BIOLOGY AND CONTROL OF THE RHODES-GRASS SCALE, ANTONINA GRAMINIS (MASK.)

By

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BIOLOGY AND CONTROL OF THE RHODES-GRASS SCALE,

Antonina graminis (Mask.)

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PREFACE

This thesis is an evaluation of data concerning biology and control of the Rhodes-grass scale carried on at the Lower Rio Grande Experiment Station at Weslaco, Texas.

Rhodes-grass, Chloris gayana Kunth., prior to the year 1942, was the chief pasture grass in the South Texas area. By 1949, no stands remained; presumably, all had been destroyed by Antonina graminis (Mask.), the Rhodes-grass scale.

At this time, the owners of the King Ranch at Kingsville, Texas requested Federal and State aid to study this insect. The Bureau of Entomology and Plant Quarantine, now designated as the Entomology Research Branch, employed Dr. H. L. Chada and the author to initiate an intensive research program into the life history and control of the insect. The State of Texas employed Mr. P. T. Riherd to investigate possibilities of biological control of the scales.

I wish to express my sincere appreciation to my co-workers Dr. H. L. Chada, Mr. P. T. Riherd and other members of the Lower Rio Grande Experiment Station. Grateful acknowledgement is made to Drs. R. G. Dahms, my former adviser and A. M. Schlehuber, Professor of Agronomy and Agronomist in charge of Small Grain Investigation for advice in the construction of this thesis.

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Everett Austin Wood, Jr.

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INTRODUCTION

Antonina graminis (Mask.), the Rhodes-grass scale belongs to the order Homoptera, family Coccidae, sub-family Pseudococcidae. Its hosts are confined to members of the family Gramineae.

The Rhodes-grass scale was first reported in the United States in 1942 from specimens collected at Kingsville, Texas by Mr. Nico Diaz, chief agronomist for the King Ranch, and identified by Dr. Harold Morrison, Entomology Research Branch, Washington, D. C.

The chief host of the scale is Rhodes-grass, Chloris gayana Kunth. (Fig. 1). Prior to 1942, Rhodes-grass was the leading pasture grass in the Kingsville, Texas area. By 1945, 100,000 acres[1] were destroyed, and by 1949, no stands remained; however, heavily infested Rhodes-grass continued to survive along roadsides where moisture was more readily available.

Very little was known about the life-history of this species, and the literature was limited to short notations concerning locations and host plants of the insect.

Due to the unusual morphological characteristics and habits of the scales, it was necessary to devise new biological techniques to complete this study.

Review of the Literature

Antonina graminis (Mask.) was first described as Sphaeroccocus graminis Mask. by W. M. Maskell (1897) from specimens collected in

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[1]Estimated by Mr. Nico Diez

Figure 1. Rhodes-grass, Chloris gayana Kunth. with a typical infestation of Antonina graminis (Mask.). Note excretory filaments. (Reproduced from Texas Agri. Exp. Sta. Circ. No. 116, 1948.)

Fig. 1

Hong Kong, China. Maskell (1898) changed the name to Chaetococcus graminis. E. E. Green (1908) named it Antonina indica from specimens taken from "Hariali" grass in Bengal, India. The present accepted name is Antonina graminis (Mask.), assigned by Dr. Harold Morrison, Entomology Research Branch, Washington, D. C.

The original description in 1897 by W. M. Maskell is as follows:

Sphaeroccocus graminis, sp. nov. (K-1520) on grass, Hong Kong. Adult female dark brown, globular, losing feet and antennae. Abdomen ending in a small depression. Epidermis covered with numerous, circular spinnerets.

E. E. Green (1908) provided a much more accurate description using the name Antonina indica as follows:

Antonina indica (Nov.). Female enclosed in a felted white sac, which fits closely upon the body of the insect but easily removable in a single piece. This covering often becomes yellowish in color, especially after death of the insect. The sac is open at the part covering the rostrum and the posterior extremity from which point a brittle, glistening tubular filament projects. The liquid excreta are carried to the extremity of the tube where they gather in the form of a globular bubble which eventually bursts scattering the liquid in a fine spray.

Adult female dark, purplish brown usually paler on the ventral surface. The stigmatic and genital orifices dusted with a white, mealy powder. Surface smooth. Form sub-ovoid to sub-circular; sometimes narrowed behind, sometimes broadly rounded. The actual extremity is very slightly indented at the anal region but is otherwise evenly rounded without any indication of anal lobes. Terminal segment not demarked.

The derm is first soft and pliant, but the posterior extremity soon becomes densely chitinous and finely rugose. In very old examples, the whole of the derm becomes more or less rigidly chitinized.

Examples from Hakgola (elevation 6,000 feet) are very weakly chitinous. Antennae rudimentary; reduced to two or three joints and a few spiny hairs at the apex. The junctions of the joints are often very indistinct. Spiracles large and conspicuous with a strong, chitinous, cup-shaped orifice, in the sides of which are numerous ceriferous (parastigmatic) pores. Derm with numerous circular pores, of two sizes, intermingled with some minute, spinelike hairs. The pores, which are sometimes (especially in older examples) rendered more conspicuous by a thickened chitinous rim, are more densely covered on the marginal and post-abdominal regions, especially on a transverse zone that lies across the anal pit. Anal ring sunk in a well defined pit, with six stout setae. In the original description of the species, it is stated that the anal setae do not reach the margin of the body; but I find this condition is not constant. In many examples the anal setae project slightly beyond the margin.

Posterior margin of body with a few stout spiniform hairs. Length (under compression) 1.5 to 3.5 mm. Breadth 1.0 to 3.0 mm.

Male not observed in any stage. Newly hatched larvae oblong oval; cream colored, the median area tinged with purple. Very active. At the base of stem and upon rhizomes of various grasses. Usually attended by ants.

P. H. Timberlake (1920) described Anagyrus antoninae Timb., which was found to be the chief parasite of the Rhodes-grass scale in the Hawaiian Islands.

Ramakrishna Ayyar (1921) found the insect infesting grass roots in southern India. Takahashi (1928) discovered an infestation on grass in Formosa and the Philippine Islands. This species was first reported from Japan by Kumana (1932) where it was found at the base of a stem of Imperata arundinacea (Rupr.).

The first report of this species attacking sugar cane came from Van Zwalwenberg (1933), who found the insect among the aerial roots, just above the ground. Phillips (1934) observed ants of the genus Pheidole feeding upon honeydew secreted by the scales. James (1934) collected specimens infesting the roots of Pennisetum clandestinum Hochst and Digitaria abyssinia Stapf. in the Province of Kenya. Schmidt (1937) reported large numbers of bees being attracted to heavy infestations.

An anonymous author (1940) from Queensland, Australia presented the first recommendations concerning control of the scale infesting lawns and bowling greens. The author recommended the use of 1 fluid ounce of nicotine sulphate, 1½ ounces of soap and 2½ gallons of water applied with a sprinkling can so as to thoroughly wet the grass. A large tarpaulin placed over the treated area increased the action of the fumigant. The author also recommended the application of fertilizers to improve the vigor of the grass.

Takahashi (1939) reported the scale from Saipan of the Marianna Island group. Mamet (1943) reported it in Mauritius infesting Cenchrus echinatus L. and Cynodon dactylon (L.) Pers.

Bruner, Scaramuzza, and Otero (1945) were the first to record this species in Cuba where it was found lightly infesting Echinochloa colonum (L.) Link. Potes (1946) discovered specimens in Colombia attacking Panicum purpurascens Raddi. Zimmerman (1948) studying taxonomy and the host range of the scale in Hawaii presented the following information:

> Respiration is accomplished by means of four spiracles located ventrally, two posterior and two anterior. Just posterior to each posterior spiracle is a small, distinct pit-like invagination, which is the outstanding character for determining species. Multilocular disc pores are confined to the midregion of the abdominal venter and to the area about each spiracle, there being none along the margins of the body or on the dorsal side. All trilocular pores are thick walled and the same size.
>
> Host plants mentioned by Zimmerman were: Cynodon dactylon (L.) Pers., Panicum spectabile Nees and Panicum variegatum Hort.

Riherd (1950) was first to attempt biological control of this insect in the United States. Riherd and Chada (1952) observed five species of predators feeding upon the larvae. These included two lady beetles, Hyperaspidius vittigera (Mann) and Hyperaspidius undulata (Say). A small checkered beetle, Hydnocera chapini (Vole) and a brown lacewing, Sympherobius barberi (Bks.) were also observed feeding upon the scale larvae.

Geographical Distribution

Antonina graminis (Mask.) has been reported from the following zoogeographic regions:

1. Australasian - Fiji, Garapan, Hawaii, Palau, Queensland, Saipan and Victoria
2. Ethiopian - Kenya
3. Neartic - Alabama, Florida, Louisiana, Mississippi, Texas and Northern Mexico
4. Neotropical - Colombia, Cuba and Puerto Rico
5. Oriental - Ceylon, Formosa, India, Mauritius, Philippine Islands, South China and Sumatra
6. Paleartic - Japan

METHODS AND MATERIALS

Life History

Reproduction

A large number of adult scales, irrespective of size, was dissected to determine the average number of eggs produced by the Rhodes-grass scale. After removing the cottony covering which envelops the insect's body, the scales were submerged in water contained in a watch glass. A lined paper forming a grid was glued to the bottom of the glass to facilitate counting and a binocular microscope was employed for magnification. The integument of each scale was ruptured, forcing the eggs on to the grid where they could be counted. Egg and larval measurements were also made using an ocular micrometer disc. Eggs, larvae and adults are illustrated in Figs. 2 and 3.

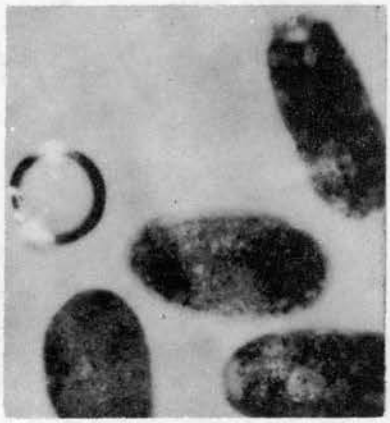
The number of larvae produced by the scales was determined by using the following equipment: a simple cage was constructed consisting of a six-inch length of pipe sealed at one end with black taffeta cloth and at the opposite end with a rubber stopper hollowed to hold a 28 x 83 mm. shell vial (Fig. 4B). The taffeta cloth was used to prevent a condensation of moisture within the cage which was sealed with melted paraffin.

Infested Johnson grass, Sorghum halepense (L.) Pers., containing a known number of scales of various sizes were placed within each cage. Three cages were used for each scale size category, replicated 3 times and labeled small, medium and large. The larvae, being positively phototropic, were attracted to the vials and counted daily.

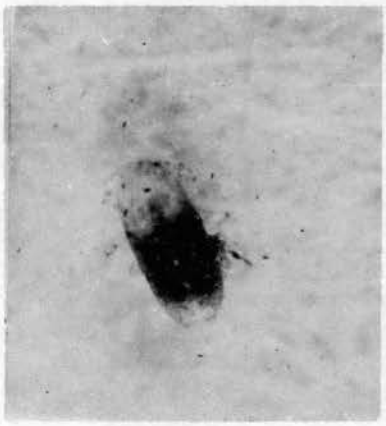
Figure 2. A. Eggs of the Rhodes-grass scale. About 30 times natural size.

B. Scale larva. About 20 times natural size.

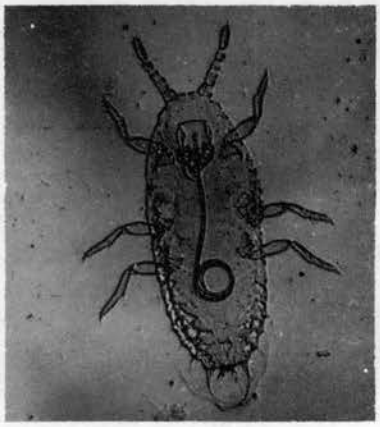
C. Scale larva cleared in Hoyer's solution. Note stylet fascicles withdrawn into the crumena. About 40 times natural size.



A



B



C

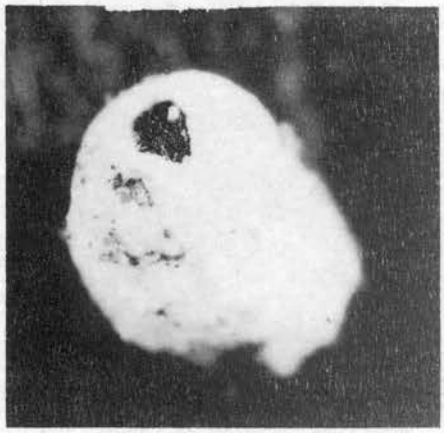
Fig. 2

Figure 3. A. Adult enclosed in cottony envelope. About 30 times natural size.

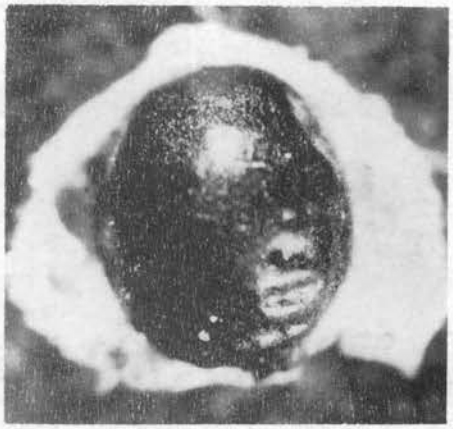
B. Adult with envelope partially removed. About 30 times natural size.

C. Adult cuticula cleared exposing ventral surface. Posterior extremity is densely chitinized. Spiracles are 8 shaped figures. Stylets may be observed as hair-like filaments at anterior end of cuticula. About 40 times natural size.

D. Cluster of Rhodes-grass scales on a node of Johnson grass. Note excretory filament with globular exudation at extremity. About 10 times natural size.



A



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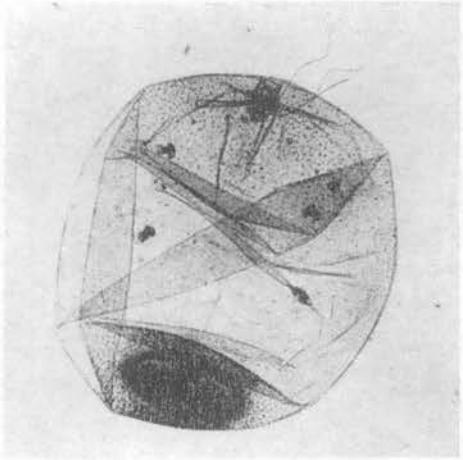

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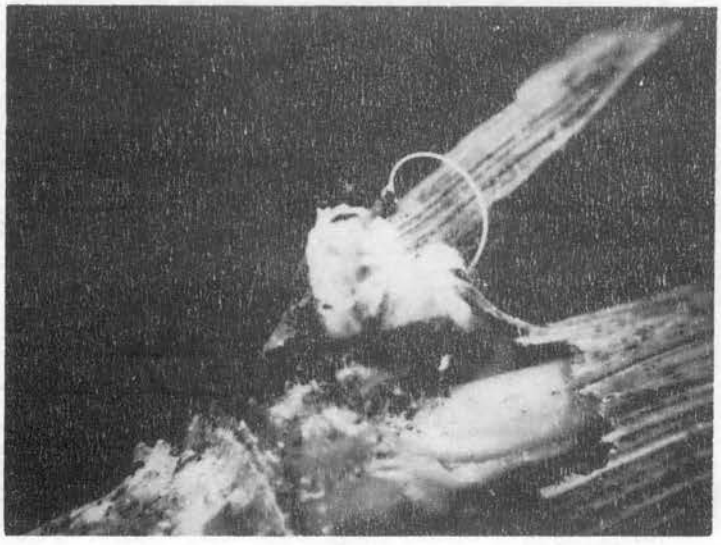


D

Fig. 3

Since this insect usually secretes itself underneath leaf sheaths at the nodes, it was very difficult to confine the larvae to living host plants. Paragrass, Panicum purpurascens Raddi, having large, exposed nodes and being very susceptible to scale attack, was chosen as the host plant for this experiment.

A desirable rearing cage for the scales must be darkened, well ventilated, durable and confine the minute larvae to a limited area. The technique used to construct such a cage was as follows: a 4 x 2 inch section of screen wire was cut with the individual wires bent to form a smooth edge. A hole large enough to encompass a hollowed No. 2 cork was cut in the screen and the whole was rolled into a cylinder and wired together. A section of black taffeta cloth was then fitted over the cylinder and glued into place. A flap was then cut in the cloth exposing the cork and the taffeta cloth. A reproduction of this cage is presented in Fig. 4A.

The cylinder was next lowered over the plant to the desired position. Two circular pieces of taffeta cloth were slit halfway to accomodate the stem and sealed with paraffin to form the bottom and top of the cage. The plant stem was taped at the point of contact to prevent injury to the plant.

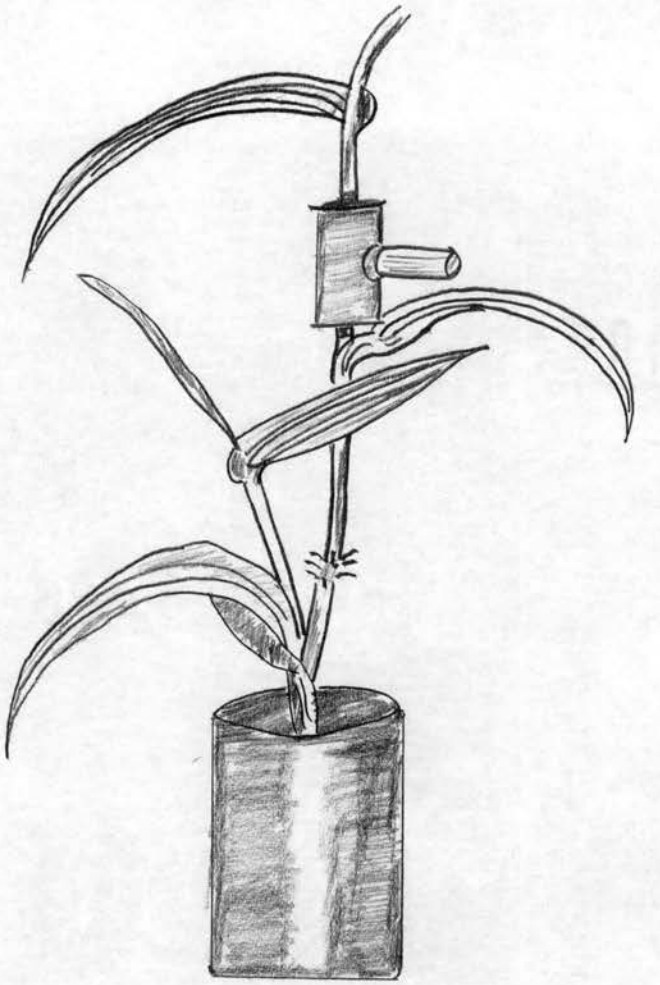
Larvae were introduced into the cage and the opening was plugged. After several days, the plug was removed and replaced with a small vial which was used to collect the progeny.

Ecdysis

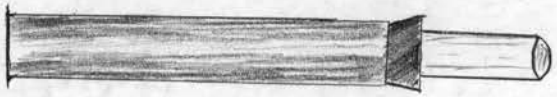
The external protective covering was macerated and the exuvia was separated from the cottony envelope to determine the exact number of exuvia. Attempts to dissolve the protective envelope without

Figure 4. A. Life history cage constructed to confine scale larvae to living host plants.

B. Cage used for fecundity studies.



A



B

Fig. 4

destroying the exuvia were unsuccessful. Chemicals used were: xylene, ether, 10 per cent KOH, acetone and hot, distilled water.

Temperature Effects

Adults were exposed to temperatures of -5.5, -4.0, -2.0, 0.0 and 29.5° C by placing cuttings of infested Johnson grass into a controlled temperature chamber while control specimens were maintained at 29.5° C. The cuttings were removed periodically and placed in shell vials for observation. Since the adult scales present no other readily observed evidence of life, rate of reproduction was used as the criterion of mortality. The number of progeny was counted at intervals and compared numerically with those produced by control specimens. As an additional check concerning mortality, the scales were examined under a binocular microscope several days after reproduction ceased in order to ascertain the condition of the haemocoele. If a dark, viscous haemocoele was evidenced upon rupturing the integument, the specimen was considered dead.

Rearing

Larvae of the scales were incubated in large flour cans (Fig. 5) which were filled with infested grass cuttings, inverted and sealed with paraffin to prevent escape of the larvae. Three holes were cut into the side of the can and 2-inch pipes were welded into the holes. A No. 3, hollow, rubber stopper containing a No. 2 shell vial was fitted over each pipe. The vials were removed at intervals and the larvae used for infesting test plants.

Figure 5. A. Inverted flour can used as an incubator for culturing scale larvae.

B. Cross-section of collecting vial.

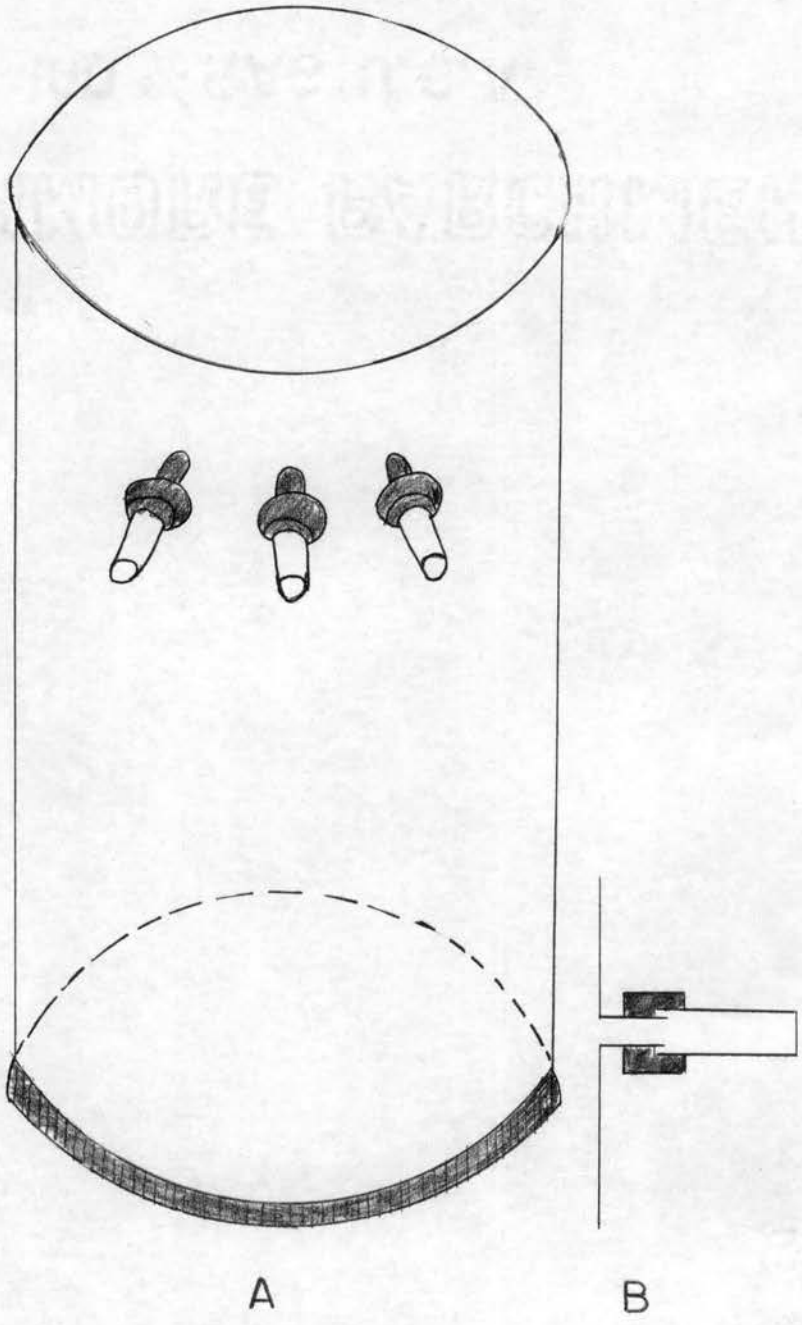


Fig. 5

Host Plants

Periodic surveys were conducted in the Gulf Coast states to determine possible new host plants and the infested areas were mapped as presented in Fig. 6. Each new host plant was identified by local agrostologists or sent to Washington, D. C. for positive identification. Additional grass species were grown in pots and subjected to scale attack to determine their susceptibility.

Control

Chemical

Due to the scale's extremely high biotic potential, an insecticide must be nearly 100 per cent effective and possess residuality. In the laboratory, a few insecticides were screened by spraying infested material until the scales were drenched. The treated specimens were then placed in vials and examined periodically. If the material was effective, it was applied to host material in the field to determine toxicity and phytotoxicity.

Susceptible grasses were grown in pots, flats, and in nutrient solution. These plants were infested with larvae cultured in the incubators and treated with various insecticides after an infestation developed.

In the field, Bermuda-grass lawns and golf greens were used in the majority of the experiments. Square yard areas were staked and treated. A gallon jug, equipped with a watering nozzle, was used for treating small plots. Large areas were treated with a 5-gallon hand sprayer using carbon dioxide gas tubes for compression.

A gravity type, hand pulled sprayer was also developed for use

Figure 6. Known distribution of the Rhodes-grass scale in the United States in 1952.

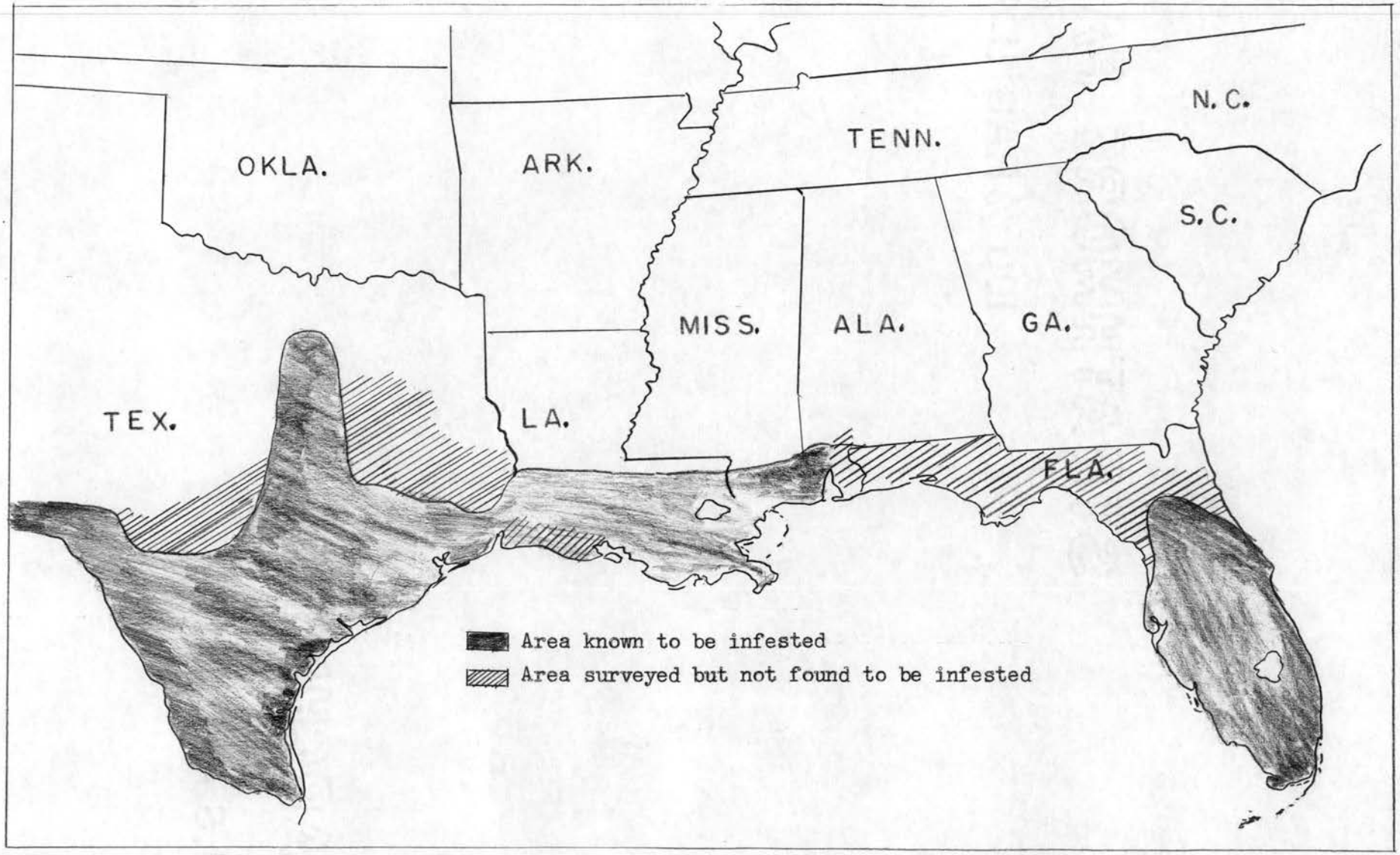

OKLA.
ARK.
TENN.
N. C.
S. C.
MISS.
ALA.
GA.
TEX.
LA.
FLA.
Area known to be infested
Area surveyed but not found to be infested

Fig. 6

on large areas. This sprayer consisted of a 50-gallon tank mounted upon a platform built upon aeroplane wheels. The boom consisted of a length of pipe with 1/8-inch holes drilled every two inches which provided even coverage.

Five 2 by 2 inch samples were taken periodically from each square yard treated and examined throughly under the binocular microscope to determine the per cent of infestation. All scale specimens, both living and dead, were counted. Dried scales, or those exhibiting a thick, viscous haemocoele, were considered dead. Since numerous dead specimens were often found in the check samples, Abbott's formula[1] was used to determine per cent control. The following insecticides were used:

Arsenicals

Milarsenite	As_2O_3 + organic waste material
Sodium arsenite	$Na_3AsO_3.NaAsO_2$

Oils

Humble white oil No. 95 (U.S.P.), X-100 emulsifier

Soltrol 140, Triton E-1956 emulsifier plus .5 per cent DDT

Soltrol 140, Triton, X-100 emulsifier

Organic Phosphates

Compound 3885	S-mercaptoacetyl-urea-O, O-diethyldithiophosphate
Compound 3901	S-mercaptoacetyl-urea-O, O-dimethyldithiophosphate
Demeton	O,O, diethyl-O-ethylmercaptoethyl-thiophosphate

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[1] $\frac{X-Y}{X}$ x 100 = per cent control; X = number alive in check, Y = number alive in treated, and X-Y = number killed by treatment.

Dow C-1014	commercial preparation of octamethylpyro-phosphoramide
Geary E 20/58	0,0-diethyl se-2-(ethylio) ethyl ester of phosphoroselenoic acid
Parathion	0,0-diethyl-0-p-nitrophenyl thiophosphate
Pestox III	Bis (bis-dimethylamino phosphonous) anhydride
Sodium fluoroacetate	Bis [2-(fluoroethoxy) ethoxy] methane

Other

Lime sulphur	
Nicotine sulphate	
Sodium selenate	Na_2SeO_4

Biological

From an economic viewpoint, biological control is the most practical if it can be applied successfully. Anagyrus antoninae Timberlake, a hymenopterous parasite of the family Encyrtidae, was reared from parasitized scale material in the insectary which was maintained at 21° to 24° C. Flats 18x18x6 inches, filled with moist soil, were enclosed by rearing cages 18x18x8 inches and covered with 60-mesh plastic screens. Johnson grass cuttings, heavily infested with scales, were set upright in the soil; the parasites were then released within the cages.

Approximately two weeks after parasitization, the material was placed in emergence boxes 6x6x15 inches and covered with a black cloth. An emergence hole, 1½ inches in diameter, was contained by a hollow cork and a vial 28x83 mm. Being positively phototropic, the parasites were attracted to the vials, where they were collected and used to

maintain a culture. When a large supply of parasitized material was obtained, it was taken to the field in paper sacks where the parasites were released at favorable locations.

Plant Resistance

It was observed during surveys, that a few species of grass were consistently free of scales while other species in the same locale were heavily infested. These resistant species were grown in flats and pots and heavily infested with scale larvae. Daily observations were made comparing these infestations with those of susceptible grasses.

Plots, containing five leading forage grasses in the South Texas region, were located at San Benito, Raymondville, and Mission, Texas. Scale counts, under irrigated and non-irrigated conditions, were made periodically at these locations to establish relative susceptibility.

RESULTS

Life History

In the southern Rio Grande Valley area the scale lived 3½ months and there were approximately five overlapping generations per year. Infestations were first noted during the latter part of February upon the nodes of susceptible grasses and the second generation was well established within two months. The last generation overwintered upon the rhizomes of Johnson grass or other host grasses which were protected from intermittent periods of cold. Those which survived, continued to reproduce when the temperatures are above freezing; however, reproduction was held to a minimum at this time.

Reproduction

The scales were ovoviviparous and parthenogenetic in development. The eggs were developed within the egg tubes and retained within the body of the adult until ready to hatch. Sexual maturity was reached about two months after birth. Each female produce from 150 to 200 eggs which measured an average of 498 μ in length and 183 μ in width. The average larval measurements were 830 μ in length and 235 μ in width. Eggs and larvae are shown in Fig. 2.

Upon maturation, the egg was deposited and hatching occurred immediately. The embryonic membrane was ruptured by the alternate expansion and contraction of the embryo. The head was first freed, followed by the first pair of legs which also aid in the extrication. The larva then escaped the confines of the cottony envelope which was provided with an opening posteriorly and anteriorly.

Dispersion and Feeding

After escaping, the larvae seek the first vertical object and either became established or were blown away by gusts of wind. Many were transported by vehicle of commerce as evidenced by localized infestations along railroad tracks and highways.

Being positively thigmotropic, the larvae secreted themselves beneath the leaf sheath, at nodes of susceptible grasses and began to feed. Feeding was accomplished by means of the long, thread-like stylets which were forced into the phloem of the plant by strong muscles. Once established, the larvae were transformed into sessile, sack-like forms. Being gregarious, colonies of 50 or more individuals clustered around a single node.

Ecdysis

The first molt occured within ten days and was characterized by the loss of all appendages including the cuticular covering of the stylets. Subsequent molts cannot be observed due to the cottony secretion which envelops the insect's body; however, a search of the cottony envelope from adult scales, revealed three cast exuviae embedded in the envelope. The life cycle follows this pattern: adult-egg-first larval instar-second larval instar-third larval instar-adult.

Excretion

Excreta in the form of "honeydew" was eliminated by means of a waxy, hollow filament which may be an inch or more in length (Figs. 1 and 3D). Upon reaching the end of this tube, the exudation formed a spherical, translucent droplet which bursts into a fine spray.

This filament acted as a conveyor to eliminate contamination from the colony, and upon being broken a new one was secreted within a few days. The colonies were often attended by ants and bees which were attracted to the sugary exudations.

Longevity

Without food, the larvae lived approximately 5 days; while the adults continued to exist 4 to 6 weeks upon Johnson grass cuttings. Under normal conditions, the scales lived approximately $3\frac{1}{2}$ months.

Temperature Effects

The Rhodes-grass scale endured high temperatures for limited periods of time; however, continued exposure resulted in desiccation. Fecundity was markedly impaired as temperature and length of exposure was increased. Temperatures of 41.5° C produced 100 per cent mortality within 120 hours. Exposures to below freezing temperatures resulted in a gradual decline of reproductive ability as the temperature decreased and the length of exposure increased (Table 1).

Table 1. The Reaction of the Rhodes-Grass Scale to Temperature Gradients.

Treatment			Number of Larvae Produced					
Degrees Centigrade	Hours exposed	No. of scales in test	Days after Exposure				Avg. per scale	Per cent of check in same test
			7	20	25	Total		
-5.5	1	25	11	0	0	11	0.44	0.35
-5.5	2	32	5	0	0	5	0.16	0.11
-5.5	3	17	1	0	0	1	0.06	0.04
-5.5	4	15	0	0	0	0	0.00	0.00
-4.0	1	12	14	0	36	50	4.17	18.96
-4.0	2	26	0	0	52	52	2.00	9.09
-4.0	3	10	0	0	18	18	1.80	8.18
-4.0	4	8	0	0	10	10	1.25	5.68
-4.0	5	10	0	0	7	7	0.70	3.18
-4.0	6	20	0	0	9	9	0.45	2.04
-2.0	1	22	5	78	-	83	3.77	13.67
-2.0	2	15	0	52	-	52	3.47	12.62
-2.0	3	4	0	12	0	12	3.00	10.91
-2.0	4	5	0	15	0	15	3.00	10.91
-2.0	5	9	0	25	0	25	2.78	10.11
-2.0	6	12	0	32	0	32	2.67	9.71
-2.0	7	14	0	12	0	12	0.86	3.12
-2.0	24	42	6	0	0	6	0.14	0.51
41.5	24	20	-	575	-	575	28.80	50.00
-2.0	48	68	0	0	0	0	00.00	0.00
0.0	48	6	-	32	0	32	5.33	61.63
0.0	49	11	28	0	0	28	2.55	29.65
0.0	50	16	50	0	0	50	3.13	36.39
0.0	51	12	12	0	0	12	1.00	11.63
0.0	65	16	3	0	0	3	0.19	2.20
0.0	66	6	20	0	0	20	3.33	38.48
0.0	67	11	20	0	0	20	1.82	21.16
0.0	68	12	7	0	0	7	0.58	6.74
0.0	69	10	0	0	0	0	0.00	0.00
0.0	72	15	2	0	0	2	0.13	1.51
0.0	90	15	0	0	0	0	0.00	0.00
41.5	96	10	150	-	-	150	15.00	26.00
41.5	120	30	0	0	0	0	0.00	0.00
29.5	-	16	629	815	342	1786	111.60	100.00

Host Plants

The host plants of the Rhodes-grass scale are confined to the family Gramineae and 79 known hosts are listed below:

Scientific Name	Common Name
Sub-family Festucoideae	
Tribe Agrostideae	
Agrostis palustris Huds.	Creeping bentgrass
Sporobolus asper R. Br.	Dropseed
Sporobolus poiretii Hitchc.	Smutgrass
Sporobolus texanus Vasey	Texas dropseed
Tribe Chlorideae	
Bouteloua curtipendula (Michx.) Torr.	Sideoats grama
Bouteloua filiformis (Fourn.) Grif.	Slender grama
Bouteloua hirsuta Lag.	Hairy grama
Buchloe dactyloides (Nutt.) Engelm.	Buffalograss
Chloris ciliata Swartz	Fringed chloris
Chloris cucullata Birch.	Hooded windmillgrass
Chloris gayana Kunth.	Rhodes-grass
Cynodon dactylon (L.) Pers.	Bermuda-grass
Dactyloctenium aegyptium (L.) Beauv.	Crowfootgrass
Eleusine indica (L.) Gaertn.	Goosegrass
Leptochloa filiformis (Lam.) Beauv.	Red Spangletop
Trichloris pluriflora Fourn.	Four flower trichloris
Tribe Festuceae	
Arundo donax L.	Giant reed
Dactylis glomerata L.	Orchardgrass
Eragrostis curvula (Schrad.) Nees	Weeping lovegrass
Eragrostis lehmanniana (Schrad.) Nees	Lehmans lovegrass
Eragrostis reptans (Michx.) Nees	Creeping lovegrass
Eragrostis secundiflora Persl.	Red lovegrass
Eragrostis trichoides (Nutt.) Wood	Sand lovegrass
Festuca arundinacea Schreb.	Alta fescue
Festuca elatior var. *arundinacea* (Schreb.) Winn	Ky. 31 fescue
Pappaphorum bicolor Fourn.	Pink pappasgrass
Triodia ablescens (Vasey) Watt and Standl.	White triodia
Vaseyochloa multinervosa (Vasey) Hitchc.	Texasgrass
Tribe Hordeae	
Agropyron smithii Rydb.	Western wheatgrass

Tribe Phalarideae

Phalaris tuberosa var. stenoptera (Hack) Hitchc.	Hardinggrass

Tribe Zoysieae

Hilaria belangeri (Stued.) Nash	Curly mesquite

Sub-family Panicoideae

Tribe Andropogoneae

Andropogon caucasius Trin.	Caucasian bluestem
Andropogon littoralis Nash	Seacoast bluestem
Andropogon saccharoides Swartz	Silver beardgrass
Andropogon sericeus R. Br.	Silky bluestem
Elyonurus tripsacoides Humb.	Pan American Balsam scale
Imperata arundinacea Cyrilli	
Imperata brevifolia Vasey	Satintail
Saccharum officinarium L.	Sugar cane
Sorghastrum nutans (L.) Nash	Indiangrass
Sorghum halepense (L.) Pers.	Johnsongrass
Sorghum vulgare Pers.	Sorghum
Sorghum vulgare sudanensis (Piper) Hitchc.	Sudangrass
Trachypogon montifari (H.B.K.) Nees	Crinkleawn

Tribe Paniceae

Brachiari ciliatissima (Buckl.) Chase	Fringed signalgrass
Cenchrus abyssinica Stapf.	
Cenchrus echinatus L.	Hedge-hoggrass
Cenchrus pauciflorus Benth	Field sandbur
Cenchrus setigerus Steud.	Birdwoodgrass
Digitaria abyssinia Stapf.	
Digitaria decumbens Stent.	Pangolagrass
Digitaria didactyla Willd.	Wooly fingergrass
Digitaria runyoni Hitchc.	Dune fingergrass
Digitaria sanguinalis (L.) Scop.	Large crabgrass
Echinochloa colonum (L.) Link	Jungle rice
Echinochloa crusgalli (L.)	Barnyardgrass
Eremochloa ophiuroides (Munr.) Hack	Centipedegrass
Panicum antidotale Retz.	Blue panicum
Panicum fasciculatum var. reticulatum (Torr.) Beal	Brown top panicum
Panicum hallii Vasey	Hall's panicum
Panicum maximum Jacq.	Guineagrass
Panicum nodatum Hitchc. and Chase	Sarita panicum
Panicum purpurascens Raddi	Paragrass
Panicum spectabile Nees	
Panicum texanum Buckl.	Texas millet
Panicum torridium Gaud.	Torrid panicgrass

Panicum reptans L.	Sprawling panicum
Panicum variegatum Hort.	Variegated panicum
Panicum virgatum L.	Switchgrass
Paspalum monostachyum Vasey	Sulfdune paspalum
Paspalum plicatulum Michx.	Brownseed paspalum
Pennisetum ciliare (L.) Link	Buffelgrass
Pennisetum clandestinum Hochst.	Kikuyugrass
Rhynchelytrum roseum (Ness) Stapf. and Hubb.	Natalgrass
Setaria sp. Beauv.	Bristlegrass
Setaria geniculata (Lam.) Beauv.	Knotroot bristlegrass
Setaria macrostachya H.B.K.	Plains bristlegrass
Setaria verticellata (L.) Beauv.	Bur bristlegrass
Stenotophrum secundatum (Walt.) Kuntz	St. Augustinegrass

Pasture grasses found to be highly resistant to the Rhodes-grass scale are represented by the following:

Andropogon annulatus Forsk.	Diez bluestem
Andropogon ischaemum L.	K. R. bluestem
Andropogon nodosus Willem.	Angletongrass
Hyperrhenia hirta (L.) Stapf.	African bluestem
Paspalum dilatatum Poir.	Dallisgrass

Control

Chemical

The systemic insecticides consistently gave good control in both laboratory and field work and exhibited no phytotoxicity when applied at recommended rates. All other insecticides either produced a low mortality, were phytotoxic, or possessed no residuality.

Demeton at the rate of one gram of actual material in one thousand ml. of water per square yard became the recommended control for scale infested lawns and golf greens.

A 1000 square foot area of the Llano Grande golf course at Mercedes, Texas was treated with demeton at the recommended rate and reduced the scale population from 15.2 per square inch to 0.25 per square inch after 28 days.

As oil is widely used in the control of many scale insects, it was decided to apply light oils to the Rhodes-grass scale in a series of laboratory test as indicated in Table 2.

Table 2. Laboratory Screening Tests of Emulsified Oils.

Insecticide	Per Cent Concentration	Per Cent control 7 days	Host plant	Amount applied
White oil No. 95 (Humble - USP) X-100 emulsifier	50.0	100.0	Johnson grass	2.3 ml. per plant
	25.0	4.8		
	12.0	0.0		
	5.0	0.0		
	3.0	0.0		
	0.0	0.0		
Soltrol 140 X-100 emulsifier	50.0	100.0	Johnson grass	2.3 ml. per plant
	25.0	0.0		
	12.0	0.0		
	5.0	0.0		
	3.0	0.0		
	0.0	0.0		
25% Soltrol 140 5% DDT Triton E-1956 emulsifier	1.25	77.9	Johnson grass	2.3 ml. per plant
	1.00	64.7		
	0.75	55.8		
	0.50	63.6		
	0.25	63.6		
	0.125	18.6		
	0.00	0.0		

Soltrol 140 when applied at a 50 per cent concentration caused 100 per cent scale mortality; however, at lower concentrations it was ineffective. The same results were obtained with Humble white oil No. 95.

Addition of DDT, to a 25 per cent oil emulsion increased scale mortality; however, oil at these rates was not considered economically feasible for scale control.

In the translocation experiments, summarized in Tables 3 and 4, demeton gave consistently excellent control and exhibited no phytotoxicity.

Table 3. Laboratory translocation Tests of Emulsified Insecticides in Nutrient Solution.[1]

Insecticide	Per cent Concentration	Per cent control 30 days	Residuality	Phytotoxicity
Demeton	.100	100.0	Excellent	None
	.040	100.0	Excellent	None
	.020	100.0	Excellent	None
	.013	100.0	Excellent	None
Geary E 20/58	.1000	100.0	Excellent	None
	.0100	41.9	Poor	None
	.0010	5.5	None	None
	.0001	0.0	None	None
Parathion	.20	76.3	None	Severe
	.10	20.8	None	Moderate
	.01	86.2	None	Slight
Pestox III	.170	100	Excellent	None
	.100	100	Excellent	None
	.010	100	Excellent	None
	.001	62.2	Poor	None
	.0001	49.0	Poor	None

[1]Hyponex: Nitrogen--7 per cent
Phosphoric acid--6 per cent
Water soluble Potash--19 per cent
Chlorine--.05 per cent
Hydroponic Chemical Co., Inc., Copley, Ohio

Residuality was excellent, protecting the plant from subsequent infestations for six weeks or more. The same rates applied to field plots, resulted in a comparative reduction in degree of control.

Geary E 20/58 and Pestox III in nutrient solution were very effective, but were discontinued after causing only minor reductions of

scales in the field. Parathion, in nutrient solution, proved to be highly phytotoxic and appeared to kill only the insects near the base of the plant. It was assumed that these specimens were killed by fumigation. In the field, parathion was ineffective against adults but very effective when applied to the larvae.

Table 4. Laboratory Translocation Tests of Emulsified Insecticides in Flats.

Insecticide	Gms. of actual toxicant per sq. ft.	Ml. of water per sq. ft.	Host plant	Per cent control 30 days	Residual toxicity	Phyto-toxicity
Compound 3885	.100 .010 .001	1000	Johnson grass	89.9 7.2 4.2	Poor None None	None None None
Compound 3901	.100 .010 .001	1000	Johnson grass	58.0 0.0 0.0	Poor None None	None None None
Demeton	.100 .010 .001	1000	Rhodes-grass	99.2 69.5 0.0	Excellent Good None	None None None
Demeton	.200 .100 .010	500	Rhodes-grass	72.5 33.9 0.0	Excellent Good None	None None None
Dow C-1014	.200 .100 .010	500	Rhodes-grass	2.9 2.0 0.0	None None None	None None None
Pestox III	.200 .10 .01	500	Rhodes-grass	21.1 9.7 11.3	Good None None	None None None

Data obtained from treatment of infested grasses grown in flats (Table 4) also demonstrated the superiority of demeton over the other

materials used. Experiments disclosed that control usually is increased with an increased application of water.

Demeton applied to heavily infested golf greens (Table 5) at the recommended rates produced 85 to 100 per cent control and protected these grasses from subsequent infestations for a period of two months. These applications were not economically feasible when applied to pasture grasses.

Applications of nicotine sulphate and lime sulphur were ineffective at very heavy rates even with the additional use of a tarpaulin to increase fumigation action.

Biological

Anagyrus antoninae Timberlake was the most important parasite of the Rhodes-grass scale found in this study. The parasite moved nervously over the plant seeking an adult. Upon finding the host, the parasite inspected it with her antennae, unsheathed the ovipositor and inserted a small, white egg within the haemocoele. Oviposition may occur several times before seeking a new host.

Each female parasite was capable of depositing 40 to 50 eggs during the life span, which was approximately three weeks. The minimum length of time from oviposition to emergence was 15 days at a temperature of 29.5° C, with an average emergence time of 18 days. The maximum time of emergence is 30 days at temperatures below 10° C. Males emerged first and copulation occured soon thereafter. Parthenogenetic reproduction may occur, but results in excusively male progeny.

Experiments, under controlled laboratory conditions, demonstrated that the parasite was very effective in the control of the scale;

Table 5. Field Insecticide Tests Upon Bermuda-grass Golf Greens

Insecticide	Formulation	Gms. of actual toxicant per sq. yd.	Gal. of water per sq. yard	Per cent control 30 days	Residual toxicity	Phyto-toxicity
Demeton	Emulsion	3.63	.50	87.2	Excellent	None
		1.81		89.2	Excellent	None
		0.18		12.9	Poor	None
	Emulsion	1.82	.50	44.3	Good	None
		.73		30.9	Moderate	None
		.36		34.0	Moderate	None
		.24		32.9	Moderate	None
Demeton	Wettable powder	3.63	.50	100.0	Excellent	None
		7.26		100.0	Excellent	None
Dow C-1014	Emuslion	8.72[1]	.48	61.4	Good	None
		4.36[1]	.48	91.6	Good	None
		2.91[2]	.16	87.1	Good	None
		1.45[2]	.16	91.5	Good	None
Lime sulphur	Emulsion	9.1	1.00	00.0	None	None
Milarsenite[3]	Powder	54.5	1.00	98.9	None	Severe
Nicotine sulphate	Emulsion	11.4	2.50	20.9	None	None
Pestox III	Emulsion	.73	.04	0.0	None	None
		.36		73.3	Good	None
		.18		48.8	Moderate	None
		.09		27.4	Poor	None
		.73	.16	26.9	Moderate	None
		.36		44.2	Moderate	None
		.18		42.0	Moderate	None
		.09		40.0	Moderate	None
Soltrol 140	Actual material	14.25		53.6	None	Slight
		6.83		37.7	None	None
		2.85		45.2	None	None
		2.28		42.9	None	None
		1.71		18.7	None	None

[1]Insecticide applied three times at weekly intervals.
[2]Insecticide applied one time.
[3]Three per cent sodium arsenite + milorganite.

however, the environmental conditions of the south Texas area were not conducive to the propagation of this parasite. Removal of any organism from one environment to another always presents difficulties. When subjected to a cool, moist habitat accompanied by an abundance of host material, the parasite flourished. During the period of release, under dryland range conditions, a severe drouth existed which destroyed large acreages of grassland and precluded the possibility of a fair test. Although the parasite was well established in areas favorable for its propagation, it is still a matter of speculation whether or not biological control of the Rhodes-grass scale can be accomplished under dryland range conditions.

Plant Resistance

Perhaps the most satisfactory method of combating any economic insect is by the introduction of resistant varieties. For range control of the Rhodes-grass scale, this method appeared to be the most satisfactory.

Two grasses, Andropogon annulatus Forsk. and Andropogon ischaemum L., have been infested repeatedly with scale larvae without an apparent infestation.

These two grasses are more winter-hardy and apparently more palatable to livestock than Rhodes-grass. It has been observed that cattle will graze patches of these grasses in preference to Rhodes-grass growing in the same field.

Data obtained from grass plots at Mission, San Benito and Raymondville, Texas under irrigated and non-irrigated conditions, indicate that the scales are more numerous on Rhodes-grass under conditions of drouth than under irrigated conditions as indicated in Table 6.

Table 6. Average Number of Rhodes-grass Scales per Stem. Texas

Test plant	Irrigated			Non-irrigated		
	Mission	San Benito	Raymond-ville	Mission	San Benito	Raymond-ville
Chloris gayana Kunth.	4.64	4.80	13.08	1.61	11.88	14.87
Panicum maximum Jacq.	2.18	1.42	3.41	0.11	1.32	1.23
Panicum antidotale Retz.	0.13	0.29	0.69	0.02	0.24	0.70
Andropogon nodosus Willem.	0.00	0.00	0.00	0.00	0.00	0.00
Andropogon ischaemum L.	0.00	0.00	0.00	0.00	0.00	0.00

SUMMARY AND CONCLUSIONS

Antonina graminis (Mask.), the Rhodes-grass scale belongs to the order Homoptera, family Coccidae, sub-family Pseudococeidae. The host range is limited to members of the family Gramineae. The chief host of this insect is Rhodes-grass, Chloris gayana Kunth., which was considered as one of the leading pasture grasses in southern Texas prior to 1942. By 1945, 100,000 acres of Rhodes-grass were destroyed and this loss was attributed mainly to the Rhodes-grass scale.

Life history studies disclosed the life span of this insect was approximately $3\frac{1}{2}$ months with five generations per year. All reproduction was by ovoviviparous parthenogenesis and each scale produced an average of 150 young. This species was found to be thigmotropic and gregarious, living in colonies beneath the protective leaf sheaths of susceptible grasses.

Dispersion was accomplished chiefly by wind, vehicles of commerce, and crawling of the active larval stage.

The long thread-like mandibles and maxillae of all stages of the scale penetrated through the epidermal cells to obtain food from the phloem. Larvae may live up to 5 days and adults up to 6 weeks without food.

The scale molted three times before reaching maturity; the first molt being characterized by the loss of all appendages. Subsequent molts cannot be observed due to the cottony secretion which envelops the insect's body.

Excretory exudations were eliminated by means of a slender, hollow, waxy filament which acted as a conveyor to reduce contamination of the colony. Upon being broken, a new tube was secreted within a few days.

Under favorable conditions the scale endured temperature variations from 41.5° to -4° C and below 0° C for limited periods of time. Continued exposure resulted in a decline of reproductive ability as length of exposure was increased.

Periodic surveys were conducted to determine the distribution and host plants of this insect. Infestations were found to be limited by approximately the 33rd parallel of both the north and south latitudes. Seventy-nine species of grasses were found to be suitable host plants for the scale. Two grasses, Andropogon annulatus Forsk. and Andropogon ischaemum L., were found to be highly resistant.

In order to find a suitable chemical control, two arsenicals, two oils, eight organic phosphates, lime sulphur, nicotine sulphate and sodium selenate were tested. Only the phosphates showed any promise and of these, demeton at 1 gram of actual material per 1000 ml. of water per square yard gave satisfactory control under field conditions.

Chemical control of this scale conditions was not economically feasible, although the use of insecticides on lawns and golf greens proved practical.

A hymenopterous parasite, Anagyrus antoninae Timberlake, was very efficient under controlled conditions; however, under dryland range conditions neither this parasite nor its host survived. The feasibility of biological control under dryland range conditions therefore is still a matter of speculation.

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