Short-term nutritional supplementation of ewes in low body condition affects follicle development due to an increase in glucose and metabolic hormones

C Viñoles1,2, M Forsberg2, G B Martin3, C Cajarville4, J Repetto4 and A Meikle5

1 School of Agriculture ‘Emilia Vigil de Olmos’, Universidad del Trabajo del Uruguay (UTU), Uruguay, 2 Centre for Reproductive Biology, Department of Clinical Chemistry, Swedish University of Agricultural Sciences (SLU), Uppsala, Sweden, 3 School of Animal Biology, University of Western Australia, Perth, Australia, 4 Department of Nutrition, Faculty of Veterinary Medicine, Montevideo, Uruguay and 5 Department of Cellular and Molecular Biology, Faculty of Veterinary Medicine, Lasplaces 1550, Montevideo, Uruguay

Correspondence should be addressed to C Viñoles Gil, Department of Cellular and Molecular Biology, Faculty of Veterinary Medicine, Lasplaces 1550, Montevideo, Uruguay; Email: cvinoles@adinet.com.uy

Abstract

This study tested whether the effects of a short period of nutritional supplementation given to ewes during the luteal phase on follicle development and ovulation rate is associated with an increase in circulating concentrations of FSH, glucose or metabolic hormones. Oestrus was synchronised with two prostaglandin injections given 9 days apart and the supplement consisted of corn grain and soybean meal. Corriedale ewes with low body condition were randomly assigned to 2 groups: the control group (C; n = 10) received a maintenance diet while the short-term supplemented group (STS; n = 10) received double the maintenance diet over days 9 to 14 of the oestrous cycle (day 0 = ovulation). Ovaries were examined daily by ultrasound and blood was sampled three times a day during the inter-ovulatory interval for measuring reproductive and metabolic hormones. On days 9, 11 and 14 of the oestrous cycle, half of the ewes from each group (n = 5) were bled intensively to determine the concentrations of glucose, insulin, IGF-I and leptin. Plasma FSH, progesterone, oestradiol and androstenedione concentrations were similar among groups. Dietary supplementation increased plasma insulin concentrations from the first to the sixth day of supplementation and increased glucose concentrations on the third day, compared with control ewes. Plasma leptin concentrations were higher in STS ewes from the second to the fifth day of supplementation. The pattern of IGF-I concentrations was similar among groups. In STS ewes, the nutritional treatment prolonged the lifespan of the last non-ovulatory follicle, so fewer follicular waves developed during the cycle. In STS ewes, increased concentrations of glucose, insulin and leptin one day before ovulatory wave emergence were associated with increased numbers of follicles growing from 2 to 3 mm and with stimulation of the dominant follicle to grow for a longer period. We suggest that the mechanism by which short-term nutritional supplementation affects follicle development does not involve an increase in FSH concentrations, but may involve responses to increased concentrations of glucose, insulin and leptin, acting directly at the ovarian level. This effect is acute, since concentrations of all three substances decrease after reaching peak values on the third day of supplementation. The status of follicle development at the time of maximum concentrations of glucose and metabolic hormones may be one of the factors that determines whether ovulation rate increases or not.


Introduction

Nutrition is one of the most important factors affecting ovulation rate. For example, supplementation for 4–6 days with lupin grain, a high energy and high protein supplement, is sufficient to increase the ovulation rate in sheep (Gherardi & Lindsay 1982, Oldham & Lindsay 1984, Stewart & Oldham 1986). This effect is not associated with a specific component in the lupin grain but to an increase in energy-yielding nutrients (Teleni et al. 1989b, Downing & Scaramuzzi 1991). To be effective, short-term supplements have to be fed on days 9 to 13 of the oestrous cycle or 6 days before luteolysis (Nottle et al. 1985, 1990, Stewart & Olhdam 1986, Downing et al. 1995). This is the time range when the ovulatory wave emerges (for review see Viñoles 2000). However, the wave-like pattern of follicle development was not clear
when the critical period of supplementation was described, so it was not possible to associate daily follicular growth with the endocrine and metabolic responses to the nutritional treatment.

Data on the effects of nutritional stimuli on follicle-stimulating hormone (FSH) levels during the cycle are equivocal, with reports for (Rhind et al. 1985, Rhind & McNeilly 1986) and against (Findlay & Cumming 1976, Rhind et al. 1989, Xu et al. 1989, Smith & Stewart 1990) the induction of a change. When we used ultrasonography to study the static effect of nutrition on daily follicular development, we found that the increased ovulation rate in ewes with high body condition was associated with increased FSH and decreased oestradiol concentrations during the follicular phase (Viñoles et al. 2002). We suggested that lower oestradiol concentrations inhibited FSH less, thus allowing more gonadotrophin responsive follicles to grow and become selected to ovulate.

An alternative mechanism of the immediate nutritional effect on follicular growth might involve direct actions at the ovarian level of glucose and metabolic hormones, such as insulin-like growth factor I (IGF-I) and leptin, since glucose transporter proteins and specific receptors for these hormones are present in the follicles (Williams et al. 2001, Muñoz-Gutiérrez et al. 2004). Since the final stages of follicle growth are the most sensitive to low FSH levels, but increased FSH concentrations are not a consistent finding, it has been proposed that nutrition changes the ability of gonadotrophin-dependent follicles to use the small amounts of FSH that are available (Scaramuzzi & Radford 1983, Souza et al. 1999). Insulin and IGF-I, the concentrations of which increase after a short-term supplementation, could fulfill this role because they increase responsiveness to gonadotrophins and suppress apoptosis in follicles (Monget & Martin 1997, Poretsky et al. 1999, Scaramuzzi et al. 1999, Williams et al. 2001). Leptin influences whole-body glucose homeostasis and the action of insulin, and its concentrations are sensitive to short-term alterations in food intake (Cunningham et al. 1999, Marie et al. 2001). Insulin stimulates the secretion of leptin by adipocytes and, by promoting lipogenesis, it may indirectly increase leptin production (Poretsky et al. 1999). Metabolic hormones also regulate steroidogenesis: glucose and insulin infused together decrease it, while IGF-I infusion is stimulatory (Scaramuzzi et al. 1999). Leptin antagonizes the stimulatory effect of insulin on theca cell steroidogenesis, ultimately leading to a decrease in oestradiol secretion (Spicer & Francisco 1997).

The aim of the present study was to test whether a 6-day nutritional supplement, applied over days 9 to 14 of the cycle, affected follicle development and ovulation rate through an increase in circulating FSH concentrations, and whether these responses were associated with increases in circulating concentrations of glucose, insulin or leptin.

### Materials and Methods

#### Animals

The experiment was conducted at the experimental farm of the Agronomy Faculty in Cerro Largo, Uruguay (latitude 31.5° S) during the period February to April 2002, using 20 Corriolade ewes aged 4.6 ± 0.2 years (mean ± s.e.m.) that had had one lamb the previous year. They had a body weight of 36.4 ± 0.6 kg and a body condition score of 1.8 ± 0.1 units (1 = emaciated, 5 = obese; Suiter 1994). The body condition of the animals was selected based on the experimental model applied to male sheep to study the effects of nutrition on reproduction (Blache et al. 2002). Body condition and body weight were measured weekly. Ewes were randomly assigned to 2 groups: the control group (C; n = 10) received a maintenance diet, while the short-term supplemented group (STS; n = 10) received double the maintenance diet for 6 days, from days 9 to 14 of the cycle (day 0 = ovulation). During the experimental period, all ewes received the diet in one meal at 1200 h, except during the 6-day supplementation period when STS ewes received two meals of equal amounts at 1200 h and 1300 h. During the experiment, the animals were maintained under 13 h light per day. The mean temperature inside the barn was 20.1 ± 0.5°C, 26.4 ± 0.5°C and 24.8 ± 0.5°C at 0730 h, 1400 h and 1930 h respectively. Four weeks before the beginning of the experiment, ewes were placed in individual pens (dimensions: 1.21 m x 1.51 m) where they were kept. The sequential order of the animals was considered for all procedures.

#### Diets

Ewes were fed a maintenance diet (AFRC 1993) formulated for restricted intake (46 g dry matter (DM) per kg of metabolic body weight; BW0.75) to avoid refusals. The basal diet consisted of 70% hay (Trifolium alexandrinum) and 30% concentrate (80% corn grain and 20% soybean meal). This allocation supplied 6.4 MJ metabolisable energy (ME) and 94 g crude protein (CP) per ewe daily. The ME of the diet was calculated by the equation elaborated by MAAF (1975) and dry matter digestibility (DMD) was calculated by applying the equation described by Oddy et al. (1993). Hay was chopped into a particle size of 2 to 5 cm. The chemical composition of the components of the diet is presented in Table 1.

<table>
<thead>
<tr>
<th>Components</th>
<th>DM (g/kg)</th>
<th>Ash (g/kg)</th>
<th>ADF (g/kg)</th>
<th>CP (g/kg)</th>
<th>Fat (g/kg)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Hay</td>
<td>843</td>
<td>79</td>
<td>408</td>
<td>119</td>
<td>25</td>
</tr>
<tr>
<td>Corn grain</td>
<td>882</td>
<td>15</td>
<td>46</td>
<td>93</td>
<td>36</td>
</tr>
<tr>
<td>Soybean meal</td>
<td>889</td>
<td>63</td>
<td>95</td>
<td>476</td>
<td>17</td>
</tr>
</tbody>
</table>
Water was available to ewes *ad libitum*. Mineral salt (8 g crystalline salt, El Valor, Canelones, Las Brujes, Uruguay) and bicarbonate (2% of total diet) were added to the diet. Refusals were removed at 0700 h of the following day and were low throughout the experimental period (less than 10% for hay and concentrates).

**Experimental design**

A schematic representation of the experimental design is shown in Fig. 1. Oestrus was synchronised with two intramuscular injections of prostaglandin (PG; 30 μg Cloprostenol, Dalmaprost-D, Fatro Laboratory, Montevideo, Uruguay) given 9 days apart. Oestrous behaviour was detected by testing the ewes every 12 h with vasectomised rams from the day of the second PG injection and from day 15 of the next oestrous cycle. Daily ultrasound examinations began on the day of the second PG injection and continued until 7 days after the second ovulation (Fig. 1). Ovulation was detected by the collapse of a large follicle and was considered to be day 0 of the oestrous cycle. During the second period of oestrous detection, ultrasound examinations were increased to every 12 h from oestrous behaviour until the detection of the second ovulation.

**Ultrasound examinations**

Transrectal ultrasound examinations were conducted using a real-time, B-mode scanner (Aloka SSD 500 Echo camera, Overseas Monitor Corp. Ltd, Richmond, BC, Canada) and a rigid 7.5 MHz transducer adapted to be manipulated externally in the rectum (Rubianes *et al.* 1997). The total number, the diameter and the position of all follicles ≥2 mm in diameter and all corpora lutea (CL) were assessed in both ovaries each day. A follicular wave was defined as one or more follicles growing to at least 5 mm in diameter. Groups of follicles emerging within 48 h were regarded as a single follicular wave. The subordinate follicle was defined as a follicle that reached 3 mm and could be followed by ultrasonography for at least 3 days. Morphological characteristics of follicular waves were described based on the following parameters of the largest follicle: day of emergence, maximum diameter, lifespan, interwave interval. These variables have been defined previously (Viñoles *et al.* 2001). The day of wave emergence was the day the largest follicle of the wave was retrospectively identified at 3 mm in diameter. Time of deviation was defined as the beginning of the greatest difference in diameter changes between the two largest follicles, at the examination when the second largest follicle reached its maximum diameter (adapted from Ginther *et al.* 1996). The morphological growing phase was defined as the number of days from emergence to maximum diameter of the largest follicle of each wave, and the morphological dominance phase as the number of days from deviation to the emergence of the next wave. Ovulation rate was determined by counting the number of CL 7 days after ovulation. The predictive value and sensitivity of ultrasonography for evaluating the presence or absence of CL is 100% and 96% respectively. The predictive value of ultrasound for the number of follicles is 98–100% for 2 mm, 4 mm and ≥5 mm diameter follicles except for those of 3 mm diameter (71%). The sensitivity of ultrasonography is high for all sized follicles (90–95%) except those of 2 mm diameter (62%; Viñoles *et al.* 2004).

---

**Figure 1** Schematic representation of the experimental design. PG, injection of a prostaglandin analogue; *, glucose measurements.
Blood sampling for glucose and hormonal measurements

From the day of the second PG injection until the day of the second ovulation, blood was collected in all ewes three times a day into heparinised tubes. Blood was sampled in relation to feeding time (hour 0) at −5 h, 1.5 h and 7 h. To describe acute changes in metabolic hormones and metabolites in relation to feeding, in half of the ewes from each group (n = 5) blood was sampled on days 9, 11 and 14 of the cycle. Samples were taken via an indwelling jugular catheter for a period of 24 h—every hour from 2 h before feeding (Fig. 1). The volume of blood taken each time was 5 ml. Blood was placed in heparinised tubes and maintained on ice until it was centrifuged within 10 min of collection. Plasma was stored at −20°C until assayed.

Progestosterone concentrations were estimated in all samples by a direct solid-phase radioimmunoassay (RIA) using DPC kits (Diagnostic Product Co., Los Angeles, CA, USA) as previously described (Meikle et al. 1997). The RIA had a sensitivity of 0.2 nmol l−1. The intra-assay coefficients of variation for low (3 nmol l−1), medium (24 nmol l−1) and high (50 nmol l−1) controls were 5%, 6% and 3% respectively. The corresponding inter-assay coefficients of variation were 12%, 10% and 4% respectively.

Oestradiol-17β concentrations were determined in all samples by RIA using DPC kits (oestradiol double antibody, KE2D, Diagnostic Product Co.) as previously described (Meikle et al. 1997). The detection limit of the assay was 4 pmol l−1. The intra-assay coefficient of variation was 25% at 5 pmol l−1 and <10% for concentrations between 11 and 180 pmol l−1. The inter-assay coefficients of variations for three control samples were 30% (7 pmol l−1), 11% (33 pmol l−1) and 16% (63 pmol l−1).

Plasma FSH concentrations were determined in samples taken at −5 h and 7 h with respect to feeding by RIA (Viñoles et al. 1999). The sensitivity of the assay was 0.2 μg l−1. The intra-assay coefficient of variation was 9% at 3 μg l−1, 10% at 5 μg l−1 and 9% at 7 μg l−1. The inter-assay coefficients of variations for three control samples were 13% (3 μg l−1), 13% (5 μg l−1) and 14% (7 μg l−1).

Androstenedione concentrations were determined twice a day by RIA (DSL-4200, Diagnostic Systems Laboratories, Inc., Texas, TX, USA) in samples taken at −5 h and 7 h daily, from the day of emergence of the last non-ovulatory wave to ovulation. To increase the sensitivity of the assay, 150 μl tracer and 25 μl antibody were used (50% binding for the 0 standard). On the first day, samples (250 μl) were extracted with 2.5 ml ether and 500 μl of the standard (1725 pmol l−1) was extracted in 4.5 ml ether. A pool of plasma from ewes known to have low (110 pmol l−1), medium (225 pmol l−1) and high (385 pmol l−1) androstenedione concentrations from previous studies were used as quality controls. After extraction, 25 μl PBS were added to each tube and 1000 μl PBS were added to the standards. The samples and standards were incubated overnight at 4°C. On the second day, the standard curve was made by serial dilutions of the standard (1:2). The standard curve ranged from 7 pmol l−1 to 432 pmol l−1. An aliquot of 25 μl from each dilution was used in the standard curve. The antibody and tracer were added to the samples and the standard curve and incubated overnight at room temperature. On the third day, 500 μl precipitating reagent were added to the tubes and, after incubation for 30 min at room temperature, the tubes were centrifuged for 30 min at 4°C. The detection limit of the assay was 16 pmol l−1. The inter-assay coefficients of variation were 7% for the low, 3% for the medium and 9% for the high control. The inter-assay coefficients of variations for the low, medium and high controls were 14%, 6% and 9% respectively.

IGF-I concentrations in plasma were measured in all samples by double-antibody RIA (Gluckman et al. 1983). Interference by binding proteins was minimised by acid-ethanol cryoprecipitation, as validated for ruminant samples (Breier et al. 1991). The samples were assayed as duplicate 100 μl aliquots and the limit of detection was 3.0 ng/ml. Six replicates of two control samples containing 7.7 and 27.7 ng/ml were included in the assay and were used to estimate the intra-assay variation (10.5% and 6.9%).

Leptin was analysed in all samples by RIA using antibodies raised in an emu (Blache et al. 2000). The samples were assayed as duplicate 100 μl aliquots and the limit of detection was 0.1 ng/ml. Six replicates of three control samples containing 1.3, 1.6 and 2.4 ng/ml were included in the assay. The intra-assay coefficients of variation were 6.9%, 7.9% and 10.2%. All the samples were analysed in one assay.

Insulin concentrations were measured in all samples by a direct solid-phase radioimmunoassay (RIA) using DPC kits (Diagnostic Product Co.). According to the manufacturer, the sensitivity of the assay is 8.6 pmol l−1. For low (14 pmol l−1), medium (251 pmol l−1) and high (667 pmol l−1) controls, the intra-assay coefficients of variation were 11%, 5% and 2% and the inter-assay coefficients of variation were 38%, 7% and 4% respectively.

Glucose concentrations were measured in fresh blood from one hour before until six hours after feeding (10 samples each day, Fig. 1) using an Elite glucose meter (Bayer, Montevideo, Uruguay). The normal reading was in the range of 20 to 600 mg/dl l−1.

Statistical analyses

The number of follicular waves and the number of ovulatory follicles in three size classes (≤5 mm, 6 mm and >6 mm) were compared using Fisher’s exact probability test. The morphological characteristics of the largest follicle of the last non-ovulatory wave and the ovulatory wave were compared by analysis of variance using
the mixed procedure of the Statistical Analysis System (SAS Institute Inc., Carg, NC, USA). The model included the fixed effect of treatment and the random effect of ewe within group. For the analysis of repeated measurements, the mixed procedure of the SAS was used. The model included the fixed effects of treatment and day, and their interaction. The covariance structure was modelled using the random effect of ewe within group plus autoregressive order 1, to account for the correlation between sequential measurements within the same animal (Littell et al. 2000). Data with repeated measurements included: (1) body weight, (2) body condition, (3) plasma concentrations of progesterone, FSH, oestradiol, IGF-I and leptin, all of which were analysed considering the period before treatment (day 0 to day 8) as a covariate in the model, and (4) plasma concentrations of glucose, insulin, leptin and IGF-I included the fixed effect of time after feeding in the model. Mean values were compared by the method of Least Square Means. The relationships between increases in FSH concentrations and follicular wave emergence were analysed using a skewness method (Viñoles et al. 2002). Profiles of oestradiol and androstenedione associated with the growing profile of the largest follicle of the last two waves of the interovulatory interval were studied individually. For each ewe and each hormone, the baseline and standard deviation of the baseline value were calculated as the average of the lowest 25% of all values measured. The time at which the concentration of a hormone began to increase was defined as the time when at least two consecutive values increased above the baseline by threefold the standard deviation of the baseline. Factors affecting plasma leptin concentrations were evaluated by multiple regression analysis using a backwards elimination procedure in SAS, using the data obtained during the frequent bleeding. The mean values for concentrations of leptin, glucose, insulin, and IGF-I from 2 to 6 h after feeding (period 1; n = 30), and for leptin, insulin and IGF-I from 7 and 24 h after feeding (period 2; n = 30), were calculated. The dependent variable was plasma lep- tin concentrations and the independent variables included group, day, period and plasma concentrations of glucose, insulin and IGF-I. Since the effects of period and glucose concentrations were not significant in the model, a mean for the 24-h period was calculated for leptin, insulin and IGF-I and included in the final model (n = 30). Data are presented as least square means ± S.E.M. Differences were considered significant if P < 0.05.

Results

Body weight and body condition score

Body condition score and body weight were similar in groups C (1.8 ± 0.1 and 36.3 ± 0.7 kg respectively) and STS (1.9 ± 0.1 and 36.0 ± 0.7 kg respectively) during the period before treatment, but body weight was higher in the STS group (38.3 ± 0.7 kg) than in the C group (36.2 ± 0.7 kg; P < 0.01) during the period after nutritional treatment, perhaps due to the increased ruminal and intestinal content in the supplemented animals.

Follicular measurements

Ewes from the STS group developed fewer follicular waves than ewes from the C group (2.9 ± 0.1 vs 3.4 ± 0.2; P < 0.05). In the STS group, 9 of the 10 ewes developed three follicular waves while the other ewe developed two waves during the cycle. Ewes in the control group developed three (n = 6) and four (n = 4) follicular waves. The mean time of emergence of the last non-ovulatory wave and the ovulatory wave was on days 5.5 ± 0.7 and 11 ± 0.7 for the STS group and on days 5.8 ± 0.7 and 12 ± 0.7 for the C group respectively.

Table 2 shows the morphological characteristics of the last non-ovulatory follicle and the ovulatory follicle for each group. The last non-ovulatory follicle had a longer lifespan in the STS group (Fig. 2 and Table 2). The ovulatory follicle had a longer morphological growing phase and its morphological dominance phase (P = 0.06) and lifespan (P = 0.07) tended to be longer in STS ewes (Fig. 2).

Ovulation rate was similar in the STS (1.1 ± 0.3) and C groups (1.1 ± 0.3). The number of follicles recruited into the ovulatory wave was similar among groups. However, in the STS group, more follicles that had been recruited into the ovulatory wave grew from 2 to 3 mm (2.0 ± 0.4 vs 0.9 ± 0.4) and less follicles grew from 2 to 4 mm (2.0 ± 0.3 vs 3.0 ± 0.3). More ewes had ovulatory follicles of 6 mm (7/11) and > 6 mm (3/11) in the STS group than in the C group (5/11 and 1/11 respectively; P < 0.05).

Progestosterone, FSH, oestradiol and androstenedione concentrations

Figure 3 shows that the concentrations of progesterone, oestradiol and FSH were similar among groups from day 9

Table 2 Morphological characteristics of the last non-ovulatory follicle and the ovulatory follicle (emergence = day 0) in ewes that received a short-term nutritional supplement (STS, n = 10) and in control ewes (n = 10). Results are least squares means ± S.E.M.

<table>
<thead>
<tr>
<th></th>
<th>STS group</th>
<th>Control group</th>
</tr>
</thead>
<tbody>
<tr>
<td>Last non-ovulatory follicle</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Deviation (day)</td>
<td>1.9 ± 0.4</td>
<td>1.5 ± 0.4</td>
</tr>
<tr>
<td>Morphological growing phase (day)</td>
<td>2.8 ± 0.5</td>
<td>3.4 ± 0.5</td>
</tr>
<tr>
<td>Morphological dominance phase (day)</td>
<td>4.8 ± 0.5</td>
<td>3.8 ± 0.5</td>
</tr>
<tr>
<td>Lifespan (days)</td>
<td>10.2 ± 0.6a</td>
<td>7.8 ± 0.6b</td>
</tr>
<tr>
<td>Ovulatory follicle</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Deviation (day)</td>
<td>1.2 ± 0.4</td>
<td>1.8 ± 0.4</td>
</tr>
<tr>
<td>Morphological growing phase (day)</td>
<td>4.5 ± 0.4</td>
<td>3.4 ± 0.4b</td>
</tr>
<tr>
<td>Morphological dominance phase (day)</td>
<td>5.2 ± 0.5</td>
<td>3.7 ± 0.5</td>
</tr>
<tr>
<td>Lifespan (days)</td>
<td>6.0 ± 0.3</td>
<td>5.1 ± 0.3</td>
</tr>
</tbody>
</table>

a,bMeans within rows with different superscripts are significantly different (P < 0.05); a,bMeans within rows with different superscripts tended to be different (P > 0.05).
to day 17 of the oestrous cycle (Fig. 3). Concentrations of FSH reached a maximum \((2.3 \pm 0.2 \mu g l^{-1})\) 1.3 \(\pm 0.2\) days before the emergence of the ovulatory wave, remained high for 1.6 \(\pm 0.2\) days and fell to minimum values 2.5 \(\pm 0.2\) days after wave emergence. Oestradiol concentrations reached a maximum \((19.5 \pm 0.9 pmol l^{-1})\) 4.1 \(\pm 0.3\) days after emergence and remained high for 2.9 \(\pm 0.2\) days. Androstenedione concentrations reached a maximum \((356 \pm 30 pmol l^{-1})\) 3.9 \(\pm 0.4\) days after emergence and remained high for 1.0 \(\pm 0.1\) days.

**Leptin and IGF-I concentrations**

Figure 3 shows that leptin concentrations were higher in STS than in C ewes from day 10 to day 13 of the oestrous cycle \((P < 0.05)\). In both groups, leptin concentrations started to increase on day 9 of the oestrous cycle and then decreased from day 11 to the end of the oestrous cycle.

Plasma leptin concentrations fluctuated with time relative to feeding but there was no clear post-prandial surge (Fig. 4). In the STS group, leptin concentrations were higher on day 11 than on days 9 and 14 while, in the C group, leptin concentrations were lower on day 14 than on days 9 and 11 \((P < 0.05)\), in agreement with the observed decrease in leptin concentrations from day 11 of the cycle (Fig. 3). Factors affecting plasma leptin concentrations were group, day and plasma insulin concentrations \((P < 0.001; r^2 = 0.5; n = 30)\). The effects of period \((-2\) to \(6\) h and \(7\) to \(24\) h after feeding) and glucose concentrations were not significant.

The IGF-I pattern was similar in STS and control ewes (Fig. 3). IGF-I concentrations decreased from days 9 to 11–12 \((P < 0.05)\) and increased from day 14 to day 17 of the oestrous cycle \((P < 0.001)\). The effects of time after feeding, day and their interaction were all significant \((P < 0.001)\). Figure 4 shows that IGF-I concentrations were higher on day 9 than on days 11 and 14 in both STS and control ewes \((P < 0.001)\), in agreement with the pattern observed during the oestrous cycle (Fig. 3).

**Glucose and insulin concentrations**

Glucose concentrations decreased in both groups from \(-1\) h to \(1.5\) h relative to feeding \((P < 0.05)\) and increased gradually from \(2\) to \(5\) h after feeding \((P < 0.001)\) on days 9, 11 and 14 (Fig. 4). In the control group, glucose concentrations were similar on days 9 and 11 but increased on day 14 \((P < 0.001)\). In STS ewes, glucose concentrations on days 11 and 14 were higher than on day 9 \((P < 0.001)\). Glucose concentrations on day 11 were consistently higher in STS than in control ewes (Fig. 4).

Plasma insulin concentrations also changed after feeding and the changes were more pronounced in STS ewes. In the control ewes, insulin concentrations were similar on days 9 and 11 but increased on day 14 \((P < 0.001; Fig. 4)\). In STS ewes, insulin concentrations increased from day 9 to day 11 and decreased on day 14, although the values on day 14 were still higher than those on day 9 \((P < 0.001)\). Plasma insulin concentrations were higher in STS than in C ewes on days 9, 11 and 14 \((P < 0.001; Fig. 4)\), with the greatest difference between groups being observed on day 11.

**Discussion**

Short-term supplementation with corn grain and soybean meal from days 9 to 14 of the oestrous cycle prolonged the lifespan of the last non-ovulatory follicle and thus decreased the number of follicle waves. This effect was not associated with changes in FSH concentrations, but it was correlated with an increase in the circulating concentrations of glucose, insulin and leptin. The short-term supplementation did not affect ovulation rate.

The static effect of nutrition is consistently associated with an increase in ovulation rate (Rhind & McNeilly 1986, Rhind et al. 1989, Xu et al. 1989). We have previously found that ewes in high body condition have a high ovulation rate which is accompanied by high FSH and low oestradiol concentrations during the follicular

---

**Figure 2** Growth profile of the last non-ovulatory follicle (LNOF) and the ovulatory follicle (OF) in ewes that received a short-term nutritional supplement (STS, \(n = 10, \bullet\)) and in ewes fed at maintenance level (\(n = 10, \circ\)). Results are least squares means \(\pm s.e.m.\)

---
phase, compared with ewes in low body condition (Viñoles et al. 2002). However, the effectiveness of the immediate effect of nutrition in increasing ovulation rate is not consistent (Stewart 1990). In the present study, a 6-day nutritional treatment, fed from days 9 to 14 of the oestrous cycle, did not affect ovulation rate. However, a 7-day supplement fed from days 8 to 14 of the oestrous cycle in the same breed and during the same season, increased ovulation rate by 14% (Viñoles 2003). The inconsistent effect of short-term supplementation on ovulation rate suggests that an increase in ovulation rate may depend on follicular status at the beginning of the nutritional treatment, among other factors.

At the beginning of the treatment, the last non-ovulatory follicle was at the end of its growing phase and the supplementation prolonged its lifespan (Fig. 2). This delayed atresia may have been induced by changes in the circulating concentrations of glucose and metabolic hormones. Insulin concentrations increased from the first day of supplementation, with maximum values for glucose and insulin observed on the third day after supplementation commenced, in agreement with Teleni et al. (1989a). Insulin has a direct effect on adipocytes to stimulate leptin secretion (Poretsky et al. 1999, Marie et al. 2001). In our study, the supplement increased leptin concentrations from the second to the fifth day of supplementation, with higher values on the third day after the start of feeding. We suggest that increasing concentrations of glucose, insulin and leptin, from the first to the third day of feeding, prolonged the lifespan of the last non-ovulatory follicle and thus delayed the occurrence of atresia, slowing down follicle turnover (fewer follicular waves in supplemented ewes).

The impact of a short-term supplementation at the ovarian level may depend upon hormone dynamics. In supplemented ewes, peak glucose, insulin and leptin concentrations occurred one day before ovulatory wave emergence. Although we are aware of the limitations of ultrasonography for determining follicles in smaller size classes (Viñoles et al. 2004), the supplement increased the number of follicles growing from 2 to 3 mm. However, fewer follicles continued to grow up to 4 mm one day after ovulatory wave emergence compared with the untreated controls. If increased concentrations of glucose, insulin and leptin are important signals for the initial stimulation of gonadotrophin-responsive follicles, their concentrations may need to remain high in order to promote the selection of more than one follicle into the ovulatory wave. Still, high but decreasing concentrations of insulin and leptin on the fifth day of supplementation stimulated the ovulatory follicle to grow for a longer period, thus increasing the proportion of ewes with follicles larger than 6 mm. Similarly, Muñoz-Gutiérrez et al. (2002) found that the diameter of aromatase-positive follicles was increased by a nutritional supplement (lupins), and follicles >6 mm in diameter were seen only in supplemented ewes. Alternatively, increased concentrations of metabolic hormones favoured the selected follicle which exerted its dominant effect and reduced the number of follicles reaching 4 mm in supplemented ewes.

The changes in follicle development observed in the present study were independent of FSH action. The results of this study and a previous one (Viñoles et al. 2002) suggest that the static and the immediate effects of nutrition on follicle development are not mediated through common mechanisms, since, unlike the static effect, the immediate effect of nutrition is not mediated by changes in the FSH–oestradiol feedback system, but by an increase in glucose, insulin and leptin concentrations.

Figure 3 Changes in the concentrations of progesterone (diamonds) and oestradiol (triangles; top panel), FSH (triangles, middle panel), and leptin (circles) and IGF-I (squares; bottom panel) from days 9 to 17 of the oestrous cycle in ewes that received a short-term nutritional supplement (STS, n = 10) from day 9 to 14 of the oestrous cycle (solid symbols) and in ewes fed at maintenance level (n = 10, open symbols). Results are least squares means ± S.E.M.
in glucose, insulin and leptin concentrations (Fig. 5). Glucose and metabolic hormones act directly at the ovarian level to regulate steroidogenesis (Poretsky et al. 1999, Williams et al. 2001, Muñoz-Gutiérrez et al. 2004). Insulin and glucose infused together decrease the secretion of androstenedione and, to a lesser extent, oestradiol, while leptin can directly attenuate insulin-induced steroidogenesis in theca and granulosa cells (Spicer & Francisco 1997, Downing et al. 1999). In this study, androstenedione and oestradiol concentrations were similar among supplemented and control ewes, suggesting that the immediate effect of nutrition is not acting via the regulation of steroidogenesis, but most probably by stimulating glucose uptake by the follicles (Fig. 5, Muñoz-Gutiérrez et al. 2004).

The pool of follicles available for the action of glucose and metabolic hormones may play a key role in stimulating an increase in ovulation rate. Ewes in high body condition had a higher number of gonadotrophin-dependent follicles than did ewes in low body condition (Rhind & McNeilly 1986, Rhind et al. 1989, Xu et al. 1989, Viñoles et al. 2002). Moreover, a better response to nutritional supplementation has been described in ewes that are in high rather than low body condition (Leury et al. 1990). We suggest that the lack of an increase in ovulation rate observed in this study may be due to the low body

![Figure 4](image-url)
condition of the animals, since metabolic signals acting on a larger pool of follicles may have a better chance of rescuing follicles from atresia, thus increasing ovulation rate.

IGF-I plays a critical role in the selection of the dominant follicle and stimulates glucose metabolism by acting through the type I receptor (Fortune et al. 2004, Muñoz-Gutiérrez et al. 2004). In the present experiment, plasma IGF-I concentrations were similar between supplemented and control ewes, as shown in other studies (Downing et al. 1995). However, circulating concentrations of IGF-I may not be a good indicator of the amount available to the follicle, since IGF-I availability is regulated by proteases and binding proteins produced at the level of the follicle (Roche 1996, Mihm & Austin 2002, Fortune et al. 2004). Short-term nutritional supplementation decreases the percentage of IGF binding protein-2 positive follicles and changes the expression of mRNA IGF-I receptor (Muñoz-Gutiérrez et al. 2004) so, if IGF-I is involved in the response of the follicles to improved nutrition, then studies of the follicular microenvironment are necessary to clarify its role.

Our findings suggest that the effect of a short-term nutritional supplement on follicle development may be mediated by glucose and metabolic hormones acting directly at the ovarian level (Fig. 5). However, the impact of short-term supplementation on ovulation rate may depend on the factors mentioned above, such as follicular status, circulating concentrations of glucose and metabolic hormones, hormone dynamics, and the pool of follicles available for the action of these hormones at the time the supplement is fed. Considering all these factors together, it is not surprising that the outcome of the immediate effect of nutrition on ovulation rate is not consistent between and within laboratories, since the model for studying the effect has not yet been standardised for the female sheep (for review see Viñoles 2003).

We conclude that the effect of short-term nutritional supplementation on follicle development is not mediated by an increase in FSH concentrations, but by increased concentrations of glucose, insulin and leptin acting directly at the ovarian level. This effect is acute, since concentrations decrease after reaching peak values on the third day of supplementation. The status of follicle development at the time of maximum concentrations of glucose and metabolic hormones may be one of the factors that determine whether ovulation rate increases or not in response to nutritional treatment.
Acknowledgements

The authors wish to thank R García and O Cáseres for their excellent work and R Quadrelli for his support during the frequent blood sampling. We are grateful to E van Lier for her advice with the catheterisation procedure. Thanks are due to Y Pardiniñas for placing all the facilities of the Experimental Station ‘Bañoado de Medina’, Agronomy Faculty in Uruguay at our disposal. Thanks to Fatro Laboratory for the donation of prostaglandin and J C Sosa for the donation of mineral salt. We are grateful to E Rubianes for technical cooperation. We are grateful to M A Carlsson (SLU) for developing the androstenedione assay and to N Lundelheim and R Littell for their advice in statistical analyses. We would also like to thank D Blache, M Blackberry and K Hunt at the RIA Laboratory of Animal Science (UWA) for their support in the hormone analyses and J Milton for his advice in nutritional equations. We acknowledge Dr A F Parlow, and the NHPP and NIDDK analyses and J Milton for his advice in nutritional equations. We are grateful to E Rubianes for technical cooperation. We also wish to thank R Garcı́ and O Ca´seres for their advice with the catheterisation procedure. Thanks are due to Y Pardiniñas for placing all the facilities of the Experimental Station ‘Bañoado de Medina’, Agronomy Faculty in Uruguay at our disposal. Thanks to Fatro Laboratory for the donation of prostaglandin and J C Sosa for the donation of mineral salt. We are grateful to E Rubianes for technical cooperation. We are grateful to M A Carlsson (SLU) for developing the androstenedione assay and to N Lundelheim and R Littell for their advice in statistical analyses. We would also like to thank D Blache, M Blackberry and K Hunt at the RIA Laboratory of Animal Science (UWA) for their support in the hormone analyses and J Milton for his advice in nutritional equations. We acknowledge Dr A F Parlow, and the NHPP and NIDDK analyses and J Milton for his advice in nutritional equations. We are grateful to E Rubianes for technical cooperation. We also wish to thank R Garcı́ and O Ca´seres for their advice with the catheterisation procedure. Thanks are due to Y Pardiniñas for placing all the facilities of the Experimental Station ‘Bañoado de Medina’, Agronomy Faculty in Uruguay at our disposal. Thanks to Fatro Laboratory for the donation of prostaglandin and J C Sosa for the donation of mineral salt. We are grateful to E Rubianes for technical cooperation. We are grateful to M A Carlsson (SLU) for developing the androstenedione assay and to N Lundelheim and R Littell for their advice in statistical analyses. We would also like to thank D Blache, M Blackberry and K Hunt at the RIA Laboratory of Animal Science (UWA) for their support in the hormone analyses and J Milton for his advice in nutritional equations. We acknowledge Dr A F Parlow, and the NHPP and NIDDK analyses and J Milton for his advice in nutritional equations.

References


Gherardi PB & Lindsay DR 1982 Response of ewes to lupin supplementation at different times of the breeding season. Australian Journal of Experimental Agriculture and Animal Husbandry 22 264–267.


Muñoz-Gutiérrez M, Blache D, Martin GB & Scaramuzzi RJ 2002 Folliculogenesis and ovarian expression of mRNA encoding aromatase in anoestrous sheep after 5 days of glucose or glucosamine infusion or supplementary lupin feeding. Reproduction 124 721–731.

Muñoz-Gutiérrez M, Blache D, Martin GB & Scaramuzzi RJ 2004 Ovarian follicle expression of mRNA encoding the type 1 insulin like growth factor receptor (IGF-IR) and insulin like growth factor binding protein 2 (IGFBP2) in anoestrous sheep after 5 days of glucose, glucosamine or supplementary feeding with lupin grain. Reproduction 128 747–756.


Nottle MB, Seamark RF & Setchell BP 1990 Feeding lupin grain for five days prior to a cloprostenol-induced luteolysis can increase ovulation rate in sheep irrespective of when in the oestrous cycle supplementation commences. Reproduction, Fertility and Development 2 189–192.


Rhind SM, Leslie LD, Gunn RG & Doney JM 1985 Plasma FSH, LH, prolactin and progesterone profiles of Cheviot ewes with different levels of intake before and after mating, and associated effects on reproductive performance. Animal Reproduction Science 8 301–313.

Rhind SM, McMillen S, McKelvey WA, Rodriguez-Herrejon FF & McNeilly AS 1989 Effect of the body condition of ewes on...


Received 28 October 2004
First decision 22 November 2004
Accepted 22 November 2004