishment Methods* (<http://edis.ifas.ufl.edu/ag107>). Even with repeated tillage following glyphosate application, smutgrass will likely emerge with bahiagrass, and smutgrass seedheads will be present by the following summer growing season. One year after seeding and during the rainy season, apply 0.5 lb/acre hexazinone (Velpar at 32 oz/acre or Velossa at 27 oz/acre). Recent research has suggested that hexazinone application one year after seeding resulted in >90% control of smutgrass for two years after application. However, the newly renovated pasture should be scouted the following year, and a second application of hexazinone may be warranted if smutgrass densities remain high.

See the EDIS publication “Smutgrass Control in Perennial Grass Pastures” for more information.

Cogongrass

This species is quickly becoming our most troublesome perennial grass throughout central and south Florida. Most become discouraged while trying to manage cogongrass because it generally takes several years of application to remove a single patch. Two herbicides are available for control, including glyphosate and imazapyr (Arsenal, Chopper, etc.), but both are non-selective. While imazapyr is generally considered to be more effective than glyphosate, care should be taken while using this product as it can harm desirable trees.

The best time for application is in the fall (October through December), but before the first frost event. Apply glyphosate at 4 qt/acre for broadcast situations (dense, large stands) or a 3% solution when treating small patches. Alternatively, apply imazapyr at 48 oz/acre for broadcast situations, or a 1% solution when treating small patches. For small patches, it is best to treat 5 to 10 feet beyond the visible perimeter of the patch to ensure that you are applying to all emerged leaves of cogongrass, regardless of the herbicide utilized. Return to the treated areas in 6 month intervals and retreat as necessary; it is allowable to return in 12 month intervals when using imazapyr as it is generally more effective long-term than glyphosate.

For more detailed information on cogongrass management in grazing areas, see the EDIS document entitled “Cogongrass Biology, Ecology, and Management in Florida Grazing Lands.”

Broomsedge

There are approximately 18 different species and varieties of broomsedge in Florida. A general perception is that if you lime the pastures that the broomsedge will go away. While this may be true for a subset of these 18, it is not true across the board. Some of these species grow a low soil pH, but some also grow quite well at high soil pH. Since there are no selective herbicides for broomsedge control in pastures or hayfields, spot-treating with 2 to 3% glyphosate is likely the best option.

We are currently investigating the role of nutrients on broomsedge. This study is currently in its third year, and we don't expect to see much difference at this point in time. It is our hope that by increasing the fertility in the pasture, that we will begin to see a decline in broomsedge over time.

An option for bermudagrass and stargrass hayfields is to broadcast glyphosate at 1 pt/acre within 7 to 10 days after cutting. This may result in some initial injury to the stargrass and bermudagrass, but it will grow out of this quite quickly.

Vaseygrass, Guineagrass, and Indian cupscale

These three species are fairly difficult to manage in improved pastures and hayfields. In bahiagrass, there are no selective options for control. Management options for bermudagrass,

stargrass, and limpograss are outlined below. For additional information on vaseygrass and guineagrass, see EDIS publication entitled “Identification and control of johnsongrass, vaseygrass, and guineagrass in pastures.”

Table 1. Control of perennial grasses in bermudagrass, stargrass, and limpograss

Bermudagrass		
Roundup, others	Glyphosate 1 to 4 oz/gal	Apply as a spot treatment when actively growing. Surrounding forage will be killed by overspray.
	Glyphosate 8 to 16 oz/acre	Spray 7 to 10 days after cutting for hay. Injury will be more severe if sprayed after 10 days.
Pastora 1 to 1.5 oz/acre	Nicosulfuron + metsulfuron	Can be applied at any time when grass weeds are actively growing, but bermudagrass injury will be less severe if treated within 7 to 10 days after cutting. Will usually take two applications; 1.5 oz at first application followed by 1.0 oz at the second application. Do not apply more than 2.5 oz/acre/year. May be tankmixed with glyphosate at 7 to 10 days after cutting.
Impose/Panoramic 4 oz/acre	imazapic	DO NOT apply to bahiagrass. DO NOT apply during spring transition or severe bermudagrass injury will occur. In summer months, expect 3–4 weeks of bermudagrass stunting after application, followed by quick recovery and rapid growth. This will reduce harvest yields of that cutting by 30%–50%. If this yield reduction is not acceptable, do not use these herbicides. Yield reductions of subsequent cuttings have not been observed. For control of crabgrass, sandspur, nutsedges, and vaseygrass, use 4 oz./acre For suppression of bahiagrass, use 12 oz./acre
Stargrass		
Roundup, others	Glyphosate 1 to 4 oz/gal	Apply as a spot treatment when actively growing. Surrounding forage will be killed by overspray.
	Glyphosate 8 to 16 oz/acre	Spray 7 to 10 days after cutting for hay. Injury will be more severe if sprayed after 10 days.
Impose/Panoramic 4 oz/acre	imazapic	DO NOT apply to bahiagrass. DO NOT apply during spring transition or severe stargrass injury will occur. In summer months, expect 3–4 weeks of bermudagrass stunting after application, followed by quick recovery and rapid growth. This will reduce harvest yields of that cutting by 30%–50%. If this

		yield reduction is not acceptable, do not use these herbicides. Yield reductions of subsequent cuttings have not been observed. For control of crabgrass, sandspur, nutsedges, and vaseygrass, use 4 oz./acre For suppression of bahiagrass, use 12 oz./acre
Limpograss		
Roundup, others	Glyphosate 1 to 4 oz/gal	Apply as a spot treatment when actively growing. Surrounding forage will be killed by overspray.
Impose/Panoramic 4 to 12 oz/acre	imazapic	DO NOT apply to bahiagrass. Applications during spring transition have caused some temporary chlorosis, but limpograss generally outgrows this injury. No long-term stand loss should be observed. Controls crabgrass, sandspur, nutsedge, vaseygrass, and suppresses bahiagrass at higher rates (12 oz).

Smutgrass Control in Perennial Grass Pastures¹

Brent Sellers, J. A. Ferrell, and N. Rana²

Introduction

Smutgrass (Figure 1)—an invasive bunch grass, native to tropical Asia—is a serious weed of improved perennial grass pastures, roadsides, natural areas, and waste areas in Florida. Results of a survey conducted by the South Florida Beef Forage Program in 2003 indicated that smutgrass ranks as the second-most problematic weed species in Florida pastures, behind tropical soda apple (which is the most problematic weed). However, because practices to control tropical soda apple have been widely adopted in Florida since that survey was conducted, it is likely that smutgrass has by now become the most problematic weed species in Florida pastures.

Two smutgrass species are found in Florida—small smutgrass (*Sporobolus indicus*; Figure 2) and giant smutgrass, which is also known as West Indian dropseed (*Sporobolus indicus* var. *pyramidalis*; Figure 3). Small smutgrass was once the predominant smutgrass species throughout Florida. By the 1990s, however, giant smutgrass had become the most common smutgrass species throughout central and south Florida. Giant smutgrass continues to move northward in Florida.



Figure 1. Smutgrass infestations are common in bahiagrass pastures throughout Florida.

Credits: B. Sellers, UF/IFAS

Mature smutgrass plants are unpalatable to livestock, but some grazing of mature smutgrass does occur. New regrowth of smutgrass, similar in quality to that of bahiagrass, is grazed for two to three weeks after burning or mowing. However, it is difficult to graze cattle on smutgrass due to the need to rotate cattle among smutgrass-infested paddocks so that growth of the smutgrass does not reach a stage where cattle will not graze the plants.

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Figure 2. Small smutgrass, once the most troublesome smutgrass species in Florida, is still found throughout the state.

Credits: B. Sellers, UF/IFAS



Figure 3. Giant smutgrass first became a problem in south Florida and is spreading north through the state. In central and south Florida, giant smutgrass is more problematic than small smutgrass.

Credits: B. Sellers, UF/IFAS

Biology

Both smutgrass species—small and giant—are perennial bunch grasses. Average bunch size of small smutgrass is approximately 8–10 inches in diameter while giant smutgrass diameter is approximately 12–18 inches.

Small smutgrass has a compact seedhead (Figure 4) with the panicle branches touching the panicle. The small smutgrass seedhead is almost always infected with a black fungus. Small smutgrass plants produce approximately 1,400 seeds per seedhead and 45,000 seeds per plant.



Figure 4. Small smutgrass seedheads are appressed near the raceme, giving the seedhead a cylindrical appearance.

Credits: B. Sellers, UF/IFAS

In contrast, giant smutgrass has an open seedhead with panicle branches directed somewhat upward (Figure 5). The seedhead of giant smutgrass is usually not infected with the black fungus, but giant smutgrass plants are sometimes infected with this fungus. Little information exists concerning seed production of giant smutgrass, but some indications suggest this plant may be a more prolific seed producer than small smutgrass.

Seed production of both species occurs throughout the growing season, and new seedheads are produced shortly after mowing or burning. The seeds, which are red to orange in color, remain attached to seedheads for some time after maturing and are spread by adhering to livestock and machinery or by movement via water and wind.

Natural seed germination has been shown to average less than 9%, and seed are thought to remain viable in the soil for at least two years.



Figure 5. Giant smutgrass seedheads are open and generally not infested with the black smut fungus that typically infests small smutgrass, but sometimes smut fungus is also found on giant smutgrass.

Credits: B. Sellers, UF/IFAS

Control

Cultural practices to control smutgrass species have not been successful to date. Mowing decreases the diameter of the clumps, but often results in increased density. Burning is thought to increase the germination of seeds in the soil seed bank. However, both burning and mowing allow for approximately two to three weeks of grazing. Smutgrass forage quality during this two- to three-week window is often equal to or slightly greater than bahiagrass.

Chemical control of smutgrass includes applying hexazinone at 1.0 lb/acre (Velpar at 2 qt/acre; Velossa at 1.67 qt/acre) to small and giant smutgrass. A surfactant may be added to Velpar (Velossa contains a surfactant), but recent research has indicated that a surfactant is not necessary since the herbicide works primarily by root uptake. Mowing smutgrass prior to hexazinone application does not increase control. Hexazinone should be applied from June through September, when rainfall is typically sufficient to move the herbicide into the root zone for uptake. There is little foliar activity from hexazinone on smutgrass. If rainfall does not occur within a two-week period after application, the herbicide treatment will likely fail. There are no grazing

restrictions for hexazinone if the application rate is below 1.13 lb /acre. However, there is a 38-day haying restriction.

Hexazinone is a highly effective herbicide but is also expensive. Experiments were recently conducted to determine when hexazinone should be applied to maximize smutgrass control, especially in light of the best timing for application to realize return on the herbicide investment. An economic analysis indicated that hexazinone should not be applied until smutgrass density is approximately 50 percent of the area of a pasture. Applications of this herbicide prior to this level of infestation will not result in enough additional bahiagrass biomass (i.e., ability to increase stocking rate) to justify the cost of application. However, in terms of preventing smutgrass infestation, it may be economically justifiable to spray highly infested areas of a pasture, even before 50% of the entire pasture is infested.

Oak trees are extremely sensitive to hexazinone, and care should be taken to stay at least 100 ft away from oak trees. If smutgrass is present under or near oak trees, spot applications of 3% glyphosate are effective.

Forage Grass Tolerance

Bahiagrass will turn slightly yellow about 15–20 days after spraying with hexazinone at the recommended rates. However, bahiagrass will recover and turn dark green within about 40 days. This green color will be darker than the non-treated pastures. Bermudagrass will turn yellow with some necrosis for approximately 30 days before new green growth occurs.

Recommendations

General

- Do not apply hexazinone within 100 feet of oak trees, because application within this range may cause death of the tree.
- Read the Velpar or Velossa label for complete instructions on reapplication interval, safety, grazing, and haying restrictions.
- Cattle may graze treated pastures if applications are less than 4.5 pt/acre Velpar and 3.75 pt/acre Velossa.
- To realize economic gains from hexazinone application, smutgrass infestation should be approximately 50 percent of pasture.
- If the initial smutgrass density covers more than 80 percent of the pasture area (if 8 out of 10 regular steps touch the base of smutgrass plants), complete renovation of the pasture should be considered.

Bahiagrass/Bermudagrass Pastures

- Graze pasture in the spring until the beginning of the rainy season.
- Apply 2.0 qt/acre Velpar (1.67 qt/acre Velossa) during the summer rainy season but not later than the end of September. Apply when plants are actively growing and rainfall is dependable and consistent.
- Fertilization after hexazinone application will increase forage production and allow bahiagrass to quickly fill the open areas created by dying smutgrass.

Floralta Limpograss

- Hexazinone is not currently labeled for smutgrass control in limpograss.

Stargrass

- Hexazinone is not currently labeled for smutgrass control in stargrass.

Mulato

- Hexazinone is not currently labeled for smutgrass control in Mulato as it will be severely injured—DO NOT USE.

Pasture Renovation

In highly infested bahiagrass pastures where smutgrass groundcover exceeds 70%–80%, pasture renovation should be considered. Spray the entire pasture with 4 qt/acre glyphosate and begin tillage practices no earlier than three weeks after application. Repeated tillage will destroy newly emerged smutgrass and will aid in depleting the soil seedbank. The final seedbed should be a smooth, flat surface devoid of vegetation. For additional information on bahiagrass varieties and seeding rates, see EDIS publications AG342/SS-AGR-332, *Bahiagrass (Paspalum notatum): Overview and Management* (<http://edis.ifas.ufl.edu/ag342>) and AG107/SS-AGR-161, *Forage Planting and Establishment Methods* (<http://edis.ifas.ufl.edu/ag107>).

Even with repeated tillage following glyphosate application, smutgrass will likely emerge with bahiagrass, and smutgrass seedheads will be present by the following summer growing season. One year after seeding and during the rainy season, apply 0.5 lb/acre hexazinone (Velpar at 32 oz/acre or Velossa at 27 oz/acre). Recent research has suggested that hexazinone application one year after seeding resulted in >90% control of smutgrass for two years after application. However, the newly renovated pasture should be scouted the following year, and a second application of hexazinone may be warranted if smutgrass densities remain high.

Cogongrass (*Imperata cylindrica*) Biology, Ecology, and Management in Florida Grazing Lands¹

B. A. Sellers, J. A. Ferrell, G. E. MacDonald, K. A. Langeland, and S. L. Flory²

Cogongrass is found on every continent and is considered a weedy pest in 73 countries. In the U.S., cogongrass is found primarily in the Southeast. It was accidentally introduced into Alabama in the early 1900s, and purposely introduced as a potential forage and soil stabilizer in Florida (and other states) in the 1930s and early 1940s. However, soon after investigations began it was realized that cogongrass could be a weedy pest. Since its introduction, cogongrass has spread to nearly every county in Florida. In some cases, it has completely taken over pastures so that it is the only species present. This is a common thread where cogongrass invades; it quickly displaces desirable species and requires intensive management.

There are many reasons why cogongrass is such a prolific invader. It is a warm-season, perennial grass species with an extensive rhizome root system. In fact, at least 60% of the total plant biomass is often found below the soil surface. In addition to the rhizome root system, cogongrass adapts to poor soil conditions, and its fires burn so hot that they eliminate nearly all native species. Cogongrass is drought tolerant and has prolific wind-dispersed seed production. Additionally, it can grow in both full sunlight and highly shaded areas, although it is less tolerant to shade.

Cogongrass spreads through its creeping rhizome system and seed production. The rhizomes can penetrate to a depth of 4 feet, but most of the root system is within the top 6 inches of the soil surface. The rhizomes are responsible for long-term survival and short-distance spread of cogongrass. Long-distance spread is accomplished through seed production. Seeds can travel by wind, animals, and equipment. Seed viability is significant in north Florida and other states of the Southeast; however, there are no confirmed cases of viable seed production in central and south Florida.

An established cogongrass stand invests heavily in its perennial root system. These infestations are capable of producing over 3 tons of root biomass per acre. This extensive network of rhizomes is capable of conserving water while the top growth dies back during prolonged drought. This is essentially a survival mechanism to keep the rhizome system alive. Another key to cogongrass invasion is that the root system may produce allelopathic chemicals that reduce the competitive ability of neighboring plants.

Identification

Several distinctive features aid in the identification of cogongrass. First, cogongrass infestations usually occur in circular patches. The grass blades tend to be yellow to

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2. B. A. Sellers, associate professor, Agronomy Department, Range Cattle Research and Education Center, Ona, FL; J. A. Ferrell, associate professor, Agronomy Department; G.E. MacDonald, professor, Agronomy Department; K. A. Langeland, professor, Agronomy Department; and S. L. Flory, assistant professor, Agronomy Department; Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL 32611.

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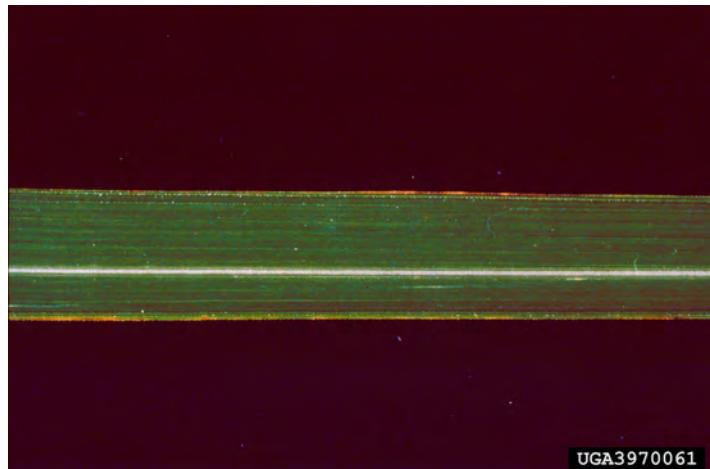
Figure 1. Cogongrass plants are yellow to green in color. Note that the edges of the leaf tend to have more yellow than green.

Credits: G. Keith Douce, University of Georgia, www.forestryimages.org.

green in color (Figure 1). Individual leaf blades are flat and serrated, with an off-center prominent white midrib (Figure 2). The leaves reach 2–6 feet in height. The seed head (Figure 3) is fluffy, white, and plume-like. Flowering typically occurs in spring or after disturbance of the sward (mowing, etc.). Seed heads range from 2 to 8 inches in length and can contain up to 3,000 seeds. Each seed contains silky-white hairs that aid in wind dispersal. When dug, the rhizomes (Figure 4) are white, segmented (have nodes), and are highly branched. The ends of the rhizome are sharp pointed and can pierce the roots of other plants.

Forage Value

Cogongrass has been used in Southeast Asia as forage because it is the dominant vegetation on over 300 million acres. In these areas it was found that only very young shoots should be grazed or cut for hay. At this stage, the leaves lack sharp points and razor-like leaf margins. For about four weeks following a prescribed burn, crude protein of regrowth is comparable to bahiagrass. Crude protein of mature stands rarely attains the minimal 7% level needed to sustain cattle, making supplementation essential for livestock production. Cogongrass yields are relatively low, even under heavy fertilization, and usually do not exceed 5 tons per acre.



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Figure 2. Cogongrass leaves have serrated edges and a prominent, white, off-center midrib.

Credits: L. M. Marsh, Florida Department of Agriculture and Consumer Services, www.forestryimages.org.



UGA2155056

Figure 3. Cogongrass seed heads are fluffy and white. Each plant produces nearly 3,000 seeds.

Credits: John D. Byrd, Mississippi State University, www.forestryimages.org.

Management

For many years researchers all over the world have studied cogongrass control. During this time nearly all available herbicides have been tested, but few effective products have been found. For example, all of the commonly used pasture herbicides such as metsulfuron, 2,4-D, triclopyr, Velpar, and other combinations have little to no activity on cogongrass. Only glyphosate (Roundup, etc.) and imazapyr (Arsenal, Stalker, etc.) herbicides have been found to be effective, but long-term control is rarely achieved.

Imazapyr is an extremely effective herbicide that controls a variety of weeds, from herbaceous to woody species. One or two applications of imazapyr (0.75 lb/acre) will often effectively control cogongrass for 18–24 months. However, there are several disadvantages to using this herbicide. First, imazapyr will severely injure or kill forage grasses such as bermudagrass and bahiagrass. It also has a long soil half-life and will remain in the soil for several months after application. This often leads to “bare ground” for up to 6 months in the application area because of the non-selective nature of this herbicide. Imazapyr also has the potential to move down slopes during periods of rainfall, killing or injuring other species in the runoff area (oaks and other hardwood trees are especially sensitive). Second, imazapyr can only be used as a “spot-treatment” with no more than 10% of the pasture area treated per year.

Similarly, glyphosate is also a non-selective herbicide that effectively controls a variety of weeds. Unlike imazapyr, glyphosate possesses very little to no soil activity. Non-target effects caused by runoff during high rainfall events are not likely. Since glyphosate has no soil activity, it does not take very long for weeds or desirable grasses to reinfest the treated areas. Cogongrass will likely reinfest the area if only one application of glyphosate is applied during the same year. Research in Alabama has revealed that it takes approximately three years of two applications per year to reduce cogongrass rhizome biomass by 90%.

Small Infestations

Early detection of cogongrass in any setting is extremely important. A young infestation will be much easier to treat and eradicate than established infestations. In this case, we would define a small patch as 20–30 feet or less in diameter. Even for a small patch, monitoring is required after the initial application to ensure that any re-sprouting is quickly treated. See Table 1 for specific timelines and suggested herbicide rates.

Large Infestations

Large infestations are 30 feet or larger in diameter. These types of infestations can be considered as established and likely have a large, intact root system. This will require more herbicide treatments to completely eradicate cogongrass. See Table 2 for specific timelines and suggested herbicide rates.

Integrated Management

Herbicide inputs alone are rarely successful in eradicating perennial species like cogongrass. In these cases, we need to



UGA1380061

Figure 4. Cogongrass rhizomes are segmented (have nodes) where new shoots are able to grow.

Credits: Chris Evans, River to River CWMA, www.forestryimages.org.

use all of the tools we have to remove an unwanted species to reestablish a desirable species. This type of strategy is best employed in an area where cogongrass has long been established and is the predominant species present. See Table 3 for specific timelines and suggested herbicide rates.

In general, burn the area infested with cogongrass in August to September. One to four months later, treat the burned area with a mixture of imazapyr and/or glyphosate. Take soil samples prior to spring tillage the next growing season to ensure that the soil pH is adequate for your desirable forage species. Till the treated area the following spring to a depth of at least 6 inches and prepare a seedbed.

Consult with your local county Extension agent to consider your options for forage cultivars and fertility recommendations. Getting a good start on the desirable forage will help limit cogongrass reinfestations in your pasture. Continue to monitor this area in six-month intervals until the fourth year. Spot treat with glyphosate when necessary to remove any new cogongrass growth.

Table 1. Herbicide suggestions for small infestations of cogongrass in grazing areas. This includes both improved and native rangeland. These concentrations are good for mixing in small (3–30 gallon) sprayers. Please read the entire label of the suggested products prior to treating existing cogongrass stands.

	Timing	Herbicide Rate	Application Notes
1 st year	Fall (August-November)	1% Arsenal/Stalker + 0.25% non-ionic surfactant	Treat only 10% of the area to be grazed. No grazing restrictions, but do not cut for hay for 7 days. Read the herbicide label for mixing instructions.
		3% glyphosate	No grazing or haying restrictions. Read the herbicide label for mixing instructions.
		0.5% Arsenal/Stalker + 2% glyphosate + 0.25% non-ionic surfactant	Treat only 10% of the area to be grazed. No grazing restrictions, but do not cut for hay for 7 days. Read the herbicide label for mixing instructions.
2 nd year	Spring (monitor regrowth)	2–3% glyphosate	See above.
	Fall (monitor regrowth)	2–3% glyphosate	See above.
3 rd year – until eradicated	Spring – Fall (monitor regrowth)	Spot treat at the above rates for the 2 nd year.	

Table 2. Herbicide suggestions for large cogongrass infestations in grazing areas, including both improved and native rangeland. These suggestions are intended for large (>1000 gallon) sprayers. Please read the entire label of the suggested products prior to treating existing cogongrass.

	Timing	Herbicide Rate	Application Notes
1 st year	Fall (August-November)	48 oz/acre Arsenal/Stalker + 0.25% non-ionic surfactant	Treat only 10% of the area to be grazed. No grazing restrictions, but do not cut for hay for 7 days. Read the herbicide label for mixing instructions.
		3 to 4 qt/acre glyphosate	Do not graze for 8 weeks. Read the herbicide label for mixing instructions.
		24 oz/acre Arsenal/Stalker + 2 qt/acre glyphosate + 0.25% non-ionic surfactant	Treat only 10% of the area to be grazed. No grazing restrictions, but do not cut for hay for 7 days. Read the herbicide label for mixing instructions.
2 nd year	Spring (monitor regrowth)	2–3% glyphosate	No grazing or haying restrictions.
	Fall (monitor regrowth)	2–3% glyphosate	No grazing or haying restrictions.
3 rd year – until eradicated	Spring – Fall (monitor regrowth)	Spot treat at above rates for the 2 nd year.	See above.

Table 3. Control of cogongrass using an integrated approach. Adjust your timelines based on your location in Florida. For example, burning should be performed earlier in north Florida than in south Florida because of the first onset of a potential killing frost. Please read all herbicide labels prior to treating cogongrass for restrictions and mixing instructions.

	Timing	Herbicide Rate	Application Notes
1 st year	Summer – Fall (August-November)	1. Burn	Cogongrass fires burn extremely hot. Be sure to have firebreaks in place before attempting to burn cogongrass.
		2. Apply herbicide: 24 oz/acre Arsenal/Stalker + 2 qt/acre glyphosate + 0.25% non-ionic surfactant	Treat only 10% of the area to be grazed. No grazing restrictions, but do not cut for hay for 7 days. Read the herbicide label for mixing instructions.
		3. Take soil samples.	Have the soil pH tested at a reputable laboratory. Amend the soil as needed to grow desirable forage.
2 nd year	Spring	1. Tillage	Prepare a seedbed for desirable forage species. Repeated tillage will help to desiccate any remaining cogongrass rhizomes.
		2. Plant desirable forage.	Please consult your local Extension agent for up-to-date recommendations on forage cultivars and fertility recommendations.
3 rd year	Spring (monitor regrowth)	2–3% glyphosate	No grazing or haying restrictions.
	Fall (monitor regrowth)	2–3% glyphosate	No grazing or haying restrictions.
4 th year – until eradicated	Spring-Fall (monitor regrowth)	Spot treat at the above rates for the 3 rd year.	See above.

Identification and Control of Johnsongrass, Vaseygrass, and Guinea Grass in Pastures¹

H. Smith, J. Ferrell, and B. Sellers²

Johnsongrass is a common perennial grass that grows throughout the South and Midwest. It is so common and well known as a troublesome weed that any large undesirable grass is often called johnsongrass. This is problematic because it is one of three perennial grasses found in pastures. Vaseygrass and guinea grass are often misidentified as johnsongrass but they have very different herbicide recommendations. Calling a plant johnsongrass when it is really vaseygrass or guinea grass can result in the wrong recommendation and lead to an expensive herbicide failure.

Identification: Johnsongrass, Vaseygrass, Guinea Grass

All three grasses have a prominent white midrib that extends the length of the leaf. But few similarities exist beyond this characteristic.

Growth Habit

All three grasses are perennial, but only johnsongrass has a creeping rhizome system and grows in patches rather than in individual bunches. Vaseygrass and guinea grass are both bunch-type grasses without a significant rhizome system. Additionally, vaseygrass is most commonly found in wet fields or along drainage ditches. Johnsongrass and guinea grass prefer dryer sites.

Seedhead

Johnsongrass and guinea grass have an open panicle seedhead that is angular. Color and size are the key differences between johnsongrass and guinea grass seedheads. Johnsongrass seeds are much larger and have a red/black mottled color, while the guinea grass seeds are smaller and somewhat green. Vaseygrass has a very different seedhead with alternating spikelets forming silky hairs around the seeds. Seeds are produced along the entire length of the seedhead branch, which does not occur in johnsongrass or guinea grass seedheads.



Figure 1. From left to right, guinea grass seedhead (Credits: Hunter Smith); johnsongrass seedhead (Credits: Brent Sellers); vaseygrass seedhead (Credits: Brent Sellers).

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Seeds

Guinea grass has small, oval, light green seeds, which often have wrinkles. Vaseygrass seeds have similar characteristics but are flatter, with the presence of hairs. Johnsongrass has much larger, pointed seeds that develop a reddish/brown tint as they mature.



Figure 2. From left to right, guinea grass seedhead branch (Credits: Brent Sellers); johnsongrass seedhead branch (Credits: Hunter Smith); and vaseygrass spikelet (Credits: Brent Sellers).

Stems

The stems of johnsongrass and guinea grass can look very similar. Inspection of the stems will show scattered but abundant hairs along the stem of guinea grass. Stem hair on guinea grass varies because of the different biotypes. Johnsongrass stems are totally smooth with no hairs. Vaseygrass stems have hairs where the leaf meets the stem or on the stem toward the base of the plant. This is because vaseygrass will generally lose stem hairs as the stems elongate.

Leaves

Johnsongrass leaves have a large white midrib and a smooth, glossy appearance. Guinea grass leaves have a less prominent white midrib, and the undersides are rough with stiff hairs. Vaseygrass leaves are long and narrow with an indented midrib and crinkled leaf margins.



Figure 4. From left to right, guinea grass leaf blade; johnsongrass leaf blade; vaseygrass leaf blade. Credits: Hunter Smith



Figure 5. Vaseygrass leaf margin. Credits: Hunter Smith

Roots

A fifth and final identification method is to pull or dig up the roots. All three of these grasses are perennial, but johnsongrass has large white rhizomes that are easily seen if the plant is well established. Vaseygrass and guinea grass have smaller, more fibrous root structures compared to johnsongrass.



Figure 6. Guinea grass root structure. Credits: Hunter Smith



Figure 7. Vaseygrass root structure. Credits: Brent Sellers



Figure 8. Johnsongrass rhizome. Credits: Brent Sellers

Vaseygrass

Impose (bermudagrass only): Vaseygrass control can be accomplished by using 6–8 ounces per acre. This rate of Impose will be highly injurious to bermudagrass and one cutting of hay will likely be lost. This injury can be minimized if the application is made immediately after hay removal and before the bermudagrass leaf-out. Additionally, do not apply Impose until after the first hay cutting when rainfall is common.

Glyphosate: Spot spraying with 1% solution (1.2 oz/gal) can be effective. Care should be taken to avoid contact with desirable grasses.

Guinea grass

Glyphosate: Spot spraying with 1% solution (1.2 oz/gal) can be effective. Care should be taken to avoid contact with desirable grasses.

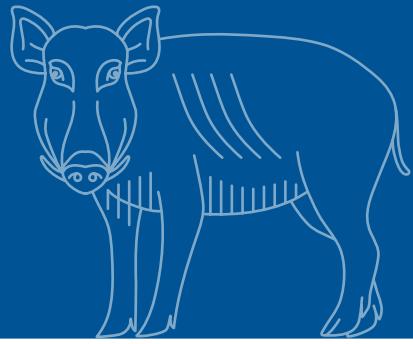
Control

Johnsongrass

Outrider: For best johnsongrass control, apply 1.33 ounces per acre when grass is actively growing and is at least 18–24 inches tall, up to the heading stage.

Impose (bermudagrass only): Use 4–6 ounces per acre on johnsongrass less than 24 inches. Higher rates can be used, but unacceptable injury on bermudagrass will likely occur. Although 4 oz of Impose can control johnsongrass, some regrowth should be expected on older stands that are large at the time of application.

Pastora (bermudagrass only): Use 1 oz/A on seedling johnsongrass (rhizomes < 18") and 1.5 oz/A on mature stands. Bermudagrass injury will occur with Pastora, but will be less than that observed with Impose. Maximum application rate of Pastora is 2.5 ounces per acre per year.



FERAL SWINE

Raoul Boughton, Assistant Professor • Bethany Wight, Research Assistant

#001 Invasive Rangeland Vertebrate Factsheet Series



SCIENTIFIC NAME: *Sus scrofa*

SYNOMYS: Wild Hog, Feral Hog, Wild Boar, Razorback, Piney Woods Rooter

HABITAT: All habitats with a water resource, especially agricultural areas and wetland/upland interface

PHYSICAL CHARACTERISTICS: Black, brown or brindled in color, juveniles striped

WEIGHT: Adults 75-250 lbs

DEMOGRAPHIC RATE: 115 day gestation, able to produce 2 litter/year, 6-8 piglets/litter in the wild. Helps populations grow rapidly

LIFESPAN: Average of 1-2 years, known to live up to 9-10 years in the wild

DISPERSAL: Female and young stay together in groups called sounders. Mature males disperse, sometimes more than 100 miles. Female dispersal activities are unknown.

An Ecological and Agricultural Invader

Breeding



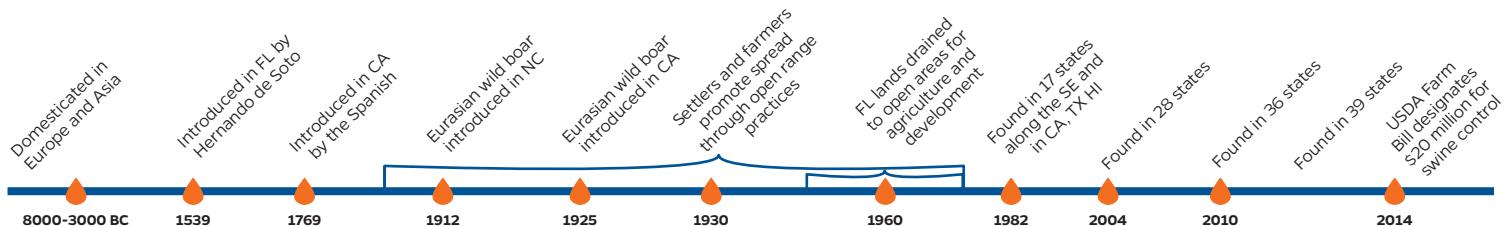
Habitat



Status

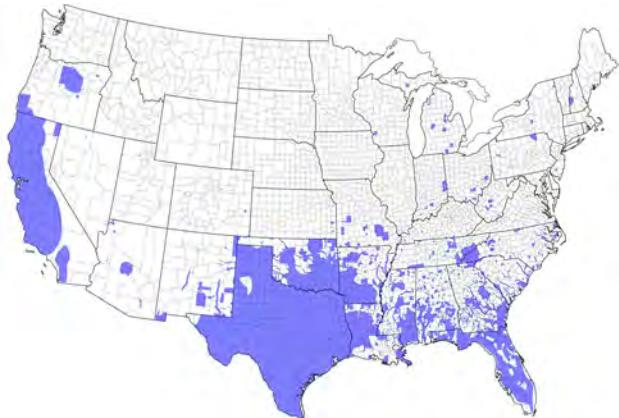


HISTORY: Feral swine are not native to the Americas and were introduced by Spanish explorers in the 1500s. In Florida, domesticated swine are thought to have first been introduced in 1539 by Hernando de Soto who settled Charlotte Harbor in Lee County. Later settlers and farmers used open range livestock practices, promoting the spread of swine. Feral swine are descendants of escaped/released domestic swine, hybrids of Eurasian wild boar x domestic swine, or wild boar in non-native habitat.



FERAL SWINE FAST FACTS

- Forage by rooting, which can negatively impact ecosystems
- No sweat glands, require water and shade to cool in hot environments
- One of the highest reproductive rate of mammals in North America
- Typically found in groups called sounders, males often solitary



Distribution of feral hogs in 2009. (Courtesy of Southeastern Cooperative Wildlife Disease Study)

DISTRIBUTION: Previously presumed to be limited to the south by harsh winters, they are now estimated to be breeding in 39 states, as far north as Michigan, North Dakota, and into Canada. The largest populations are found in Texas, California, Florida, and Hawaii. Population estimates in Florida are >500,000 which could be a great underestimate. Map courtesy of the SCWDS, University of Georgia.

Tracks



Rooting along edge of wetlands common

Wallows in shady sites used often

Rubs on posts & trees likely used as scent marks

Swine tracks

Swine feces

How You Can Help

- Do not relocate or transport feral swine
- Control feral swine on your property
- Collaborate with neighbors to control large areas
- Work with your local wildlife agency

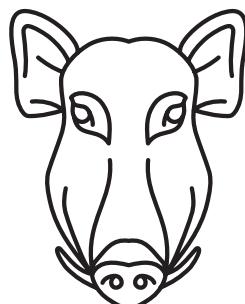
IMPACTS: The most common type of damage by feral swine is from rooting. When swine root to get food they burrow into the soil with their snouts to find roots, tubers, fungus, etc. This rooting loosens the soil, destroys native vegetation, and modifies the chemistry and nutrients of the soil. Feral swine can negatively impact not only natural ecosystems but also agricultural areas, livestock, and even residential areas. Feral swine also carry numerous diseases, some of which are transmittable to wild and domestic animals as well as humans.



Some wild boar have large tusks.

To learn more see these fact sheets at www.rangelandwildlife.com

- Feral Swine Damage Cost
- Feral Swine on Your Property
- Feral Swine Diseases
- Dealing with Damaging and Dangerous Wildlife



Estimating forage loss costs due to feral swine rooting to cow/calf production in South Central Florida.

Brittany Bankovich¹, Samantha Wisely¹, Elizabeth Boughton², Raoul Boughton¹, and Michael Avery³

¹University of Florida, WEC and RCREC, ²MacArthur AgroEcology Research Center, ³USDA Wildlife Services

The economic costs associated with feral swine and their foraging behavior has been lacking. Foraging behavior of feral swine is destructive because they till up the soil with their snouts to depths up to one foot. There are many different types of economic costs associated with feral swine on ranches including: loss of forage from rooting, renovating pastures after rooting, feral swine usage of cattle supplemental feed, and invasive weed spread, and disease spread. In this study we focus on the economic costs of forage loss due to feral swine rooting behavior. We compared rooted to unrooted plots in both improved and semi-native grasslands to assess the impact that rooting of feral swine has upon forage species. The study plots were located at the MacArthur Agro-Ecology Research Center and economic analysis approach was recommended by James McWhorter and Chris Prevatt, Highlands County Livestock Agent and RCREC Livestock Economist, respectively.

Rooting by feral swine was observed in semi-native pastures in January 2013. To quantify this rooting, we established transects and measured the area of rooted patches once in 2013 and again in 2014 after another rooting event. We mapped all freshly rooted patches that fell along transect lines that were 4m² or greater in area using a Trimble GeoXT (Fig 1a). In improved pastures, we observed initial rooting in February 2013. In March 2013, we established transects in each of two 20 ha improved pastures and mapped rooted patches (Fig 1b) using the same methodology as above. Improved pastures were not rooted again in 2014. As rooted patches were only found if they bisected a transect it is likely these are minimum estimates of rooting impact. There was at minimum 13% of area rooted in semi-native and 2% rooted in improved pasture. For each rooted patch we established paired subplots, 4x1m² subplots within the rooted patch and 4x1m² subplots outside of the rooted patches. Species composition and cover was recorded in each subplot monthly and averaged for each set of 4 subplots, and the difference between cover of forage compared between rooted and unrooted subplots was calculated.

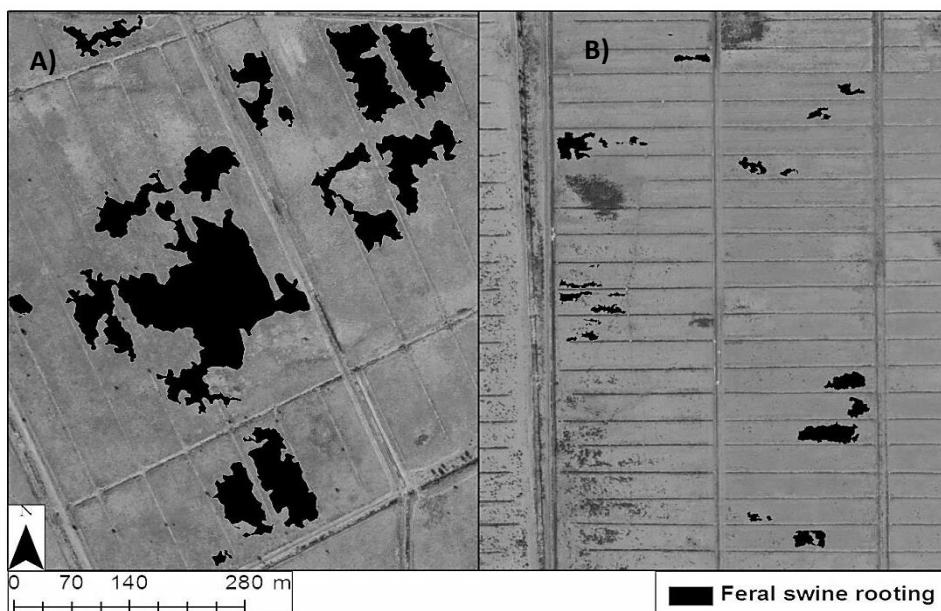


Figure 1: Mapped rooting in semi-native pasture (13% rooted) (A) and improved pasture (2% rooted) (B).

The semi-native pastures included percent cover measurements of 23 plant species, bare ground, litter, and water in a total of 1,672 subplots (152 plots over 11 sampling events). The improved pasture included percent cover measurements of 18 plant species, bare ground, litter, and water from 832 subplots (64 plots over 13 sampling events). In both pasture types, rooted subplots had less forage grass throughout the year compared to unrooted subplots. During winter when native grassland pastures are used for grazing (Oct-Feb), there was 53-68% less forage grasses in rooted plots compared to unrooted plots with an average of **60% less** forage grass per m² in rooted plots compared to unrooted plots, and the loss of forage cover remained similar the entire study (Fig 2). In improved pastures we found 67% (May 2013, 2 months into the study) and 31% (February 2014, 10 months into the study) less bahiagrass in rooted plots compared to unrooted plots suggesting a slow recovery of bahiagrass after rooting. In June, one of the most productive months for bahiagrass there was **44% less** bahiagrass cover per m² in rooted plots compared to unrooted plots (Fig 3).

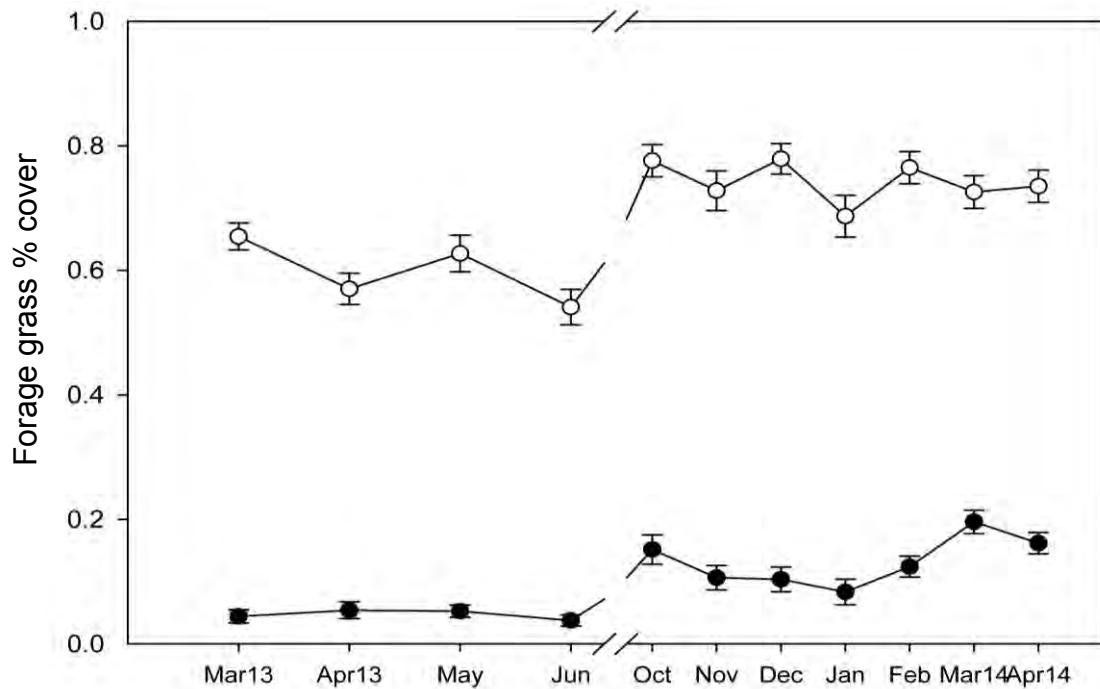


Figure 2: Forage % cover in unrooted (open circles) and rooted plots (filled circles) of semi-native pastures. Forage species included *Andropogon virginicus*, *Panicum longifolium*, *Axonopus fissifolius*, *Panicum hemitomon*, *Cynodon dactylon*, *Paspalum notatum*.

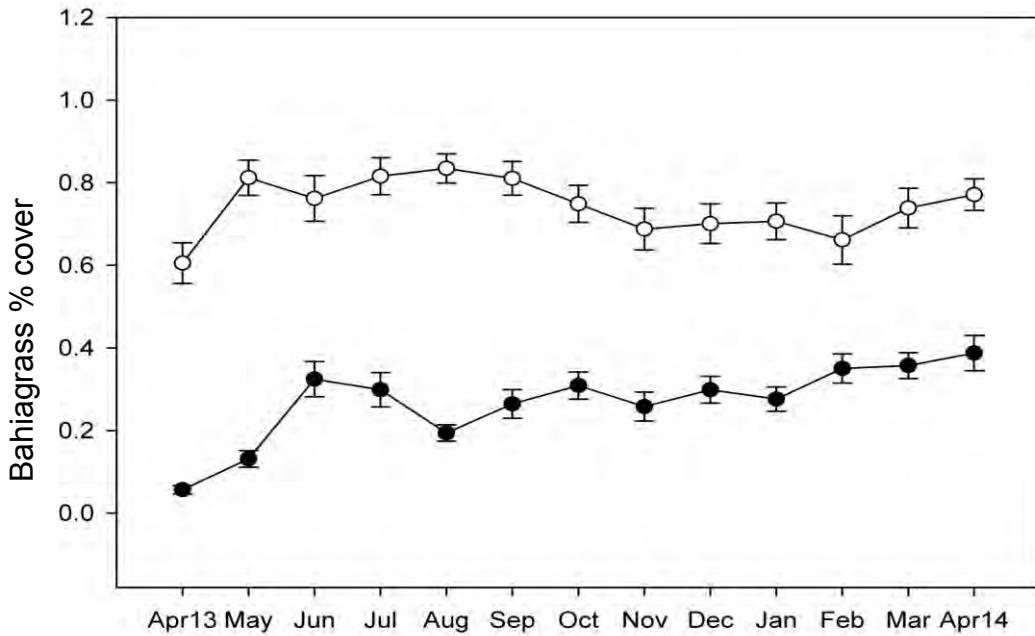


Figure 3: Forage % cover in unrooted (open circles) and rooted plots (filled circles) of improved pastures

Economic Analyses

To estimate the economic cost feral swine may be causing through the destruction of forage during rooting, as exemplified above, we modified a simple economic model based on cow/calf pairs and the amount of beef, measured as calf weight produced under relevant stocking densities (Ferrel et al 2006). The model is defined as;

$$W * CW * k / R$$

W=weaning % (we used 75%)

CW=average calf weight (we used 550lbs)

k=rooting constant adjustment based on %loss of forage per acre

R=Stocking Rate adjustment to per acre (we used 1 cow-calf pair per 3 acres for improved and 1 cow-calf pair per 20 acres for semi-native)

For example, we found that 2% of improved pasture is rooted and that within that 2% of rooting there is a 44% loss of forage (June, Fig 3). This equates to a 0.088% loss of forage per acre, or 99.12% of forage remaining. The assumption we make in the model is that this forage loss equates to an equivalent loss in beef production. With no forage loss in our equation k=1 and per acre yield of beef (for 550lb calf) at 75% weaning rate is 137.5lbs/acre. At today's market price of \$2.57 per pound this equals \$354.01 value per acre. Now if we investigate our forage loss at 0.088%, as measured in our research, we only produce 136.29lbs of beef/acre worth \$350.89, or in other words a loss of \$3.12/acre. That does not sound like a lot but if you have 5,000 acres of improved pasture that is \$15,600 a year loss just from 2%

rooting in improved pasture. The table below has been calculated for both improved pasture (stocked 3 acres per cow-calf) and semi-native pasture (stocked 20 acres per cow-calf), across a range of rooting pressures.

Table 1: Estimates of cost of rooting in improved and semi-native pasture.

Improved Pasture	Calf Production lbs/acre	Calf Value \$/acre	Cost of rooting \$/acre
Not Rooted	137.5	\$354.01	0
2% Rooted	136.29	\$350.89	\$3.12
10% Rooted	131.45	\$338.43	\$15.58
20% Rooted	125.4	\$322.85	\$31.15
Semi-native Pasture			
Not Rooted	16.5	\$42.48	0
13% Rooted	15.213	\$39.17	\$3.31
20% Rooted	14.52	\$37.38	\$5.10
30% Rooted	13.53	\$34.83	\$7.65

Finally, if we scale up to a larger region and assume rooting occurs at a 2% level across this area on improved pastures, the cost of feral swine to the industry could be quite significant. For example, in the counties of Highlands Okeechobee, Osceola, Polk, and Hardee there is 718,088 acres of improved pasture. In one year of rooting at a 2% level this could cost the region \$2,240,434 in lost beef production, or increased cost in supplementation to account for the decreased forage. We have seen areas that have experienced much greater levels of rooting than 2% (Fig 4) and we know from our data that there is a slow recovery of rooted areas back to bahiagrass, suggesting that from year to year a cumulative effect is possible. Just for thought, if the region ever experienced 10% rooting on average across improved pastures it would be equivalent to \$11,187,811 lost beef production.



Figure 4: Intense rooting (30-50%) by feral swine in improved pasture

Ferrell, J. A., Mullahey, J. J., Dusky, J. A., & Roka, F. M. (2006). Competition of giant smutgrass (*Sporobolus indicus*) in a bahiagrass pasture. *Weed science*, 54(1), 100-105.

Trapping Feral Swine

An effective way to continuously reduce or control wild swine populations.

Successful trapping is a process requiring several key factors:

1. Locating high swine use areas for trapping
2. Pre-baiting and baiting traps
3. Choosing effective trap and gate design
4. Patience and persistence

Three basic trap types are used box/cage trap, panel trap, and corral/silo trap. In addition there are several gate and trigger options. You should choose a trap type that is most efficient and cost effective for your needs. Some things to consider when managing wild swine with traps:

- Density of swine or sounder size
- Cost
- Portability and weight
- Presence of non-target species
- Number of traps needed
- Surrounding wild swine management efforts



Choosing Trap Locations

- Look for signs of high swine activity, such as evidence of:
 - Rooting, tracks, and wallows
- Site of damage not always the best place, swine spend a lot of their time in shaded areas close to a water source. Scout low-lying areas such as river or creek bottoms, wetlands and forest edges.
- Travel routes to and from these areas are ideal for higher catch opportunities and multiple sites may increase your success.
- Vehicle access is usually essential.



Pre-baiting and Baiting

- It is important to allow swine enough time to become accustomed to the location and entering the trap.
- Pre-baiting the location prior to trap placement attracts swine regularly to a specific site increasing trap success.
- After pre-baiting erect trap, secure the gate and layout bait trail. Monitor the traps with game cameras to ensure swine are readily entering the trap for at least 3 nights.
- When swine are entering freely set trap but do not bait heavily around the trigger. Instead bait heavily along the inside of the trap opposite the trigger location. *Place only a small amount of bait around the trigger which will allow more swine to enter and be drawn to larger bait pile before the mechanism is triggered.*
- Common baits used include: dry or fermented corn, vegetable/produce scraps, molasses, red Kool-Aid, and commercial attractants.

Common reasons for poor trapping success

- Bad trap placement.
- Not enough pre-baiting, suggest up to 2 weeks.
- Faulty trigger or escape due to poor trap construction.
- Too much natural food available.
- Hunting and dogs can alter swine behavior and reduce trap success.





Feral Swine Portable Wooden Box Trap

A relatively cost effective, portable trap, ideal for individual swine or small groups of swine.

- Rectangular or square made of treated lumber or wood fence panels.
- Typically 4 feet wide, 8 feet long, and 5 feet high, no top or bottom
- 5 foot height prevents swine from climbing or jumping out
- Usually heavy enough to prevent lifting by swine, but may add T-posts

PROS

- Simple to construct
- Cost effective
- Easy to store, transport and relocate

CONS

- Limited to small number of swine
- May appear confining to trap shy swine
- May require long term maintenance

Material	Quantity	Estimated Cost
2" by 4" by 10' board	4	\$5/each
1" by 4 (or 6)" by 10' board	17	\$4/each
Decking screws	1 box	\$10/box
TOTAL		Approx. \$100.00

* Does not include gate cost, see Gate sheet

Feral Swine Portable Cage Trap

A stronger portable trap, ideal for individual swine or small groups of swine.

- Rectangular trap made from heavy-gauge wire livestock panels welded to steel frame or purchased from vendor (see purchase sheet).
- Typically 4 feet wide, 6-12 feet long, and 4-5 feet high.
- Traps <5 feet tall should have top panel to prevent swine escape.
- New round design easier to transport by rolling.



PROS

- Can self-construct or purchase
- May appear more open to trap shy swine
- Easy to store, transport and relocate
- Top panel prevents swine from jumping out

CONS

- Limited to small number of swine
- Top panel may prevent escape of non-target species

PURCHASING INFORMATION

- Multiple vendors, we do not endorse any particular company or trap type

- Voorhies Outdoor Products, LLC Hog Trap - Metal trap with 3 rooter doors
 - 8' by 4' by 3'
 - \$399.99
- Forestry Suppliers—Steel Cage Hog Trap with spring loaded door
 - 8' by 4' by 3'
 - \$400.00
- Avon Park Correctional Institute—Work Study Program
 - 8' by 4" by 5' with a 3' by 5' guillotine door
 - Purchase of materials only, usually \$500/trap.



Feral Swine Panel Trap

A cost effective, portable, easy to construct trap that allows design flexibility. Can be built to catch small or large groups of swine.

- Constructed from welded mesh panels wired together on T-posts. (Min. 4" by 4" spacing on bottom 3')
- Flexibility in shape and size allows variety of possibilities to suit individual needs or materials.
- Traps should be at least 5 feet tall to prevent swine escape (can also add jump wire)

PROS

- Cost effective
- Easy for one person to self-construct
- Easy to store, transport and relocate
- Flexible design for shape and size

CONS

- Catch capability depends on size and materials

Material	Quantity	Estimated Cost
16' by 5' Panel	2	\$50-70/each
5' T-posts	8-10	\$3.60/each
TOTAL		Approx. \$150.00

* For a 8' by 8' by 5' trap, Does not include gate cost, see Gate sheet



Feral Swine Corral Trap

The most effective for trapping large groups of swine. Requires more time and effort to construct and relocate.

- Constructed from wire livestock panels fastened to 5-6 1/2' T-posts using U-bolts and cable clamps.
- Using 3-4 5' by 16' panels should make a trap large enough to catch most sounders.
- Can vary in shape but are typically round to prevent swine from piling up in corners and possibly climbing or jumping out.

PROS

- Very effective for trapping large groups
- Allows non-target species to escape
- Open appearance may appear less threatening to trap shy swine

CONS

- More time and effort to construct and relocate

Material	Quantity	Estimated Cost
16' by 5' Panel	3	\$50-70/each
6 1/2' T-posts	10	\$4.50/each
5/16 cable clamps	12	\$1.50/each
5/16 by 1 1/2' U-bolts	24	\$2.00/each
TOTAL		Approx. \$290.00

* Does not include gate cost, see Gate sheet



Feral Swine Silo Trap

A cost effective trap that allows for the capture of large groups of swine. Allows design flexibility and can be used with a variety of gates or funnels.

- Constructed from either continuous mesh panel or multiple livestock panels fastened to 5' T-posts using U-bolts and cable clamps.
- Requires more time to construct but potential for high capture rate.
- Can vary in shape but are typically round to prevent swine from piling up in corners and possibly climbing or jumping out.
- Using funnel entry cheaper than building gate.

PROS

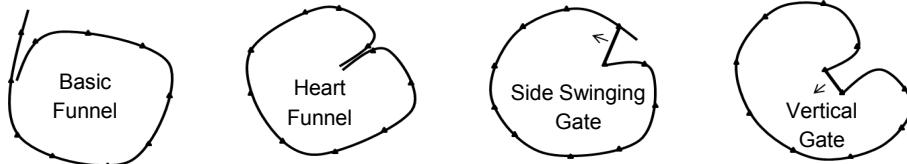
- Very effective for trapping large groups
- Cost effective
- Flexibility in design, shape, size and gate or funnel used

CONS

- More effort for one person to construct
- Better for semi-permanent sites

Material	Quantity	Estimated Cost
16' by 5' Panel	2	\$50-70/each
5' T-posts	10-13	\$3.60/each
5/16 by 1 1/2' U-bolts	24	\$2.00/each
TOTAL		Approx. \$210.00

* Does not include gate cost, see Gate sheet

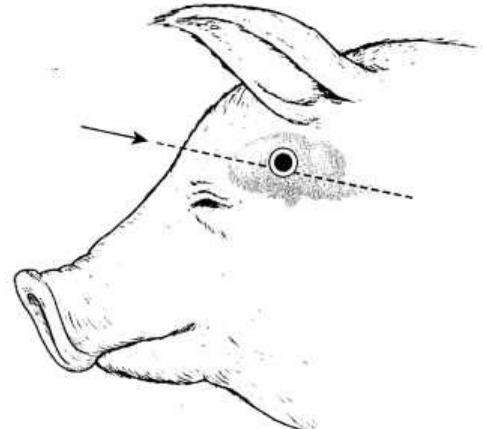


Humane Trapping and Disposal

Although wild swine are nuisance species, they are living animals that register pain and stress. Steps should be taken to minimize stress and insure they are disposed of humanely.

Humane Trapping

- Traps should be checked at least once daily and placed somewhere with shelter or shade.
- Traps should be constructed to minimize injury, smaller mesh size should be used to avoid snout injury. 4" by 4" is the minimum recommendation.
- Traps should be secured so that swine cannot lift the trap.



Humane Disposal

- Swine should be disposed of quickly.
- Shooter should approach the trap quietly to avoid panicking the trapped swine.
- Swine can be disposed of using a .22 caliber rifle or larger.
- Do not insert the rifle barrel into the trap, through the side panels. Swine may charge and hit barrel, potentially causing you or someone else injury.
- Instead shoot through the panels or down into the trap.

- A brain shot will insure a quick, humane death.
- Frontal shot should be placed about 2-3" above an imaginary line directly between the eyes.
- Midpoint shot should be placed at an imaginary line between the eye and ear.
- Careful not to shoot directly between the eyes as this is the beginning of the nasal cavity and will not result in a rapid death.

Trap Gates

There are many variations in design and materials used for trap gates. Most are made from steel. Choosing the type of gate to use depends on budget, ease of transport and trap being used.

Four Basic Gate Types

- 1) **Drop Gate or Guillotine** - Inexpensive and easily constructed. Gate is suspended by trigger line, once triggered gate will drop close. Single-catch only.
- 2) **Swing or Saloon Gate** - pivot towards inside and held with a trigger line. Once triggered heavy springs close gate quickly. Can be noisy and frighten other swine. Multi-catch.
- 3) **Rooter or Lift Gate** - hinged top of gate allows one way entry into trap. Can also be set open and then drop close with a trigger. Can be noisy and frighten other swine. Multi-catch.
- 4) **Funnel Entry** - ends of mesh panel constructed as funnel in which swine must push through to enter trap. Tynes on edge of mesh panel entry prevent swine from pushing back out. Quiet closure.

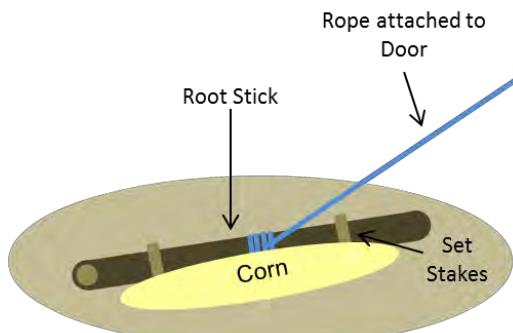


Trigger Mechanisms

Two major types of trigger mechanisms are used when trapping wild swine: the root stick and the trip wire. For both the trigger pulls a line which causes the gate to fall or swing close.

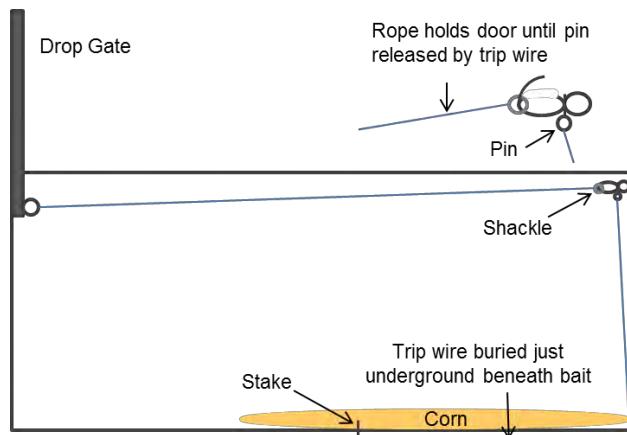
Root Sticks

- A stick is wedged beneath two holding stakes in or around a bait pile. The stick is triggered when the swine feed and root around, pushing the root stick out from under the holding stakes.
- Stick is holding weight of gate so swine must push weight of gate to dislodge stick or a pin can be used.



Trip Wire

- A line or wire buried under bait or suspended slightly above the ground attached to a triggering device (pin or shackle) that will release the gate when pressure is exerted on the line.
- Many different designs.
- Design below by USDA Wildlife Services



New Technology Traps

The newest trap designs utilize a remote triggered gate and cameras that allow you to wirelessly monitor your traps to catch entire sounders or large groups of wild swine.

Below are several examples of vendors and their products. We do not endorse any particular vendor or trap type.

Jager Pro M.I.N.E™ Trapping System

(Manually Initiated Nuisance Elimination)

- Utilizes a large corral trap (35' diameter), an automatic feeder, a 8' gate closed by a remote control device and a wireless camera that can send pictures and videos to your smartphone or email.
- Gate and camera = Approx \$2,125
- Entire M.I.N.E. Trapping system = Approx. \$3,350
- Distributed by Florida Feral Hog Control, Plant City
- www.jagerpro.com and www.floridaferalhogcontrol.com
- Check out the display under the sponsor tent!



BoarBuster - The Samuel Roberts Noble Foundation

- Utilizes a large suspended corral trap that is remotely triggered to drop over the entire group of swine. Trap is monitored with a wireless camera that can send pictures and videos to your smartphone or email.
- BoarBuster Trapping system = Approx. \$5,995
- Distributed by W-W Livestocks Systems
- First come first serve for pre-orders expected to begin delivery June 1st
- www.noble.org/Global/boarbuster

More resources for Swine Trapping Information

- A Landowner's Guide for Wild Pig Management: Practical Methods for Wild Pig Control
 - Published by Mississippi State University Extension Service & Alabama Cooperative Extension System
 - Available at www.msucares.com/pubs/publications/p2659.pdf or www.wildpiginfo.msstate.edu
- Managing Wild Pigs: A Technical Guide
 - Published in Human-Wildlife Interactions Monograph
 - Available at berrymaninstitute.org/files/uploads/pdf/managing-feral-pigs.pdf
- Trapping of Feral Pigs
 - Published by Dr. Jim Mitchell of NQ Dry Tropics, AU
 - Available at file:///C:/Users/bwright/Downloads/trapping_of_feral_pigs%20(2).pdf
- Wild Pigs: Biology, Damage, Control Techniques and Management
 - Published by the Savannah River National Laboratory
 - Available at <http://www.wildpigconference.com/pdf/SRNL-Mayer-Biology%20Damage%20Control.pdf>

Modification by an invasive ecosystem engineer shifts a wet prairie to a monotypic stand

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Abstract At the landscape scale, ecosystem engineers are expected to increase species diversity; however, diversity could decline if the ecosystem engineer is over-abundant. Thus, invasive ecosystem engineers are expected to have strong impacts, due to their high abundances and novel disturbances. An invasive ecosystem engineer, the feral swine (*Sus scrofa*), is a species that creates intense soil disturbances, altering soil and plant communities. In this study, we examine the effects of this invasive ecosystem engineer on experimental plant plots that had been protected for over a decade. Feral swine avoided recently burned plots and preferred plots with N addition. Rooted plots shifted from a bunchgrass dominated wet prairie to a monotypic stand of the native, *Lachnanthes caroliana*. Feral swine were also attracted to plots with existing patches of *L. caroliana* suggesting a potential positive feedback between swine activity and *L. caroliana* patch expansion that could result in an alternative state.

Keywords Clonal growth · Disturbance · Ecosystem engineer · Grassland · Invasive species · Plant–animal interactions

Introduction

Ecosystem engineers alter or create habitats and modulate resources resulting in changes in population dynamics, legacy effects, and altered abiotic conditions that often lead to feedbacks and ultimately alternative states (Jones et al. 1994, 1997a, b; Cuddington and Hastings 2004). At the landscape scale, ecosystem engineers are expected to increase species diversity (Jones et al. 1997a, b); however, this positive effect is contingent on the abundance of the ecosystem engineer and if engineered habitat becomes dominant species richness will decline (Jones et al. 1994; Wright et al. 2004). Thus, invasive species that are ecosystem engineers are expected to have strong impacts, due to their high abundances and novel disturbances (Crooks 2002). In particular, ungulates and omnivores that create large physical disturbances alter ecosystems in various ways; through consumption of plant material and through activities such as belowground foraging or wallowing which alter soil characteristics and potentially hydrology (Rogers et al. 2001; Trager et al. 2004; Anderson and Rosemond 2007). Invasive animals such as feral swine, sheep, goats, and rabbits are considered agents of disturbance and have marked ecosystem-level effects (Lockwood et al. 2007; Klinder 2007).

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Feral swine are an invasive species that is rapidly expanding worldwide and are viewed by some as the most supreme vertebrate modifier of plant communities (Bratton 1975). Feral swine obtain a considerable portion of their diet by rooting (Bratton et al. 1982; Baber and Coblenz 1987; Hone 1988), which involves breaking through the surface layer of vegetation at depths between 5 and 15 cm and excavating the detected food item (Genov 1981; Risch et al. 2010). Several factors determine where and when feral swine root. High food availability drives rooting; for example they feed in oak stands when acorns are available, but after exhausting acorn supply, move to swamp and marsh margins to feed on grasses, sedges, tubers, and roots (Wood and Roark 1980; Doupé et al. 2010). Additionally, it has been noted that feral swine are attracted to tall vegetation (Klinger 2007). Large soil disturbances in the matrix of vegetation create opportunities for annuals and invasive plants (Cushman et al. 2004; Barrios-Garcia and Simberloff 2013). As feral swine populations increase throughout the world, more ecosystems are subjected to this novel disturbance regime. Understanding how widespread soil tilling by an invasive animal impacts structure and function of ecosystems is a critical need (Barrios-Garcia and Simberloff 2013).

The responses of plant communities to disturbance are a function of life history characteristics of the residing species, the pool of available propagules, and rates of colonization. Ruderal plants are expected to respond positively to disturbance (Grime 1979). Kotanen (1995) found annuals proliferated within 1 year after boar rooting in a Californian prairie and large numbers of small sized, wind-dispersed seeds of annuals from the seed rain rapidly established in rooted soil in Sweden (Welander 1995). Other studies suggest that increased abundances of both non-native and native species are associated with disturbance by swine (Aplet et al. 1991; Cushman et al. 2004; Barrios-Garcia and Simberloff 2013). In addition, plants capable of rapid clonal growth could be positively affected by disturbance if they can quickly monopolize the resulting open space and freed resources (Gagnon and Platt 2008). Palacio et al. (2013) found that wild boar disturbance in the Spanish Pyrenees enhanced the prevalence of species propagating from vegetative structures (i.e., bulbs, rhizomes, stolons) and that rooting increased the size and nutrient content of bulbs. Mechanisms leading to enhanced prevalence

of species with vegetative propagation in areas disturbed by herbivores are largely unexplored. Possible pathways include abiotic changes as a result of rooting, such as increased available nitrogen (Bueno et al. 2013), or the spread and mechanical break-up of vegetative structures, or a combination. In the case of rapid clonal growth, multiple disturbances are often required for a species to form dense, monotypic stands (Paine et al. 1998; Kercher and Zedler 2004; Gagnon and Platt 2008).

In this study, we examine the effects of feral swine rooting on long-term experimental plots located in wet prairie and originally designed to examine effects of season of burn and nutrient addition. The plots had been protected from swine rooting for 10 years prior to being intensely rooted. This is one of the few studies of feral swine impacts on a plant community which contains pre-condition plant composition prior to feral swine rooting in a site that had not experienced rooting for at least 10 years; most studies compare rooted with intact areas without previous information on the affected area (Barrios-Garcia and Ballari 2012). We had two objectives: (1) Examine the spatial distribution of feral swine rooting across a long-term experiment and assess the preference of swine for particular season of burn or nutrient treatments; and (2) Quantify vegetation shifts in response to feral swine rooting.

Materials and methods

Site description

This study was conducted at the MacArthur Agro-Ecology Research Center (MAERC) at Buck Island Ranch a 4,170-ha commercial cattle ranch with approximately 3,000 cow-calf pairs. The Center is located ~30 km northwest of Lake Okeechobee. Its subtropical climate features distinct wet (May–Oct.) and dry (Nov–Apr) seasons, and an average annual rainfall of approximately 1,300 mm, and an average temperature of 26 °C in July (mid-summer) and 13 °C in January (mid-winter). Most soils are poorly drained, acidic, sandy spodosols and luvisols. The project site was located within poorly drained grassland dominated mostly by C4 native perennial grasses. Dominant species include native bunchgrasses, (e.g., *Andropogon virginicus* L., *Panicum longifolium* Torrey, and *Axonopus fissifolius* (Raddi) Kuhlm.). Common forbs

are *Lachnanthes caroliniana* (Lam.) Dandy, *Eupatorium mohrii* Greene., *Rhexia* L. spp., and *Diodia virginiana* L., and dominant woody dicots (hereafter shrubs) are *Eupatorium capillifolium* (Lam.) Small and *Euthamia graminifolia* (L.) Nutt. var. *hirtipes* (Fernald) C.E.S. Taylor and R.J. Taylor. The site is representative of wet prairie and has never been fertilized or plowed. Prior to the experiment set-up in the year of 2002 the site was utilized for grazing during the winter dry season since 1960. Similarly to the rest of the Buck Island ranch and other ranches in the region, the site area was burned every 2–3 years in the winter to manage forage and suppress woody plants. Most of south-central Florida has been drained by ditching to lower the water table in the rainy season, and this site had a low density of ditches that were dug between the 1940's and 1960's. Feral swine are abundant on the property; 200–400 feral swine were trapped or hunted per year from 2007 to 2012. It is unknown when feral swine were first observed in the region but records exist of feral swine in Florida from homesteads of the nearby Kissimmee River Valley that maintained free range domestic swine from as early as 1840 (Mayer and Brisbin 2008).

Experimental design

In 2002, MAERC initiated a split-plot experiment with season of burn as the whole plot treatment and nutrient addition as the subplot treatment in an area of relatively homogeneous mesic prairie that was previously used as semi-natural pasture. In October 2002, the experiment (105 m by 80 m) was fenced off to prevent cattle grazing and intrusion of deer and pigs. Within the fenced area twelve 20 m × 20 m plots (four rows of three) were created and these plots were each divided into four 10 m × 10 m subplots (Fig. 1). Rows were treated as blocks and burn treatment was randomly assigned within row. Burn treatments included summer burn, winter burn, and control (no burn). Within the three burn treatments a four level nutrient addition treatment was implemented which included a nitrogen (N) addition treatment, phosphorus (P) addition treatment, nitrogen and phosphorus (NP) addition, and no nutrients added (control). Twice a year lane ways were mowed to keep plots defined. Season of burning was found to significantly affect diversity and vegetation composition and nutrient

addition had weak effects (Boughton et al. 2012). Full descriptions of the experimental design and results prior to feral swine rooting are described in Boughton et al. (2012). The long-term nature of this experiment and vegetation monitoring in permanent plots every year serves as a rigorous foundation to examine the effects of feral swine environmental modification on plant communities.

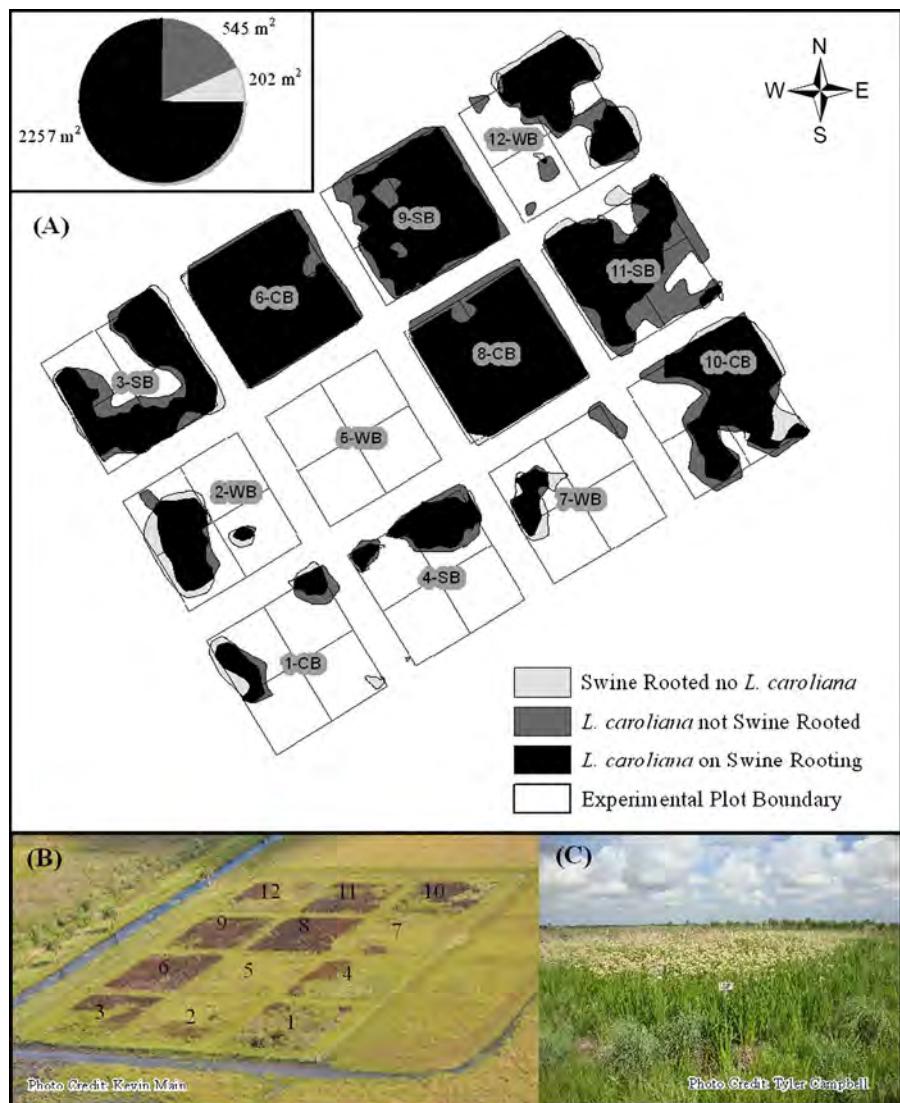
Vegetation composition

Beginning in November 2002, one species composition sampling point was randomly selected within each treatment subplot and permanently marked with an iron post covered with PVC for annual sampling of species composition. The random species composition point represented the center of a circle with an area of 10 m² (10 % of the area of the 10 m × 10 m subplot). At each sampling, individual species percent canopy cover was assigned to one of seven classes of a modified Daubenmire scale (Daubenmire 1968) (1:0–1 %, 2:2–5 %, 3:6–25 %, 4:26–50 %, 5:51–75 %, 6:76–95 %, 7:96–100 %). The 10 m² circle was split into four quarters during cover evaluation to make estimations more accurate. The midpoints of the cover classes (0.5, 3, 15, 37.5, 62.5, 85, and 97.5 %) assigned to each of the 4 quarters were then averaged to produce a species cover value for each species in the 10 m² circle.

Feral swine disturbance

In February 2012, feral swine breached the fence and rooted in over half of the fenced experimental subplots (26 out of 48 10 m × 10 m plots; Fig. 1). Plots were not impacted by feral swine until 2009 and 2010 when a small amount of rooting (<5 m²) occurred in one plot each year (observed and noted by E. Boughton). Following the February 2012 rooting event, we mapped the boundary of each rooted area in March 2012 with a Trimble GPS creating a set of “rooted” polygons. A few months later, we observed within the rooted area a large increase in the plant *Lachnanthes caroliniana*, following which we mapped the boundary of all patches of *L. caroliniana* with a Trimble GPS in July 2012 (Fig. 1.). Where *L. caroliniana* occurred, the density of cover was so high that discerning edges of patches was easily achieved.

Fig. 1 Spatial congruence of *Lachnanthes caroliniana* emergence after damage caused by feral swine (*Sus scrofa*) to experimental plots (a). Aerial view of dark brown seed heads *Lachnanthes caroliniana* taken October 2012, 8 months after feral swine rooting disturbance (b). Dense monoculture of flowering *Lachnanthes caroliniana* taken June 2012 just before mapping (c). Numbers represent the 12 fenced plots and letters burn treatment: winter burn (WB), summer burn (SB), and control or unburned (CB)



Data analysis

To examine whether feral swine targeted plots depending on their fire and nutrient regime, we compared the amount of rooted area derived from our mapped polygons and intersected them with each of the 48 subplots (10 m²). Spatial area of swine rooting was analyzed with linear mixed effects (LME) model. This analysis accounted for random variation due to the plot while analyzing the fixed effects of burn treatment and nutrient addition treatment. Spatial rooting and spatial occurrence of *L. caroliniana* congruence was compared by simple percent congruence.

We assessed community composition of rooted and unrooted plots using non-metric multidimensional scaling (NMS) ordination in PC-ORD v.5.32 with Sørensen distance, a random starting configuration, 50 runs of real data, 100 runs with random data, and 250 iterations. Species composition prior to rooting was included in the ordination to examine how much vegetation composition diverged in rooted areas. The Sørensen distance measure was selected because this measure has been repeatedly shown to be one of the most effective measures of sample similarity (McCune and Grace 2002). A total of 29 species were included in the ordination. Before conducting the

ordination we assessed descriptive statistics in PC-ORD of each plot (rows) and found the coefficient of variation was 15.49 %, indicating no transformations were necessary. We analyzed successional vectors to compare the magnitude and direction of change in rooted plots to unrooted plots following procedures outlined in McCune (1992). Components of vectors analyzed were vector length and vector direction (McCune 1992). The city-block dissimilarity index was used. Multivariate analysis of variance was used to assess vector directions in rooted versus unrooted plots.

We used LME models to examine length of successional vectors (the magnitude of change) and the change in dominant species in unrooted and rooted plots by comparing species composition pre and post rooting. In this analysis, the random factor was plot ($n = 12$) nested within block ($n = 4$). In these analyses the main goal was to determine how feral swine rooting affected vegetation but we had to account for the affect of the experimental treatments. Since the data are non-orthogonal we could not analyze the three way interaction between burn, nutrient addition, and rooting. Therefore, we followed a model simplification procedure in which interactions between factors that were not significant were removed, and the minimal adequate model is presented (Crawley 2007). Since we analyzed five species from the same community we used a Bonferroni correction to derive the appropriate alpha ($\alpha = 0.01$). Models were assessed by visually examining predicted versus standardized residuals. LME and MANOVA were carried out using the R statistical program (R Core Development Team 2010) and non-metric multidimensional scaling ordination was conducted in PC-ORD v.5.

Results

In February 2012, several feral swine breached the fence and rooted in 11 out of 12 plots (Fig. 1) resulting in large changes in plant community composition. Analysis of the spatial distribution of feral swine rooting among the experimental plots suggests that swine tended to avoid winter burn treatment plots (Fig. 2a). Winter burn had less rooting than control or summer burn (respectively, $t\text{-value} = -2.30$, $p = 0.05$, $t\text{-value} = -1.64$, $p = 0.14$). Whereas

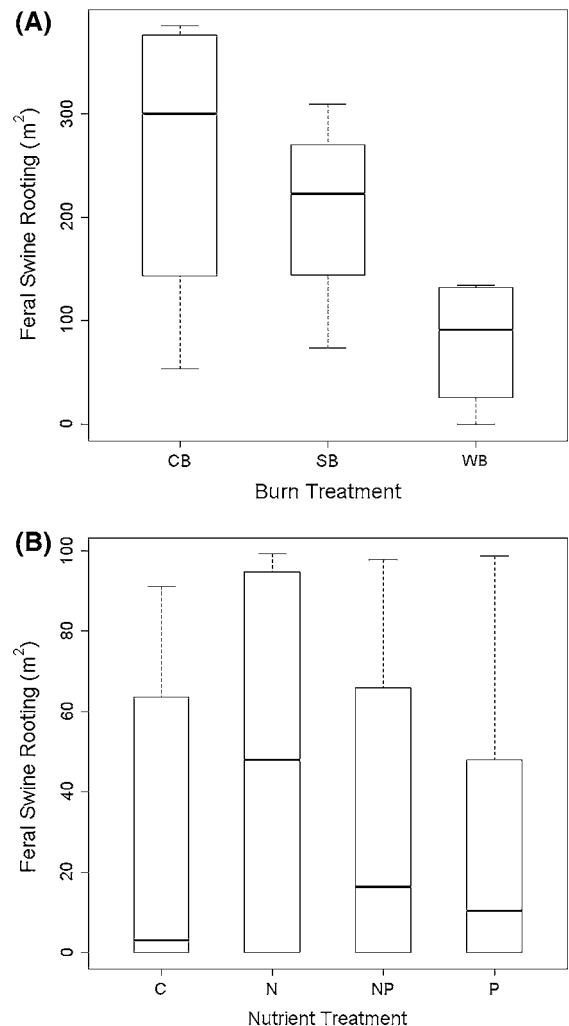


Fig. 2 **a** Feral swine rooting associated with three long term burning treatments, control or unburned (CB), summer burn (SB), winter burn (WB). **b** Feral swine rooting associated with four long term nutrient addition treatments, control—no nutrients (C), nitrogen addition (N), nitrogen and phosphorus (NP), and phosphorus (P). Boxplots show median (black line) and 25th and 75th quartiles (box). Whiskers represent extent of data

summer burn and control did not differ in rooted area ($t\text{-value} = 0.66$, $p = 0.52$). Feral swine preferentially rooted in nitrogen (N) addition plots. The amount of rooting was significantly greater in N plots compared to all other nutrient treatments (Fig. 2b; N vs control: $t\text{-value} = -2.31$, $p = 0.03$; N vs. NP: $t\text{-value} = -2.06$, $p = 0.05$; N vs. P: $t\text{-value} = -3.08$, $p = 0.004$). The amount of rooting in control, NP, and P plots did not differ ($p > 0.05$ for all comparisons).

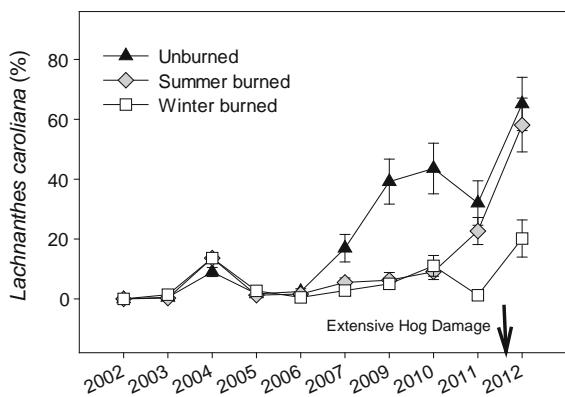


Fig. 3 Treatment responses (mean percent cover and $\pm 95\%$ CI) of *Lachnanthes caroliana* from 2002 to 2012. Treatments are: Control: *unburned*, ungrazed; *Summer burn*, and *Winter burn*, ($n = 4$ replicate plots for each treatment). Prior to swine rooting, *L. caroliana* was increasing in abundance in the unburned control plots starting in 2007. Following swine rooting there was an increase in *L. caroliana* in all treatments. Plots were not sampled in 2008

In plots where rooting occurred, we subsequently observed a large increase in cover and density of the plant, *Lachnanthes caroliana*, a native wetland monocot, in the footprint of the rooted areas. Of the $2,459\text{ m}^2$ rooted by hogs, $2,257\text{ m}^2$ (92 %) was covered by an almost monoculture of *L. caroliana* by July (Fig. 1). Only 202 m^2 (8 %) of rooted area did not become dominated by *L. caroliana* and in all but one case (15.1 m^2 in plot 7) the remaining 545 m^2 of *L. caroliana* mapped was a direct expansion surrounding a *L. caroliana* patch congruent with hog rooting. In 2002, when the project was initiated, all species composition plots had similar, very low levels of *L. caroliana* percent cover (0.013 ± 0.04). After plots were fenced, *L. caroliana* cover remained low in all treatments until 2007 when *L. caroliana* started to increase in percent cover, primarily in the unburned treatment (Fig. 3).

The NMS ordination of all plots ($n = 48$) revealed that plant composition in rooted plots were substantially different from unrooted plots and the prior year's composition (Fig. 4a). A two-dimensional solution to the ordination with low stress (11.18) was found and both axes were significant ($p = 0.02$). The total solution represented 89 % of the variation in the species composition (NMS axis 1 $R^2 = 0.19$, $p = 0.02$; NMS axis 2 $R^2 = 0.70$). Axis 2 represented feral swine rooting disturbance with low values

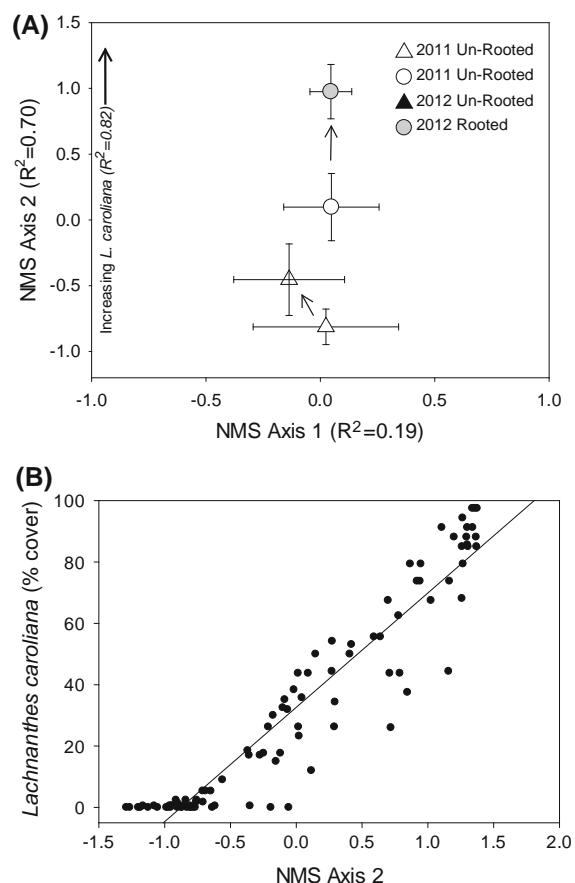


Fig. 4 **a** Non-metric multi-dimensional scaling ordination showed there was differences in species composition among plots that were rooted and unrooted. *Triangles* denote plots that were unrooted in both 2011 and 2012 and species composition shifted little. *Circles* denote plots pre- and post-rooting. **b** *Lachnanthes caroliana* percent cover was highly correlated with axis 2 of the ordination ($R^2 = 0.82$, $p < 0.001$)

corresponding to non-rooted plots and high values corresponding to rooted plots. *L. caroliana* cover was strongly positively associated with Axis 2 (Fig. 4b; $R^2 = 0.82$, $p < 0.001$). Plots that were rooted had higher cover of *L. caroliana* prior to rooting (mean: 32.87, stdev: 24.07) compared to unrooted plots (mean: 3.16, stdev: 8.86) (Fig. 4a).

Species composition in plots that were rooted shifted more compared to non-rooted plots. This was confirmed by analyzing the vectors of individual plots from 2011 to 2012. Vectors were longer in rooted plots in winter and summer burned plots compared to rooted areas in unburned plots (summer burn \times root: $t\text{-value} = 2.35$, $p = 0.03$; winter burn \times root:

Table 1 Summaries of the linear mixed effects analysis of change in dominant species percent cover the year prior to rooting and year after rooting

Dependent	Burn		Nutrients			Rooting
	SB	WB	N	P	NP	
<i>Eupatorium capillifolium</i>	25.77	22.69	4.50	-6.25	-10.36	-5.33
	6.67	5.57	1.01	-1.38	-2.33	-1.54
	<0.001	0.001	0.32	0.18	0.03	0.13
<i>Andropogon virginicus</i>	-0.71	7.37	4.18	9.14	13.89	-3.09
	-0.17	1.67	0.90	1.94	3.00	-0.82
	0.87	0.15	0.37	0.06	0.005	0.42
<i>Panicum longifolium</i>	-22.63	-19.38	1.17	3.88	-2.54	-22.10
	-4.01	-3.26	0.18	0.59	-0.39	-4.36
	0.007	0.02	0.86	0.56	0.70	0.001
<i>Axonopus furcatus</i>	-2.46	-0.38	-0.12	-3.14	0.94	-7.32
	-1.18	-0.17	-0.05	-1.28	0.39	-3.91
	0.28	0.87	0.96	0.21	0.70	<0.001
<i>Lachnanthes caroliana</i>	-3.40	-7.33	1.37	10.03	-1.64	27.61
	-0.50	-1.03	0.17	1.26	-0.21	4.57
	0.63	0.34	0.86	0.22	0.83	<0.001

Values from top to bottom within cells are effect sizes, t-values, and *p*-values. Significant values are bolded. Significance of burn and nutrient treatments are in comparison to control (unburned) and control (no nutrients added), respectively. Significance of Rooting is in comparison to un-rooted plots. SB summer burn, WB winter burn, N nitrogen, P phosphorus, NP nitrogen + phosphorus

t-value = 2.49, *p* = 0.02). Additionally, the directions of vectors of rooted plots compared to non-rooted plots were significantly different (MANOVA, Pillai = 0.22, *F* = 6.28, *p* = 0.004).

Vegetation change was driven by shifts in dominant species cover. In rooted plots, there was on average an additional 41 % cover of *L. caroliana* and a reduction of 20 % cover in *Panicum longifolium* (the dominant bunch grass prior to rooting) and other dominant species (*Axonopus* sp.) whereas there were only slight changes in non-rooted plots (Table 1; Fig. 5). Two of the dominant species (*Eupatorium capillifolium* and *Andropogon virginicus*) were not significantly affected by rooting although the trend was for decreased cover in rooted plots for all species except *L. caroliana* (Table 1).

Discussion

Feral swine are a well-known ecosystem engineer (Bratton 1975; Wood and Barrett 1979; Choquenot and Lukins 1996; Crooks 2002; Engeman et al. 2007; Barrios-Garcia and Ballari 2012), and in many parts of

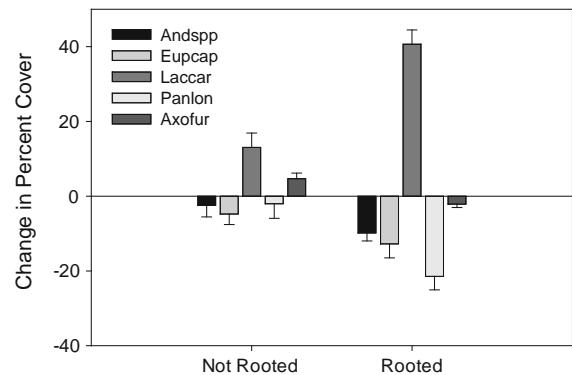


Fig. 5 Change in percent cover of dominant species from 2011 to 2012 after swine-rooting event. In rooted areas, there was on average a 40 % increase in *L. caroliana*. Andsp: *Andropogon* sp., Eupcap: *Eupatorium capillifolium*, Laccar: *Lachnanthes caroliana*, Panlon: *Panicum longifolium*, Axofur: *Axonopus furcatus*

the world, this animal is an invasive introduced species (Cushman et al. 2004; Engeman et al. 2007; Doupé et al. 2010). Intense physical disturbance of the soil by these voracious omnivores has been shown to alter plant communities and typically increase non-

natives and annuals (Aplet et al. 1991; Kotanen 1995; Cushman et al. 2004). However, other studies have found that soil tillage increases richness, evenness, and diversity of wetlands (Kirkman and Sharitz 1994; Arrington et al. 1999). At the patch scale, the effect of feral swine rooting on plant species composition will depend on several factors including species pool and species traits. Although it is well documented that feral swine rooting creates colonization sites for invasive and annual plants, swine rooting effects on perennial species that can spread by clonal fragments has been relatively little studied (but see Palacio et al. 2013). We recorded a substantial increase in cover of a plant species capable of rapid clonal growth which resulted in dense monocultures.

Distribution of rooting among the season of burn and nutrient addition treatments was not uniform; swine were attracted to root in plots with nitrogen addition and tended to avoid plots that were recently burned. That swine were attracted to N plots is not surprising since nitrogen addition resulted in greater %N in plant tissue and thus higher protein content (Newman et al. 2009). In this experiment, plant tissue in N addition plots was on average 0.98 % N (~6.13 % crude protein) compared to 0.73 % N (~4.56 % crude protein) in control (Boughton, unpublished data). Anecdotal observations at MA-ERC also suggest that swine rooting is greater in pastures fertilized with N in the past 12 months (Boughton, personal observation). Some studies have noted a correlation that feral swine rooting results in increased N in rooted patches compared to unrooted areas; however these studies lack pre-condition soil nutrients (Siemann et al. 2009; Cuevas et al. 2012; Wirthner et al. 2012). Alternatively, the finding that increased soil N is associated with rooting may be due to small scale variation in N in the soil whereby swine seek out resources associated with high N. The causation of high N in rooted patches requires further exploration. The reason feral swine avoided recently burned plots is less clear but could be due to low vegetative cover and/or lower cover of *L. caroliniana*, a plant species that feral swine appear to seek out (see “Discussion” section below).

Species composition plots that had been monitored for a decade prior to feral swine disturbance allowed us to examine vegetation shifts as a response to intense rooting. We found that feral swine disturbance shifted a bunchgrass dominated wet prairie to a near

monoculture of *L. caroliniana*. In rooted plots, *L. caroliniana* increased on average 40 % while the dominant bunch grass, *P. longifolium* was reduced in cover on average by 20 %. Vector analysis showed that rooted plots changed significantly more than non-rooted plots. Gagnon and Platt (2008) suggest that as new stressors become part of a given disturbance regime, species capable of rapid clonal growth, even seemingly innocuous natives could form monotypic stands and generate alternative ecological states. *L. caroliniana* was present in the community prior to rooting and was most abundant in long unburned plots (mean % cover: 32 %) and summer burn plots (mean % cover: 23 %) and very low in cover in winter burn treatments (mean % cover: 1 %). *L. caroliniana* cover did not differ between nutrient addition plots and in general nutrient addition had very weak effects on species composition (Boughton et al. 2012). Interestingly, plots that were rooted had higher cover of *L. caroliniana* prior to rooting compared to unrooted plots (see Fig. 4a). This suggests that either swine utilize *L. caroliniana* rhizomes as a food source or are attracted to a food source or condition (i.e. moister soils) associated with *L. caroliniana*. Regardless of the reason that swine targeted plots with *L. caroliniana*, after rooting, *L. caroliniana* cover increased fourfold suggesting there was a positive effect of swine activity on *L. caroliniana*. This resulted in patch expansion of *L. caroliniana* potentially by the spread of rhizome fragments which re-sprout vigorously (Griffith and Boughton unpublished). Similarly, perennial bulbs, even though a preferred food item of wild boar, remain abundant in most European meadows because several bulb species produce large numbers of tiny vegetative bulblets which are dispersed by rooting but not consumed (Barrett 1978).

Expanding patches of *L. caroliniana* due to feral swine activity could increasingly attract swine to root in these patches. This is a potential positive feedback where the effects of feral swine on *L. caroliniana* causes a reciprocating effect of *L. caroliniana* on swine activity which further amplifies the effect of the swine on *L. caroliniana*. If such a strong positive feedback exists, it could result in an alternative state with bunchgrasses replaced by monotypic stands of *L. caroliniana*. As feral swine increase on the landscape, patch expansion of vigorous clonal plants such as *L. caroliniana* in the matrix of bunchgrass dominated prairie could increase beta diversity at the landscape scale or, in contrast, result in

increasing homogenization of the wet prairie ecosystem. The outcome is dependent on the activity and density of the invasive engineer. A key adaptation of feral swine is their enormous reproductive potential, the highest of all free-ranging, large mammals in the United States (Wood and Barrett 1979). Feral swine show an ability to use diverse foods and habitats, high intelligence and wariness, and a resilience to control efforts, which together with high fecundity have allowed populations to grow and disperse rapidly (Choquenot and Lukins 1996; Sweeney and Sweeney 1982). Feral swine population estimate from 10 years ago suggest a total size of ~5 million with the largest populations occurring in California, Florida, Hawaii, and Texas (Pimentel et al. 2005). The population now is undoubtedly much larger and knowledge of ecosystem-feral swine feedbacks will increase our understanding of impacts that invasive ecosystem engineers can have on abiotic and biotic heterogeneity (Paine et al. 1998).

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Facts about Wildlife Diseases: Pseudorabies¹

Samantha Wisely²

What is pseudorabies?

Pseudorabies primarily affects swine; however, cattle, sheep and other mammals are susceptible to infection. Humans are not at risk of contracting pseudorabies. The superficial symptoms of this viral disease (disorientation, foaming at the mouth, and convulsions or tremors) resemble rabies symptoms, thus the name pseudorabies. (The disease is sometimes called “Mad Itch” because infected cattle and sheep will rub against objects to relieve the itching sensation on the skin.) Like rabies, pseudorabies is a viral disease, but it is caused by a different virus, one that is related to the human herpes virus. In addition to neurological signs, animals may show respiratory distress or infection of the reproductive system. The disease is often fatal in piglets, but weaned pigs, juveniles, and adults typically recover and survive after 7 to 10 days of illness (Murphy et al. 1999). Once infected, pigs become carriers of the virus throughout their lives and continue to shed the virus when stressed (USDA 2008). Pseudorabies (abbreviated PRV) is also called Aujesky’s disease and is named for the man who first described the disease in dogs, cats, and cattle in 1903.

Pseudorabies in feral and domestic swine

Once a commercial vaccine was developed, a nationwide control effort eliminated pseudorabies from the US commercial swine industry in 2004. It remains a common disease in commercial piggeries globally, however. Before the elimination of pseudorabies from commercial operations, the US economy lost \$34 billion annually to control

efforts and lost revenue (Ministry of Supply and Services Canada 1988). Current vaccines for swine are considered both safe and effective.

Although the disease was eliminated in commercial animals, feral swine populations in the United States continue to circulate pseudorabies and provide a reservoir for outbreaks. Texas, Oklahoma, Florida, and Hawaii all have dense populations of feral swine with a high prevalence of pseudorabies (Figure 1). Feral swine, therefore, pose a serious risk to commercial swine operations, livestock, companion animals, and wildlife.

Who is at risk for contracting pseudorabies?

Humans are not susceptible to contracting the pseudorabies virus.

Along with swine, cattle and sheep are susceptible to pseudorabies (Figure 2), and the disease is fatal to these animals. Once a cow or sheep is infected, it takes 2 to 5 days for symptoms to develop, and once more severe neurological, respiratory, and reproductive symptoms occur, infected livestock die within 1 to 2 days (Callan and Van Metre 2004). Sporadic outbreaks of pseudorabies occur in cattle, particularly when they are co-mingled with swine (Beasley et al. 1980). The virus is passed directly via nose to nose contact and indirectly via contact with urine or feces. The virus can live for up to two weeks in the environment.

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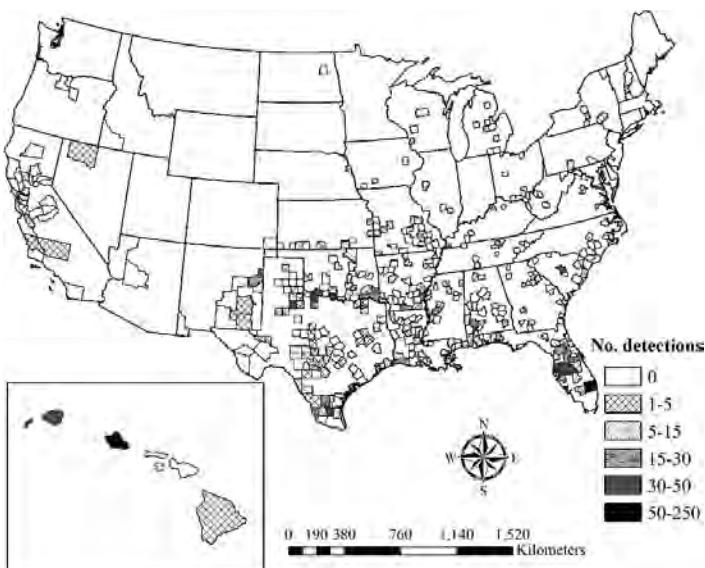


Figure 1. Seroprevalence of pseudorabies in feral swine from 2009–2012 in the United States.

Credits: Pedersen et al. 2013. Reproduced with permission from the *Journal of Wildlife Diseases*.



Figure 2. Remotely triggered camera picture of feral swine and cattle near feed tubs on a ranch in south central Florida.

Credits: Courtesy of the author.

Pseudorabies is a fatal disease in dogs and cats. Symptoms are similar to rabies including excessive salivation, scratching that can lead to self-mutilation, and a lack of coordination or paralysis, but animals infected with pseudorabies will not display an aggressive behavior as do rabid animals (Thiry et al. 2013). There is no vaccine to prevent pseudorabies in cats or dogs.

In the United States, cats and dogs become exposed to the virus when fed raw meat or offal from infected feral swine. Dogs used to hunt feral swine have additional risks because they may also become infected if they come into contact with live feral swine, or feral swine carcasses, gut piles, or feces.

Wild carnivores are susceptible to pseudorabies, and death from pseudorabies has been documented in European brown bears (Zanin et al. 1997), wolves (Verpoest et al. 2014), raccoons (reviewed in Thawley and Wright 1982), Florida panthers (Maehr et al. 1991, Glass et al. 1994), and coyotes (Raymond et al. 1997). Carnivores are considered a dead-end host to pseudorabies, i.e. the disease does not persist and circulate in populations of carnivores because animals succumb to the disease so rapidly that they rarely transmit the disease. Most documented deaths in wildlife come from captive studies where animals have been fed infected pork. More work is needed to understand the risk of pseudorabies to carnivore populations in the wild. Wildlife become exposed to pseudorabies when they prey on feral swine (adults or piglets) or eat the carcasses or gut piles of infected feral swine that are left by hunters or land managers practicing control. There is the potential for indirect transmission of pseudorabies to wildlife from swine urine or feces deposited in the environment.

How can I protect my pets from pseudorabies?

There is currently no vaccine available for cats or dogs; attenuated vaccines (i.e., live virus vaccines) that protect pigs are lethal for cats (Thiry et al. 2013) and dogs. If house cats and dogs are fed meat from feral swine, it should be thoroughly cooked. Commercial pet food is the safest product to feed pets.

Hunting wild hogs with dogs has been a sport for centuries (Figure 3) and is still popular today throughout the United States. Dogs used for hunting feral swine are particularly at risk for contracting and dying from pseudorabies. To reduce the risk of exposure, dog owners should limit contact between dogs and swine and prevent dogs from eating any part of wild pigs, unless the meat is thoroughly cooked.

Pseudorabies in Florida

In the United States, approximately 25% of adult feral swine are seropositive for pseudorabies, meaning that they have been exposed to and are likely carriers of the virus. Florida, however, has a higher-than-average feral swine population and therefore a higher prevalence of pseudorabies (Pedersen et al. 2013). The higher prevalence of pseudorabies in feral swine means a higher risk of exposure and death for Florida livestock, wildlife, and pets.

Evidence suggests that wildlife and companion animals have been impacted by pseudorabies in this state.



Fig. 100.—Sport with Dogs.—“How the Wild Boar is hunted by means of Dogs.”—Fac-simile of a Miniature in the Manuscript of the “*Livre du Roy Modus*” (Fourteenth Century).

Figure 3. Hunting wild pigs with dogs dates back to the 14th century.
Credits: This print, “How the Wild Boar is hunted by means of Dogs,” is from the manuscript *Livre du Roy Modus*. Public domain.

Numerous public hunting areas in Florida allow “hog dogs,” dogs that are trained to track wild hogs. Reports of hog dogs contracting and dying from pseudorabies occur every year throughout the state (e.g. www.promedmail.org, archive no. 20081118.3637), but the magnitude of animal deaths is unknown. The endangered Florida panther is also at risk of death from exposure to pseudorabies. As of 2014, four Florida panthers have been confirmed to have died from pseudorabies. Another 14 are suspected to have died from pseudorabies, but lab results were inconclusive (Glass et al. 1994; M.C. Cunningham, unpublished data).

Decreasing the risk of exposure

Eliminating swine-borne diseases such as pseudorabies from Florida is likely an unrealistic goal given the pervasive nature of feral swine on the Florida landscape. Steps can be taken, however, to reduce disease prevalence and the risk of exposure to pets and livestock:

1. Reduce numbers of swine through animal control, especially on rangelands where livestock are at greatest risk.
2. Keep feral swine away from congregating livestock animals, for example at pens, milking barns, and feed areas.
3. Eliminate translocation of feral swine to reduce spread of diseased animals into populations free of pseudorabies.
4. Do not feed offal or uncooked meat to dogs or other pets.
5. Minimize contact between wild pigs and hunting dogs.

A note to hunters: Although pseudorabies in feral swine does not pose a risk to humans, other diseases carried by pigs such as brucellosis can make people very ill. Wear gloves when handling uncooked meat or other carcass parts of feral swine, and if blood or other bodily fluids come

into contact with your skin or mucous membranes, wash the affected area immediately and contact your doctor. In addition, keep pets away from swine carcasses or live feral swine, and do not feed pets raw meat from feral swine.

This publication is the first in a series on *Wildlife Diseases: Risks to People and Animals*.

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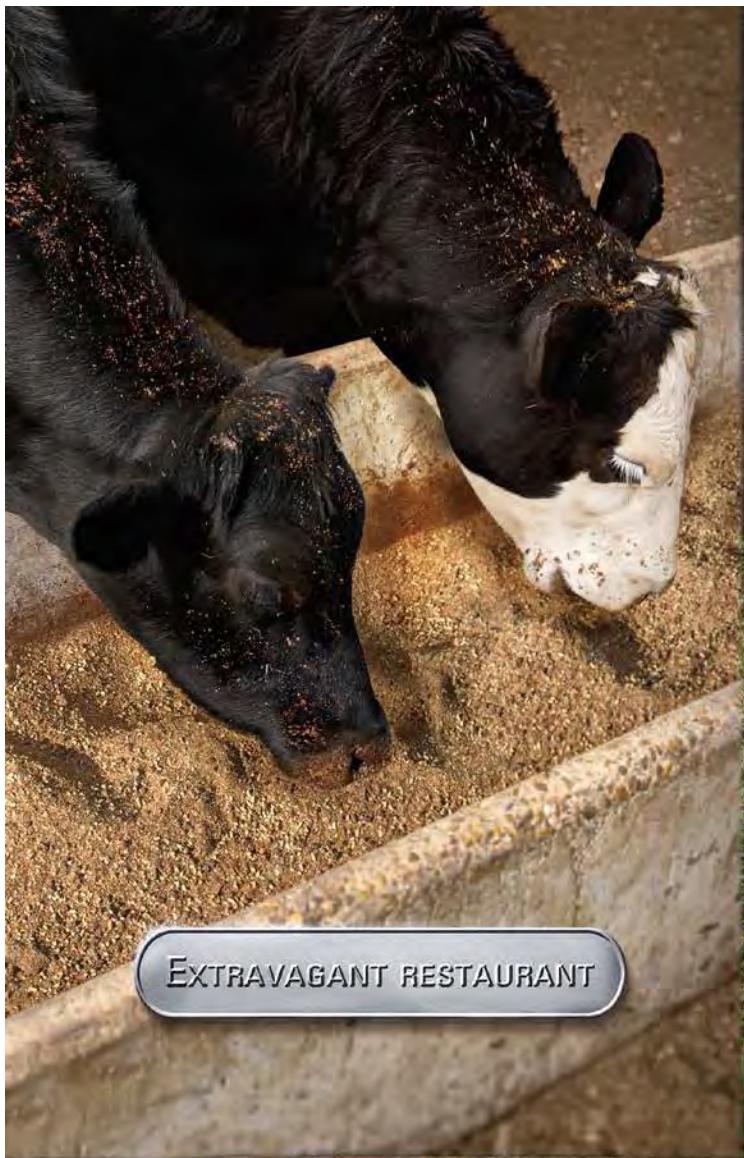
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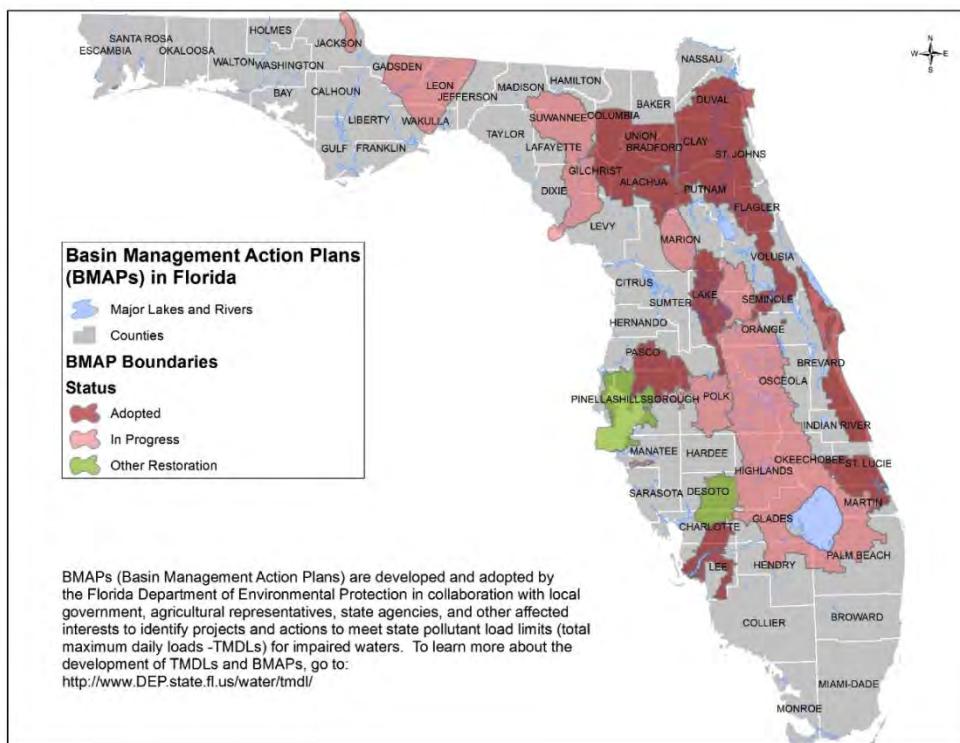
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CAUTION: Federal law restricts this drug to use by or on the order of a licensed
veterinarian.

INDICATIONS FOR USE

LONGRANGE, when administered at the recommended dose volume of 1 mL per 110 lb (50 kg) body weight, is effective in the treatment and control of 20 species and stages of internal and external parasites of cattle:

Gastrointestinal Roundworms	Lungworms
<i>Cooperia oncophora</i> – Adults and L ₄	<i>Dictyocaulus viviparus</i> – Adults
<i>Cooperia punctata</i> – Adults and L ₄	
<i>Cooperia surinabada</i> – Adults and L ₄	Grubs
<i>Haemonchus placei</i> – Adults	<i>Hypoderma bovis</i>
<i>Oesophagostomum radiatum</i> – Adults	
<i>Ostertagia lnyata</i> – Adults	Mites
<i>Ostertagia ostertagi</i> – Adults, L ₄ and inhibited L ₄	<i>Sarcopetes scabiei</i> var. <i>bovis</i>
<i>Trichostrongylus axei</i> – Adults and L ₄	
<i>Trichostrongylus colubriformis</i> – Adults	

Parasites	Durations of Persistent Effectiveness
Gastrointestinal Roundworms	
<i>Cooperia oncophora</i>	100 days
<i>Cooperia punctata</i>	100 days
<i>Haemonchus placei</i>	120 days
<i>Oesophagostomum radiatum</i>	120 days
<i>Ostertagia lnyata</i>	120 days
<i>Ostertagia ostertagi</i>	120 days
<i>Trichostrongylus axei</i>	100 days
Lungworms	
<i>Dictyocaulus viviparus</i>	150 days

DOSAGE AND ADMINISTRATION
LONGRANGE® (eprinomectin) should be given only by subcutaneous injection in front of the shoulder at the recommended dosage level of 1 mg eprinomectin per kg body weight (1 mL per 110 lb body weight).

WARNINGS AND PRECAUTIONS

Withdrawal Periods and Residue Warnings

Animals intended for human consumption must not be slaughtered within 48 days of the last treatment. This drug product is not approved for use in female dairy cattle 20 months of age or older, including dry dairy cows. Use in these cattle may cause drug residues in milk and/or in calves born to these cows. A withdrawal period has not been established for pre-ruminating calves. Do not use in calves to be processed for veal.

Animal Safety Warnings and Precautions

The product is likely to cause tissue damage at the site of injection, including possible granulomas and necrosis. These reactions have disappeared without treatment. Local tissue reaction may result in trim loss of edible tissue at slaughter. Observe cattle for injection site reactions. If injection site reactions are suspected, consult your veterinarian. This product is not for intravenous or intramuscular use. Protect product from light. LONGRANGE® (eprinomectin) has been developed specifically for use in cattle only. This product should not be used in other animal species.

When to Treat Cattle with Grubs

LONGRANGE effectively controls all stages of cattle grubs. However, proper timing of treatment is important. For the most effective results, cattle should be treated as soon as possible after the end of the heel fly (warble fly) season.

Environmental Hazards

Not for use in cattle managed in feedlots or under intensive rotational grazing because the environmental impact has not been evaluated for these scenarios.

Other Warnings: Underdosing and/or subtherapeutic concentrations of extended-release anthelmintic products may encourage the development of parasite resistance. It is recommended that parasite resistance be monitored following the use of any anthelmintic with the use of a fecal egg count reduction test program.

TARGET ANIMAL SAFETY

Clinical studies have demonstrated the wide margin of safety of LONGRANGE® (eprinomectin). Overdosing at 3 to 5 times the recommended dose resulted in a statistically significant reduction in average weight gain when compared to the group tested at label dose. Treatment-related lesions observed in most cattle administered the product included swelling, hyperemia, or necrosis in the subcutaneous tissue of the skin. The administration of LONGRANGE at 3 times the recommended therapeutic dose had no adverse reproductive effects on beef cattle at all stages of breeding or pregnancy or on their calves.

Not for use in bulls, as reproductive safety testing has not been conducted in males intended for breeding or actively breeding. Not for use in calves less than 3 months of age because safety testing has not been conducted in calves less than 3 months of age.

STORAGE

Store at 77° F (25° C) with excursions between 59° and 86° F (15° and 30° C). Protect from light.

Made in Canada.

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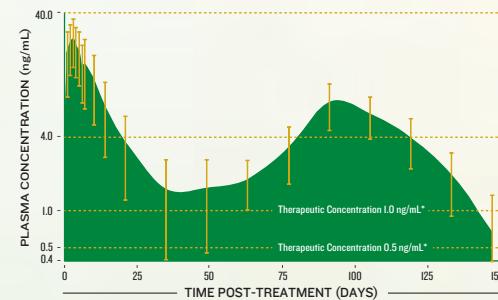
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Pharmacokinetic studies of LONGRANGE in cattle indicate that effective plasma levels remain for an extended period of time (at least 100 days).²

*Plasma concentrations between 0.5 and 1.0 ng/mL would represent the minimal drug level required for optimal nematocidal activity.



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IMPORTANT SAFETY INFORMATION: Do not treat within 48 days of slaughter. Not for use in female dairy cattle 20 months of age or older, including dry dairy cows, or in veal calves. Post-injection site damage (e.g., granulomas, necrosis) can occur. These reactions have disappeared without treatment.

¹ Dependent upon parasite species, as referenced in FOI summary and LONGRANGE product label.

² LONGRANGE product label.

³ Morley FH, Donald AD. Farm management and systems of helminth control. *Vet Parasitol.* 1980;6:105-134.

⁴ Brunson RV. Principles of helminth control. *Vet Parasitol.* 1980;6:185-215.

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