Effect of androgen treatment during foetal and/or neonatal life on ovarian function in prepubertal and adult rats

Victoria Tyndall¹, Marie Broyde¹, Richard Sharpe¹, Michelle Welsh¹, Amanda J Drake² and Alan S McNeilly¹

¹MRC Human Reproductive Sciences Unit and ²Endocrinology Unit, University/BHF Centre for Cardiovascular Science, The Queen’s Medical Research Institute, 47 Little France Crescent, Edinburgh EH16 4TJ, UK

Correspondence should be addressed to A S McNeilly; Email: a.mcneilly@ed.ac.uk

V Tyndall is now at King’s College London Medical School, London, SE1 1UL, UK
R Sharpe and AS McNeilly are now at the MRC Centre for Reproductive Health, The Queen’s Medical Research Institute, 47 Little France Crescent, Edinburgh, RH16 4TJ, UK
M Welsh is now at the School of Life Sciences, West Medical Building, University of Glasgow, Glasgow, G12 8QQ, UK

Abstract

We investigated the effects of different windows of testosterone propionate (TP) treatment during foetal and neonatal life in female rats to determine whether and when excess androgen exposure would cause disruption of adult reproductive function. Animals were killed prepubertally at d25 and as adults at d90. Plasma samples were taken for hormone analysis and ovaries serially sectioned for morphometric analyses. In prepubertal animals, only foetal + postnatal and late postnatal TP resulted in increased body weights, and an increase in transient, but reduced antral follicle numbers without affecting total follicle populations. Treatment with TP during both foetal + postnatal life resulted in the development of streak ovaries with activated follicles containing oocytes that only progressed to a small antral (smA) stage and inactive uteri. TP exposure during foetal or late postnatal life had no effect upon adult reproductive function or the total follicle population, although there was a reduction in the primordial follicle pool. In contrast, TP treatment during full postnatal life (d1–25) resulted in anovulation in adults (d90). These animals were heavier, had a greater ovarian stromal compartment, no differences in follicle thecal cell area, but reduced numbers of anti-Mullerian hormone-positive smA follicles when compared with controls. Significantly reduced uterine weights lead reduced follicle oestradiol production. These results support the concept that androgen programming of adult female reproductive function occurs only during specific time windows in foetal and neonatal life with implications for the development of polycystic ovary syndrome in women.

Reproduction (2012) 143 21–33

Introduction

Androgens have been shown to play important roles in ovarian function not only acting as substrates for oestradiol (E₂) production in preovulatory follicles but also by modulating ovarian function both directly and indirectly. Studies on both sheep (Steckler et al. 2007, 2009, Hogg et al. 2011) and primates (Abbott et al. 1998, 2002, 2007) have shown that inappropriate exposure to androgens in foetal life results in reduced or absent ovarian function in adult life and may provide models for polycystic ovary syndrome (PCOS) in women (Abbott et al. 1998, Franks 2009). Up to 22% of women may have polycystic ovaries (PCO), but only around 5–10% of women develop the syndrome (PCOS), associated with increased androgenisation, weight gain and infertility (Franks et al. 2006). Androgen receptors (ARs) are expressed in the oocyte, granulosa and thecal cells of the follicle, are highest in small follicles and expression is developmentally regulated during follicle development being down-regulated by FSH and LH (see Drummond 2006, Walters et al. 2008). Global knockout of the AR in female mice leads to reduced fertility with reduced numbers of antral follicles and increased granulosa cell (GC) apoptosis (Hu et al. 2004, Shiina et al. 2006, Walters et al. 2008). More refined studies on GC-specific AR-knockout mice suggest that these reproductive defects are attributed to lack of AR expression in GCs (Sen & Hammes 2010). Androgens promote mouse follicular development in vitro through effects on methylation and up-regulation of the FSH receptor expression in conditions that are marginal for follicle growth (Hillier & Tetsuka 1997, Murray et al. 1998a, 1998b). In contrast, excess androgens correlated with poor oocyte fertilisation and developmental rates.
(Anderiesz & Trounson 1995, Xia & Younglai 2000) and can induce atresia in developing follicles (Billig et al. 2003). Thus, it appears that in vitro the effects of androgens on follicle development in established follicle populations is dependent on the stage of follicle development and the ratio of androgens to E₂, while in the longer term AR expression in GCs is essential for normal follicle development (Drummond 2006).

Several studies on rats have demonstrated that continuous exposure of postpubertal animals to steroid hormones, either via regular s.c. dosing or continuous release pellet implant, produces an anovulatory phenotype in these animals (Faiman et al. 1988, Beloosesky et al. 2004, Misugi et al. 2006, Alexanderson et al. 2007, Baravalle et al. 2007, Manneras et al. 2007, 2008, Abbott et al. 2009). Such models recapitulate some of the features of PCOS in women who have increased ovarian and stromal volume, increased thecal 17α-hydroxylase (17αOH) activity (Nelson et al. 1999, 2001) and high plasma concentrations of LH, testoste-

| TP treatment group | Body weight (g) | Uterus weight (mg) | FSH (ng/ml) | LH (ng/ml) |
|--------------------|----------------|--------------------|------------|-----------|
| Control oil n=12   | 52 ± 2         | 28 ± 4             | 2.63 ± 0.3 | 6.5 ± 1.0 |
| Foetal n=8         | 51 ± 1         | 28 ± 2             | 1.68 ± 0.3 | 3.7 ± 0.7 |
| Full postnatal n=6 | 56 ± 3         | 137 ± 12           | 2.98 ± 0.8 | <0.8 †   |
| Foetal+postnatal n=5 | 81 ± 3†       | 121 ± 19†          | 2.10 ± 0.3 | 9.6 ± 2.3 |
| Late postnatal n=5 | 77 ± 6‡        | 202 ± 6‡           | 1.43 ± 0.5 | <0.8 †   |

Results are mean ± S.E.M. and were analysed by Student’s unpaired t-test (†P<0.01; ‡P<0.001).

(P<0.05 and P<0.01) increase in uterine weight and significant (P<0.001) reduction in plasma LH levels as expected, due to aromatisation of the TP to E₂ (Table 1).

Adult females

TP administration in all treatment groups except the late postnatal group (d15–24) resulted in an absence of vaginal opening. In adult animals at d90, TP treatment in full postnatal, foetal + postnatal and late postnatal treatment groups resulted in a significant (P<0.05 to P<0.001) increase in body weight and reduced uterine weights (Table 2). No differences in E₂ or LH levels were observed across TP treatment groups. FSH levels were significantly reduced in animals from the foetal + postnatal TP treatment group (P<0.05), and levels were undetectable (P<0.001) in the full postnatal TP treatment group (Table 2).

Mean adult ovarian weight was significantly reduced (P<0.01) in animals from the full postnatal TP treatment group due to a lack of corpora lutea (CL; Fig. 1C and Table 2). Ovaries from the foetal and late postnatal TP treatment groups appeared morphologically normal with comparable numbers of CL (data not shown) to control ovaries (Fig. 1A, B and E). Foetal + postnatal TP ovaries were very small and severely underdeveloped, embedded in fat and could not be weighed accurately. Upon histological examination (Fig. 1D), foetal + postnatal TP ovaries appeared as underdeveloped ‘streak’ ovaries set within the gonadal fat pad (Fig. 2B and D). These ovaries had significantly (P<0.01) fewer anti-Mullerian hormone (AMH)-positive follicles compared with controls (controls: 72 ± 20; foetal + postnatal testosterone: 8 ± 2).

Neonatal androgen exposure leads to a reduction in the pool of primordial follicles in the adult but not in the prepubertal rat

Compared with oil-treated controls, there were no significant differences in total follicle numbers in ovaries from any treatment groups in both immature d25 and adult d90 rats, except for a significant (P<0.05) reduction at d90 in the foetal + postnatal group (Fig. 3B). In immature rats, both full and late postnatal
treatment groups had a significantly (P ≤ 0.05) greater proportion of transitory and significantly (P ≤ 0.05) reduced proportions of antral follicles. Adults had a significantly (P < 0.01) reduced proportion of primordial follicles in postal TP treatment groups compared with controls. Furthermore, adults from the foetal + postnatal group also had a significantly reduced number of follicles (P ≤ 0.05) and proportion of antral follicles (P ≤ 0.001), since most follicles from this treatment group did not reach large antral (lgA) stage (Fig. 2B and C).

Assessment of adult anovular ovaries following full postnatal treatment

Full postnatal TP ovaries were morphologically cystic in appearance (Figs 1 and 4–7). To quantify this cystic phenotype, average antrum size of follicles containing an oocyte with a visible nucleus was measured in every tenth section (Fig. 4C). Full postnatal TP led to a significant (P ≤ 0.001) increase in antrum size compared with controls.

Follicular functional status in both control and full postnatal TP-treated animals was measured using semiquantitative analysis of AMH (Fig. 4) and aromatase expression (Fig. 5). Follicles expressing these proteins were counted and categorised in every tenth serial section. Although reduced, no significant difference in the total number of AMH-positive follicles was found (Fig. 4D), but there was a significantly (P ≤ 0.01) reduced proportion of AMH-positive small antral (smA) follicles in full postnatal TP-treated ovaries (Fig. 4E). Compared with controls, the number of aromatase-positive antral follicles was significantly (P ≤ 0.05) greater in full postnatal TP-treated animals (Fig. 5D). However, no differences were found in the proportions of lgA follicles staining between individual thecal area and follicle diameter in either treatment group (Fig. 6E). After correction for the area of the CLs in the control ovaries, the 3βHSD-positive stromal compartment (Fig. 7C and D) of full postnatal TP ovaries was significantly (P ≤ 0.05) larger than that of control ovaries and occupied a significantly (P ≤ 0.001) greater proportion of ovarian area.

Discussion

As outlined in the Introduction section, androgens can have profound positive and negative effects on ovarian follicle development as well as providing the essential substrate for E2 production by preovulatory follicles. Chronic administration of steroids or steroidogenic inhibitors during postnatal life in the rat have been assessed to determine whether any altered reproductive phenotype in the adult resembles that found in women with PCOS. Steroids used include testosterone (Beloosesky et al. 2004), DHEA (Anderson & Lee 1997, Anderson et al. 1997), E2 valerate (Brawer et al. 1986, Rosa et al. 2003), DHT and letrozole (Manneras et al. 2007) while single bolus injection of testosterone or E2 on or just after the day of birth resulted in a PCO-like phenotype in the adult animal (Barracough 1961, Brawer et al. 1978, Ota et al. 1986, Grossmann et al. 1987, Alexanderson et al. 2007). However, relatively few rodent models have fully examined the reproductive phenotype in the adult animal and contrasted it with the clinical situation (Ota et al. 1986, Misugi et al. 2006, Abbott et al. 2007). Follicle formation is a continuum in both humans and sheep, occurring during mid gestation (McNatty et al. 2000, Childs et al. 2010). In the rat, follicle formation occurs in the first few days after birth (Schindler et al. 2010), equating to approximately midgestation in humans. In this study, rats were exposed to TP in various time windows, including postnatal only or foetal + postnatal life. Thus, it is reasonable in our current study to assume that the exposure to TP and, potentially, any E2 that may be converted from the TP (Picon et al. 1985), would occur over a similar time period of follicle formation and development as in the other non-rodent models of PCOS, namely sheep (Steckler et al. 2007) and primates (Abbott et al. 2009).

Table 2 Body and organ weights and hormone concentrations across TP treatment groups in adult animals (d90).

| TP treatment group | Body weight (g) | Ovary weight (mg) | Uterus weight (mg) | E2 (pg/ml) | FSH (ng/ml) | LH (ng/ml) |
|--------------------|-----------------|------------------|--------------------|------------|-------------|------------|
| Control oil n = 5  | 213 ± 9         | 95 ± 4           | 433 ± 52           | 12.1 ± 2.1 | 1.8 ± 0.1   | 5.9 ± 1.0  |
| Foetal n = 3       | 228 ± 7         | NA               | 470 ± 75           | 16.9 ± 1.2 | 2.1 ± 0.7   | 4.7 ± 1.0  |
| Full postnatal n = 4 | 318 ± 6       | 59 ± 5           | 307 ± 15           | 4.2 ± 1.3  | < 0.8†      | 2.2 ± 0.9  |
| Foetal + postnatal n = 5 | 309 ± 5  | NA              | 158 ± 3.5         | 9.7 ± 2.9  | 1.3 ± 0.4†  | 4.3 ± 1.0  |
| Late postnatal n = 5 | 239 ± 6      | 80 ± 5           | 247 ± 38          | 9.3 ± 3.2  | 1.6 ± 0.2   | 5.1 ± 1.2  |

Results are mean ± s.e.m. and were analysed by Student’s unpaired t-test (g P ≤ 0.01; †P ≤ 0.001). NA, not available.
Foetal TP has no discernable effects upon the female rat

Foetal TP did not affect folliculogenesis or fertility in immature or adult animals, as evidenced by the presence of normal numbers of CL in the adult animals, and normal follicle numbers and proportions at both ages. Immature and adult body weights were normal, and gonadotrophin and E2 levels were comparable to those seen in controls at each stage of investigation for this TP treatment group. These findings corroborate previous investigations into the effects of foetal TP upon female reproductive development (Slob et al. 1983). It is worth noting that there may be strain differences in the response to foetal androgen exposure as foetal TP administration in Sprague–Dawley (SD) rats in one study reduced both immature and adult body weight (Wolf et al. 2002, 2004). Foetal TP-treated female rats develop masculinised features of the urogenital tract, for example, an increased anogenital distance, as described in the original papers upon which these studies were based (Welsh et al. 2008, 2010).

Postnatal TP treatment leads to changes in ovarian follicle composition in the immature animal

At d25, in late postnatal TP and foetal + postnatal TP (streak ovary) treatment groups, a higher proportion of activating transitory follicles and lower proportion of smA follicles was observed, with no effects upon total follicle numbers at this age. These results demonstrate the mitogenic effects of testosterone upon folliculogenesis, whereby androgens stimulate GC proliferation of smaller newly activated transitory and primary follicles as shown in graft experiments of ruminant follicles pre-exposed to testosterone and transplanted into chick ovaries; the grafts were shown to selectively increase the proportion of growing primary follicles (Qureshi et al. 2008). The same graft study found androgens to have a protective effect upon follicle survival, which is at odds with the reduced proportion of smA follicles found in the late postnatal and foetal + postnatal TP treatment groups of the current study. In the rat, ovarian AR expression is reduced in GCs of FSH-responsive follicles (Tetsuka et al. 1995) and other studies on rats have shown that
exogenous androgen administration at the later stages of gonadotrophin-dependent folliculogenesis can facilitate follicular atresia (Drummond 2006, Honnma et al. 2006). Indeed, the smA follicles present in the immature rat ovary do not express sufficient aromatase in GCs for the production of E$_2$ (Mahesh et al. 1987). Thus, in the current study, TP likely acts to promote GC atresia in more advanced follicles, which would account for the reduction in smA follicle proportions within these TP treatment groups at d25.

**TP treatment after birth up to d15 leads to weight gain in adult animals**

Significant weight gain was observed at d25 in immature foetal + postnatal and late postnatal TP-treated animals and in adults within all postnatal TP treatment groups. It is possible that this weight gain would affect metabolic function, but this was not assessed in this study. Other studies have only examined prenatal oestrogen (Roland et al. 2010) and glucocorticoids (Cottrell & Seckl 2009), which can induce metabolic dysfunction in mice and rats. In the current study, the increase in body weight was much less in the late postnatal group in the adults. This indicates that the window of postnatal androgen exposure which can produce a metabolic phenotype extends for at least 14 days after birth, but the effect appears to be less if treated only in the late postnatal period from d15 to 25. Furthermore, these findings extend results from other studies in which regular neonatal administration of androgens (testosterone; Nilsson et al. 1998; DHT: Alexanderson et al. 2007) or aromatase inhibitors (Letrozole; Manneras et al. 2007) has been shown to program weight gain and increased adiposity in adult animals. Recently, it has been suggested that oestrogen receptors, implying oestrogen action, exert a stronger metabolic programming effect than ARs (Beloosesky et al. 2004). In this study, we do not know to what extent, if at all, the TP would have been converted to E$_2$ in the neonatal treatment periods, and this will require further investigation.

**Foetal + postnatal TP treatment leads to the formation of underdeveloped streak ovaries**

TP administered during both foetal and postnatal life resulted in underdeveloped streak ovaries within both immature and adult animals, which by d90 of life had fewer follicles, fewer activated AMH-expressing follicles and consequent lower circulating E$_2$ levels with significantly reduced uterine weights. Secretion of AMH by GCs of growing follicles is known to inhibit further follicle activation (Durlinger et al. 2002, Rey et al. 2003, Pellatt et al. 2010). It is thus conceivable that the lower expression of AMH in the streak ovaries might allow unimpeded follicular recruitment and subsequent atresia. This could account for the reduction in follicle numbers within the adult animals by d90. The presence of streak ovaries at both d25 and d90 also implicates a combined effect of late foetal and early postnatal TP exposure, given that no effect was found in foetal TP-treated animals and a different effect was observed in the full postnatal TP-treated animals. Streak ovaries have been associated with FSH receptor (Levallet et al. 1999) and BMP15 (Rossetti et al. 2009) mutations and is associated with an autoimmune response in neonatally thymectomised mice (Scalzo & Michael 1988). The potential role of the thymus is unknown.
However, the involvement of alterations in FSH receptor function was not examined in the current study, while BMP15 does not appear to play a significant role in rodent follicle formation or development (Yan et al. 2001). Neonatal E₂ exposure in rodents prevents germ cell nest breakdown in the ovary (Kezele & Skinner 2003, Jefferson et al. 2006) and effects involving ESR1 (ER₂), ESR2 (ERβ) and the membrane-bound estrogen receptor (Chen et al. 2009). It is possible that in our study, circulating E₂ resulting from the placental or foetal and neonatal ovarian aromatisation of TP (Picon et al. 1985) and/or levels of TP acting purely as an androgen in animals exposed as foetuses were high enough to persist into early neonatal life and increase the sensitivity of the postnatal animal to further androgen exposure. This notion is supported by another study in which TP administered to pregnant SD rats during the late foetal period (e17–e18) had a priming effect upon female animals, leading to the masculinisation of female behaviour and physiology in animals, which subsequently received a small postnatal dose of 5 μg TP (Hoepfner & Ward 1988). Animals in the foetal and foetal + postnatal TP treatment group were cross fostered to untreated dams that had recently littered and/or account for the effects seen, as aromatase activity has been shown in the neonatal pituitary (Carretero et al. 2003) and ovaries (Picon et al. 1985) of female rats. Additionally, reduced primordial follicle recruitment could play a part in the conservation of the primordial pool documented in the current study.

**The postnatally androgenised rat as a PCOS model**

Full postnatal TP (d1–25) induced a PCOS-like phenotype in the adult rat and was evaluated as a PCOS model.
Animals from this treatment group had reduced ovarian weights due to anovulation and lack of CL in addition to low uterine weights that, as a bioassay for oestrogens in the rat, indicate low oestrogen levels. However, it should be noted that plasma E$_2$ levels were comparable to controls in this group. An unexpected effect was that plasma FSH levels were undetectable, and LH levels were low in this treatment group. Given that the bioassay for E$_2$ levels indicated reduced levels (though when measured plasma E$_2$ was within normal range), it seems reasonable to expect that this would result in an increase in both LH and FSH secretion (McNeilly et al. 2003).

The fact that this has not occurred suggests that the TP treatment has altered the negative feedback regulation of gonadotrophin secretion and will require further investigation.

Histological examination of ovaries from full postnatal TP-treated animals showed that they were anovular, with large fluid filled antral follicles apparent on subjective inspection. The reduction in the proportion of smA AMH-positive follicles in animals given postnatal TP may have contributed towards a larger mean number of antral follicles staining positive for aromatase found in adjacent sections and supports the idea that the functional status of follicles was altered, particularly given the low levels of FSH. This is supplemented by the fact that follicles within the ovaries of adult postnatal TP-treated animals had larger mean antrum sizes. Further studies will be required to determine whether the altered ovarian phenotype that recapitulates some of the features of PCOS are related only to long-term effects on the ovary of TP exposure in the prenatal period or to altered gonadotrophin levels with or without a presumed metabolic effect related to weight gain.

No difference was observed between ovaries from the full postnatal TP-treated animals and control animals with regard to the status of the theca. Indeed, the thecal compartment was of a comparable size, surrounded a comparable number of follicles and the 17αOH stained area was similar between control and TP-treated animals. Given that the theca only develops around follicles as they grow beyond the primary stage (Young & McNeilly 2010), these results suggest that in the rat, at least there is no long-term effect on any thecal precursor cells in the stroma in adult life. In contrast, a novel finding of this study was the increased amount and proportion of 3βHSD-positive stromal found in full postnatal TP-treated animals, which further recapitulates clinical observations in women with PCOS (Fulghesu et al. 2007, Welt et al. 2005). As no differences in circulating gonadotrophins were observed in adult full postnatal TP-treated animals, this would indicate that this increased stromal compartment could be due to primary programming of non-thecal precursor cells, as hypothesised by other investigators (Franks 2009).
In conclusion, the results from the foetal + postnatal TP group, in agreement with the findings of other studies (Hoepfner & Ward 1988), demonstrate that foetal TP may reduce the sensitivity threshold to neonatal testosterone exposure in female animals, providing one explanation for the failure of normal ovarian development observed in this treatment group. Exposure to TP during the first 25 days of life additionally led to a significant increase in both immature and adult body weight through an as yet unknown mechanism. Finally, TP administration during the first 15 days of life resulted in a PCOS-like phenotype in the female: overweight, anovulatory and with larger than normal follicles as well as an increased ovarian stromal compartment. However, these phenotypic changes occur in the absence of the abnormalities of FSH and LH typically associated with PCOS and suggest that neonatal TP exposure in the rat at least does not provide an adequate model for all aspects of PCOS.

Materials and Methods

Animals and treatments

Wistar rats were bred and maintained in the University of Edinburgh animal facility under standard conditions with licensed approval from the University Ethical Review Process and a UK Home Office Project Licence. Animals had access to water and a soy-free breeding diet made available ad libitum (SDS; Dundee, Scotland). Matings were timed and presence of a vaginal plug designated embryonic day 0.5 (e0.5). TP treatment regimes are summarised in the original paper.
Briefly, for foetal TP treatment, pregnant dams were injected s.c. with 20 mg/kg TP in 0.4 ml corn oil daily between e14.5 and 21.5. To avoid foetal mortality that can be brought about by dystocia (Welsh et al. 2008), foetuses from TP-treated dams were delivered by caesarean and cross-fostered to untreated dams that had delivered within the last 6 h (Welsh et al. 2010). For postnatal TP treatment, pups were injected with 20 mg/kg TP every 3 days (d) in three different treatment groups: i) late postnatal TP treatment, from d15 to 24; ii) foetal + postnatal TP treatment, from foetal e14.5 to e21.5, then from d1 (the day after birth) to 24; and iii) full postnatal TP treatment, from d1 to 24. At least three litters were used per treatment group. Control animals received the same volume of oil as for each treatment period and were treated in foetal and neonatal life. This treatment had previously been shown to have no effect when compared with untreated controls (Welsh et al. 2010). Animals were killed by inhalation of carbon dioxide and cervical dislocation on d25 (immature) and d90 (adult). Body and organ weights were documented and a blood sample collected by cardiac puncture for hormone analysis. Ovaries and uteri were fixed in Bouins for 6 h, transferred into 70% ethanol and processed into paraffin wax using standard methods.

**Ovarian IHC**

One ovary from each animal was serial sectioned (5 µm thickness) and mounted onto permafrost slides for analysis using multiple IHC markers. Every tenth section was used for each set of IHC analysis. Sections were stained with haematoxylin and eosin (H&E) for follicle count analysis, or DAB IHC was performed as described previously (McNeilly et al. 2000).
Primary antibodies were diluted in normal serum: mouse anti-aromatase at 1:50 μl (Gift – Prof. N Groome, Oxford Brookes University); goat anti-AMH at 1:500 (Santa Cruz Biotechnology, Santa Cruz, CA, USA: MIS C-20); rabbit anti-17βOH at 1:500 (Gift– Dr D Hales Southern Illinois University); and rabbit anti-3βHSD at 1:250 (Gift–Prof. I Mason, University of Edinburgh). Slides were counterstained with haematoxylin, dehydrated through alcohols and xylene and mounted in pertex (Histolab, Gothenburg, Sweden) for analysis.

**Morphological and stereological analysis**

Every tenth section of each ovary was H&E stained and examined at ×20 and ×40 magnification under a light microscope. Follicles were counted and classified according to a previously published method, allowing for a realistic estimate of follicle proportions with minimal data manipulation (Hirshfield & Midgley 1978, McNeilly et al. 2011). Only follicles containing a visible oocyte nucleus were included in the count, and these were classified based on their GC morphology. A layer of flat squamous GCs indicated a primordial follicle. Two or more cuboidal GCs indicated a transitory follicle. If two or less than two layers of cuboidal GCs were present around the oocyte, the follicle was primary and if more than two layers of cuboidal GCs were present around the oocyte, the follicle was secondary. If there were more than two layers of cuboidal GCs and an antrum had formed, the follicle was secondary. If more than two layers of cuboidal GCs were present around the oocyte, the follicle was primary and if more than two layers of cuboidal GCs were present around the oocyte, the follicle was secondary. If more than two layers of cuboidal GCs and an antrum had formed, the follicle was secondary.

The number of cells per follicles expressing AMH or aromatase as well as their staining intensity was used to semi-quantitatively assess follicle morphology. Morphometric analysis of antrum size, 17αOH staining and 3βHSD staining was completed using stereological equipment comprising an Olympus BH-2 microscope fitted with a Prior automatic stage (Prior Scientific Instruments Ltd., Cambridge, UK). For 17αOH and 3βHSD, stained area below 10 μm² was not included, in order to discount any background. CL and areas of non-specific staining were also masked using Image-Pro Plus version 6.2 with Stereologer-Pro 5 plug-in software (Media Cybernetics UK, Wokingham, Berkshire, UK), as published previously (McNeilly et al. 2011). Figure images were taken with a Provis AX70 (Olympus Optical, London, UK) microscope fitted to a Canon DS6031 camera (Canon Europe, Amsterdam, The Netherlands).

**Hormone RIA and ELISA**

Plasma E₂ was assayed following solvent extraction using a sensitive RIA modified for mouse plasma as described previously (McNeilly et al. 2000, 2011). All samples were assayed in a single assay with a minimum detectable concentration of 4.6 pg/ml and an intra-assay coefficient of variation of <8%. The in-house ELISAs using the same general method were performed to assess plasma FSH and LH. Capture antibody diluted in buffer (50 μl of 0.2 M sodium bicarbonate–carbonate buffer) was adsorbed onto plastic plates (Nunc Biologicals, Thermo Scientific, Epsom, Surrey, UK) overnight. Plates were then washed with wash buffer (0.1 M Tris–HCl pH 7.5) and then washed with wash buffer (Tris–HCl, pH 7.5) containing 0.001% Tween20, 1% BSA (Sigma A3294) and 0.1% bovine-γ-globulin for a further 1 h. After a further wash, 5 μl standards, quality controls and unknown samples were added to the plate in duplicate and incubated with 95 μl assay buffer overnight at 4 °C. Between steps, plates were washed using 200 μl wash buffer and dried and all antibodies/samples were diluted in assay buffer.

For the FSH ELISA, mouse FSH (NIDDK AFP5308D; Dr A Parlow, Harbor-UCLA Medical Center, Torrance, CA, USA) was used as standard (range 0.78–50 ng/ml) with anti-human FSH MAB (4 μg/ml; MedixBiochemica 6602, BiosPacific, Inc., Emeryville, CA, USA) used as capture antibody. The second antibody was biotinylated rabbit anti-rat α-subunit IC1 (NIDDK AFP66P99860; 5 μg/ml) and anti-rabbit HRP conjugate (1:10 000; Thermo Scientific #31458) was used for signal detection.
For the LH ELISA, mouse LH (NIDDK AFP5306A) was used as standard (range 0.78–25 ng/ml) with anti-bovine LH chain MAB (2 μg/ml; 518B7; Gift – Dr J Roser, UCLA) used as capture antibody. The second antibody was biotinylated anti-human LH MAB (MedixBiochemica 5303, Kauniainen, Finland; 1 μg/ml) and Amplex streptavidin-HRP conjugate (1:10 000; GE Healthcare, Chalfont St Giles, Bucks, UK) RPN4401V was used for signal detection.

After a final wash cycle, both ELISAs were visualised with 100 μl 3',5',5'-tetramethylbenzidine micro-well peroxidase blue chromogen solution for 30 min (KPL). In both assays, oxidation was stopped by addition of 6% phosphoric acid. Absorbance was read using a plate spectrophotometer as λ 450 nm minus λ 620 nm to eliminate any background from the results that were analysed using AssayZap (Biosoft, Cambridge, UK).

### Statistical analysis

All data shown are expressed as mean ± s.e.m. and analysed using GraphPad Prism 5 (GraphPad Software, Inc., San Diego, CA, USA). Two-way ANOVA was performed between any group data, for example, follicle counts, with a Bonferroni post hoc analysis to assess for specific in-group differences. Unless otherwise stated, a two-tailed unpaired Student’s t-test was used to evaluate the differences between rodent treatment groups in all other circumstances. To determine the correlation between two sets of data, the Pearson product–moment correlation (PPMC) was used. To assess for differences between two correlation coefficients, a Fisher r-to-Z transformation was performed and applied to the sample correlation coefficients (r) calculated by PPCM. In Graphpad Prism, a P value for a Fisher transformation was then calculated to test for the significant differences between two sets of overlapping correlative data.

### Declaration of interest

The authors declare that there is no conflict of interest that could be perceived as prejudicing the impartiality of the research reported.

### Funding

This work was funded by the Medical Research Council (G00007.01) to A S McNeilly.

### Acknowledgements

We thank Judy McNeilly, Linda Nicol, Ian Swanston, Nancy Evans, Mike Millar and Mark Fiskin for excellent technical help.

### References

Abbott DH, Dumesic DA, Eisner JR, Colman RJ & Kemnitz JW 1998 Insights into the development of polycystic ovary syndrome (PCOS) from studies of prenatally androgenized female rhesus monkeys. Trends in Endocrinology and Metabolism 9 62–67. (doi:10.1016/S1043-2760(98)00019-8)

Abbott DH, Dumesic DA & Franks S 2002 Developmental origin of polycystic ovary syndrome – a hypothesis. Journal of Endocrinology 174 1–5. (doi:10.1677/joe.0.1740001)

Abbott DH, Dumesic DA, Levine JE, Dunai A & Padmanabhan V 2007 Animal models and fetal programming of the polycystic ovary syndrome. In Contemporary Endocrinology: Androgen Excess Disorders in Women, IV, pp 259–272. Ed. R Azizz. Totawa, NJ: Humana Press.

Abbott DH, Tarantal AF & Dumesic DA 2009 Fetal, infant, adolescent and adult phenotypes of polycystic ovary syndrome in prenatally androgenized female rhesus monkeys. American Journal of Primatology 71 776–784. (doi:10.1002/ajp.20679)

Alexander C, Eriksson E, Stener-Victorin E, Lystig T, Gabrielson B, Lonn M & Holmang A 2007 Postnatal testosterone exposure results in insulin resistance, enlarged mesenteric adipocytes, and an atherogenic lipid profile in adult female rats: comparisons with estradiol and dihydrotestosterone. Endocrinology 148 5369–5376. (doi:10.1210/en.2007-0305)

Anderiesz C & Trounson AO 1995 The effect of testosterone on the maturation and developmental capacity of murine oocytes in vitro. Human Reproduction 10 2377–2381.

Anderson E & Lee GY 1997 The polycystic ovarian (PCO) condition: apoptosis and epithelialization of the ovarian antral follicles are aspects of cystogenesis in the dehydroepiandrosterone (DHEA)-treated rat model. Tissue & Cell 29 171–189. (doi:10.1016/S0040-8166(97)80017-1)

Anderson E, Lee GY & O’Brien K 1997 Polycystic ovarian condition in the dehydroepiandrosterone-treated rat model: hyperandrogenism and the resumption of meiosis are major initial events associated with cystogenesis of antral follicles. Anatomical Record 249 44–53. (doi:10.1002/sjc.1097-0185(19970924)249:1<44::AID-ARQ3.0.CO;2-F)

Baravalle C, Salvetti NR, Mira GA, Lorente JA & Ortega HH 2007 The role of ACTH in the pathogenesis of polycystic ovarian syndrome in rats: hormonal profiles and ovarian morphology. Physiological Research 56 67–78.

Barraclough CA 1961 Production of anovulatory, sterile rats by single injections of testosterone propionate. Endocrinology 68 62–67. (doi:10.1210/endo-68-1-62)

Beloosesky R, Gold R & Almog B 2004Induction of polycystic ovary by testosterone in immature female rats: modulation of apoptosis and attenuation of glucose/insulin ratio. International Journal of Molecular Medicine 14 207–215.

Billig H, Furuta I & Hsueh AJ 2003 Oestrogens inhibit and androgens enhance ovarian granulosa cell apoptosis. Endocrinology 133 2204–2212. (doi:10.1210/endo.133.5.2204)

Brawer JR, Naftolin F, Martin J & Sonnenschein C 1978 Effects of a single injection of estradiol valerate on the hypothalamic arcuate nucleus and on reproductive function in the female rat. Endocrinology 103 501–512. (doi:10.1210/endo-103-2-501)

Brawer JR, Munoz M & Farookhi R 1986 Development of the polycystic ovary condition (PCO) in the estradiol valerate-treated rat. Biology of Reproduction 35 647–655. (doi:10.1095/biolreprod35.3.647)

Carretero J, Vázquez G, Rubio M, Blanco E, Juanes JA, Pérez E, Burks D & Vázquez R 2003 Postnatal differentiation of the immunohistochemical expression of aromatase P450 in the rat pituitary gland. Histology and Histochemistry 18 419–423.

Champagne FA, Francis DD, Mar A & Meaney MJ 2003 Variations in maternal care in the rat as a mediating influence for the effects of environment on development. Physiology & Behavior 79 359–371. (doi:10.1016/S0031-9384(03)00149-5)

Chen Y, Breen K & Pepling ME 2009 Estrogen can signal through multiple pathways to regulate oocyte cyst breakdown and primordial follicle assembly in the neonatal mouse ovary. Journal of Endocrinology 202 407–417. (doi:10.1677/JOE-09-0109)

Childs A, Kinnell HL, Collins CS, Hogg K, Bayne RAL, Green SJ, McNeilly AS & Anderson RA 2010 BMP signaling in the human fetal ovary: insights into the development of polycystic ovary syndrome. Frontiers in Behavioral Neuroscience 3 1–9. (doi:10.3389/neuro.08.019.2009)

Drummond AE 2006 The role of steroids in follicular growth. Reproductive Biology and Endocrinology 4 16–26. (doi:10.1186/1477-7827-4-16)
Durlinger ALL, Visser VA & Themmen APN 2002 Regulation of ovarian function: the role of anti-Mullerian hormone. Reproduction 124 607–615. (doi:10.1530/rep.0.1240801)

Faiman C, Reyes FL, Dent DW, Fuler GB, Hobson WC & Thiliviers JA 1988 Effects of long-term testosterone exposure on ovarian function and morphology in the rhesus monkey. Anatomical Record 222 245–251. (doi:10.1002/ar.1092220305)

Franks S 2009 Do animal models of polycystic ovary syndrome help to understand its pathogenesis and management? Yes, but their limitations should be recognized Endocrinology 150 3983–3985. (doi:10.1210/en.2009-0652)

Franks S, McCarthy ML & Hardy K 2006 Development of polycystic ovary syndrome: involvement of genetic and environmental factors. International Journal of Andrology 28 278–285. (doi:10.1111/j.1365-2605.2005.00623.x)

Fuglsang AM, Angioni S, Frau E, Belosi C, Apa R, Mioni R, Xamin N, Grippe A, Gross DM, Desole S, Fuzzetti F et al. 2007 Ultrasound in polycystic ovary syndrome – the measuring of ovarian stroma and relationship with circulating androgens: results of a multicentric study. Human Reproduction 22 2501–2508. (doi:10.1093/humrep/dem202)

Grossmann R, Diez-Guerra FJ, Mansfield S & Dyer RG 1987 Neonatal testosterone modifies LH secretion in the adult female rat by altering the opioid-noradrenergic interaction in the medial preoptic area. Brain Research 375 207–210. (doi:10.1016/0006-8993(87)90832-3)

Hillier SG & Tetsuka M 1997 Role of androgens in follicle maturation and atresia. Bailliere’s Clinical Obstetrics and Gynaecology 11 249–260. (doi:10.1016/S0955-3522(97)80036-3)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hoepfner BA & Ward IL 1998 Prenatal and neonatal androgen exposure interact to affect sexual differentiation in female rats. Behavioral Neuroscience 102 61–65. (doi:10.1037/0735-7044.102.1.61)

Hogg K, McNeilly AS & Duncan WC 2011 Prenatal androgens and Testicular Function study of polycystic ovaries and ovarian atresia. In (99)00058-5)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)

Hirshfield AN & Midgley AR Jr 1978 Morphometric analysis of follicular metabolic disturbances and modulate gene expression in adipose tissue. The Journal of Clinical Endocrinology and Metabolism 48 1043–1049. (doi:10.1210/jcem.39.12.143)
Walters KA, Allan CM & Handelsman DJ 2008 Androgen action and the ovary. *Reproduction* 78 380–389. (doi:10.1530/reprod.107.064089)

Webber LJ, Stubbs S, Stark J, Trew GH, Margara R, Hardy K & Franks S 2003 Formation and early development of follicles in the polycystic ovary. *Lancet* 362 1017–1021. (doi:10.1016/S0140-6736(03)14410-8)

Welsh M, Saunders PT, Fiskin M, Scott HM, Hutchison GR, Smith LB, Richard M & Sharpe RM 2008 Identification in rats of a programming window for reproductive tract masculinization, disruption of which leads to hypospadias and cryptorchidism. *Journal of Clinical Investigation* 118 1479–1490. (doi:10.1172/JCI34241)

Welsh M, MacLeod DJ, Walker M, Smith LB & Sharpe RM 2010 Critical androgen-sensitive periods of rat penis and clitoris development. *International Journal of Andrology* 33 e144–e152. (doi:10.1111/i.1365-2605.2009.00978.x)

Welt CK, Taylor AE, Fox J, Messerlian GM, Adams JM & Schneyer AL 2005 Follicular arrest in polycystic ovary syndrome is associated with deficient inhibin A and B biosynthesis. *Journal of Clinical Endocrinology and Metabolism* 90 5582–5587. (doi:10.1210/jc.2005-0695)

Wolf CJ, Ostby JS, LeBlanc GA & Gray LE Jr 2002 Effects of prenatal testosterone propionate on the sexual development of male and female rats: a dose–response study. *Toxicological Science* 65 71–86. (doi:10.1093/toxsci/65.1.71)

Wolf CJ, LeBlanc GA & Gray LE Jr 2004 Interactive effects of vinclozolin and testosterone propionate on pregnancy and sexual differentiation of the male and female SD rat. *Toxicological Science* 78 135–143. (doi:10.1093/toxsci/ki018)

Xia P & Younglai EV 2000 Relationship between steroid concentrations in ovarian follicular fluid and oocyte morphology in patients undergoing intracytoplasmic sperm injection (ICSI) treatment. *Journal of Reproductive and Fertility* 118 229–233. (doi:10.1530/reprod/118.2.229)

Yan C, Wang P, DeMayo J, DeMayo FJ, Elvin JA, Carino C, Prasad SV, Skinner SS, Dunbar BS, Dube JL et al. 2001 Synergistic roles of bone morphogenetic protein 15 and growth differentiation factor 9 in ovarian function. *Molecular Endocrinology* 15 854–866. (doi:10.1210/me.15.6.854)

Young J & McNeilly AS 2010 Theca – the forgotten cell of the ovarian follicle. *Reproduction* 140 489–504. (doi:10.1530/REP-10-0094)

Received 30 June 2011
First decision 17 August 2011
Accepted 19 October 2011