

# Arginine vasopressin infusion is sufficient to model clinical features of preeclampsia in mice

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Copeptin, a marker of arginine vasopressin (AVP) secretion, is elevated throughout human pregnancies complicated by preeclampsia (PE), and AVP infusion throughout gestation is sufficient to induce the major phenotypes of PE in mice. Thus, we hypothesized a role for AVP in the pathogenesis of PE. AVP infusion into pregnant C57BL/6J mice resulted in hypertension, renal glomerular endotheliosis, intrauterine growth restriction, decreased placental growth factor (PGF), altered placental morphology, placental oxidative stress, and placental gene expression consistent with human PE. Interestingly, these changes occurred despite a lack of placental hypoxia or elevations in placental fms-like tyrosine kinase-1 (FLT1). Coinfusion of AVP receptor antagonists and time-restricted infusion of AVP uncovered a mid-gestational role for the AVPR1A receptor in the observed renal pathologies, versus mid- and late-gestational roles for the AVPR2 receptor in the blood pressure and fetal phenotypes. These findings demonstrate that AVP is sufficient to initiate phenotypes of PE in the absence of placental hypoxia, and indicate that AVP may mechanistically (independently, and possibly synergistically with hypoxia) contribute to the development of clinical signs of PE in specific subtypes of human PE. Additionally, they identify divergent and gestational time-specific signaling mechanisms that mediate the development of PE phenotypes in response to AVP.

**Conflict of interest:** MKS, DAS, and JLG have submitted Patent Cooperation Treaty applications (PCT/US2014/015327, PCT/US2014/015631, and PCT/US2018/027152) and hold a US Patent (no. 9,937,182) describing a role for AVP in the potential diagnosis and therapeutic treatment of preeclampsia. Ongoing research by MKS, DAS, and JLG developing diagnostic tests for preeclampsia that involve measurements of the AVP system are supported in part by a seed grant from Carmentix Pte Ltd/Esco Ventures.

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## Introduction

Preeclampsia (PE) is a common and life-threatening disease of pregnancy characterized by hypertension, proteinuria, renal glomerular endotheliosis (RGE), fetal growth restriction, placental hypoxia, headache, and edema along with various other nonspecific symptoms (1, 2). Clinical signs and symptoms of PE typically manifest around mid-gestation no earlier than week 20, and progress in severity until birth or early fetal and placental delivery. If PE is allowed to progress, maternal seizures, coma, and maternal and fetal death can occur. Fetal and placental delivery remain the only cure.

While the initiating cause of PE is unknown, it is known that women with PE exhibit placental dysfunction early in pregnancy (3). Fetal trophoblasts responsible for invading and dilating maternal spiral arteries that supply blood to the growing fetoplacental unit are dysfunctional in PE (3), and this is thought to lead to abnormally small spiral arteries, poor placental perfusion, and placental and fetal hypoxia.

It is generally accepted that PE is a heterogeneous disease, as highlighted by the complex clinical diagnostic criteria defining the disease (4). Increasing evidence supports the concept that the variability in clinical presentation of PE is at least in part due to the existence of distinct subtypes of PE. For example, it was reported that gene expression clustering in placental tissue can be used to define multiple unique molecular subtypes of PE (5). Further, a growing number of independent animal models of PE have been developed

in recent decades involving genetic, surgical, and pharmacological interventions targeting various distinct mechanisms and resulting in partially overlapping development of specific clinical features of PE (6–8). Due to the heterogeneity of clinical symptoms and molecular mechanisms involved in PE, there is currently no single comprehensive animal model that captures all the clinical and molecular features of PE (6–8). To understand the molecular mechanisms of the various subtypes of PE, it is important to continue to develop novel preclinical models of the disease.

In 2014, our group demonstrated that copeptin, a stable biomarker of arginine vasopressin (AVP) secretion (9), is elevated in women with PE as early as the sixth week of gestation (10). While this was the earliest gestational time point examined in any such study, other independent research teams around the globe have now similarly demonstrated elevations in copeptin both before and after the clinical diagnosis of PE using an array of commercially available assays (11–17), supporting the conclusion that AVP secretion is increased early and throughout pregnancies that eventually develop PE. Importantly, we have also demonstrated that infusion of AVP throughout gestation is sufficient to initiate major PE phenotypes in mice, including elevated systolic blood pressure (SBP), proteinuria, RGE, and intrauterine growth restriction (10). Nonetheless, the role of AVP in the pathogenesis of human PE and the mechanism(s) by which AVP leads to PE phenotypes in mice remain unclear.

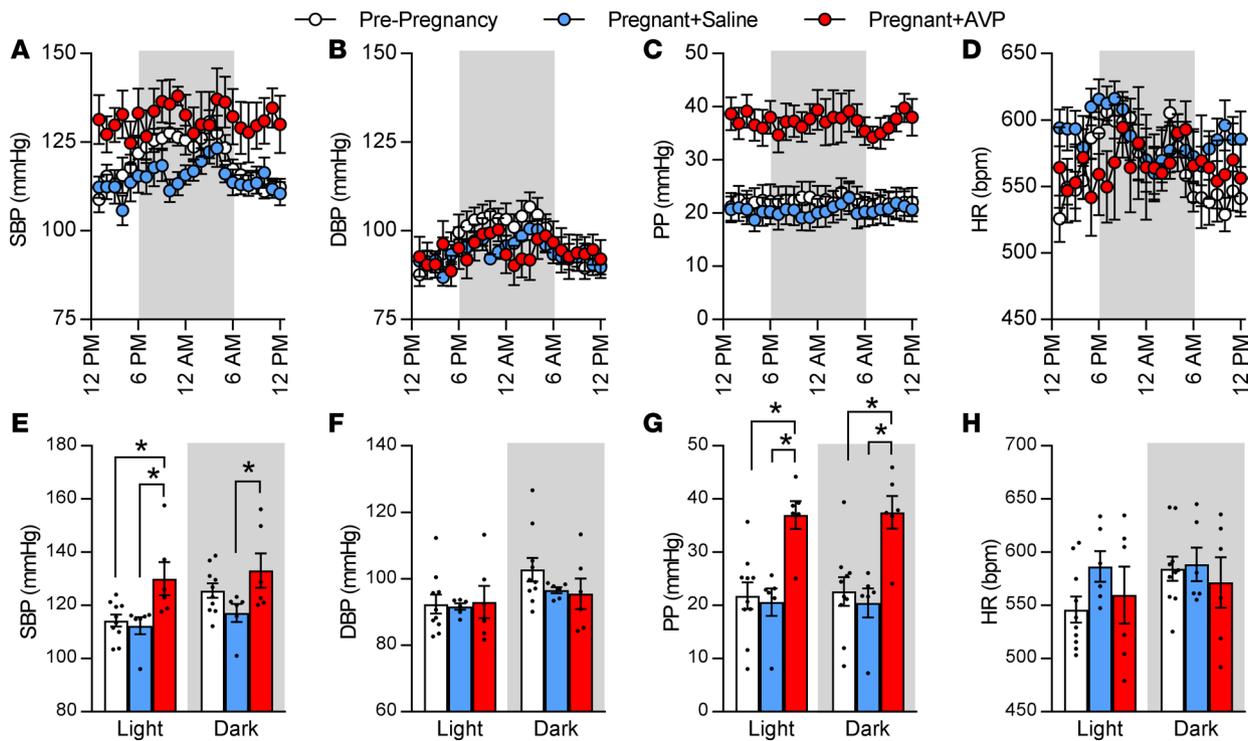
Therefore, the overall objectives of the current study were to explore the utility of AVP infusion to model the clinical syndrome of PE, and to clarify the receptor- and gestational time-dependent actions of AVP that elicit phenotypes of PE in mice.

## Results

**Model development.** Our first objective was to validate and extend the results of our previous initial characterization of the effects of AVP infusion throughout mouse pregnancy (10). Most importantly, we discovered that significant variation exists in the quality and quantity of AVP supplied between lots of AVP offered both within, and between, individual commercial vendors. Initial animal studies performed using distinct lots of AVP from multiple vendors resulted in variable induction of PE phenotypes in mice, which correlated with the quality and quantity of AVP supplied (data not shown). Therefore, careful analytical analyses, including mass-spectrometric confirmation of both the quantity and purity of AVP supplied, was required for every vial of AVP used for these studies (data not shown). Throughout the studies described below, Sigma-Aldrich AVP (catalog V9879), lot 025M4856V and 126M4842V were utilized. Sigma-Aldrich lot SLBF6741V and Phoenix Peptides (catalog 065-07) lot 432419 were specifically avoided. AVP was infused at 24 ng/hr into the subcutaneous space of the flank via osmotic minipump; this dosing schedule was selected based on a preliminary dose-response evaluation in which 0.24 and 2.4 ng/hr infusion rates had no discernable effects, while 240 ng/hr caused excessive fetoplacental death and resorption and/or prevented pregnancy (Supplemental Figure 1; supplemental material available online with this article; <https://doi.org/10.1172/jci.insight.99403DS1>). Subcutaneous infusion of AVP at 24 ng/hr resulted in a 47% increase in plasma AVP in AVP-infused dams compared with saline-infused dams (at gestational day 12.5 [GD 12.5], pregnant with saline infusion:  $n = 5$ ,  $33.7 \pm 5.1$ , vs. pregnant with AVP infusion:  $n = 5$ ,  $49.8 \pm 6.0$  pg/ml, 1-tailed  $t$  test  $P = 0.04$ ). Although nonpregnant human plasma AVP concentrations are typically lower, endogenous AVP is elevated to this level in mice, rats, and humans in both pregnant and nonpregnant states in response to various distinct challenges (18–20).

**Blood pressure.** To characterize the cardiovascular phenotypes of the AVP-infusion model of PE, we first examined blood pressure (BP) using radiotelemetric transducers. Compared with prepregnancy baseline, mice chronically infused with saline (0.11  $\mu$ l/hr, s.c.) exhibited no changes in SBP (Figure 1, A and E), diastolic BP (DBP) (Figure 1, B and F), pulse pressure (PP) (Figure 1, C and G), or heart rate (HR) (Figure 1, D and H) during the third week of gestation (GD 13.5–17.5). In contrast, mice chronically infused with AVP (24 ng/hr, s.c.) exhibited robust elevations in SBP and PP, with no changes in DBP or HR. Importantly, infusion of AVP in nonpregnant female C57BL/6J mice did not alter SBP, urine volume, or urine protein concentration (Supplemental Figure 2).

**Vascular function.** These data led us to hypothesize that infusion of AVP leads to vascular dysfunction and altered uterine and placental blood flow, as has been documented in PE pregnancies (21, 22) and in various models of the disease (23–27). Toward that end, we measured endpoints of arterial stiffness and resistance via pulse wave velocity (PWV) and Doppler ultrasonography during GD 10.5–13.5 in mice. Aortic PWV (Figure 2A), a measure of arterial stiffness, did not change with AVP infusion. Measures of uterine artery function, including peak systolic velocity (PSV) (Figure 2B), end diastolic velocity (EDV) (Figure 2C), or resistive



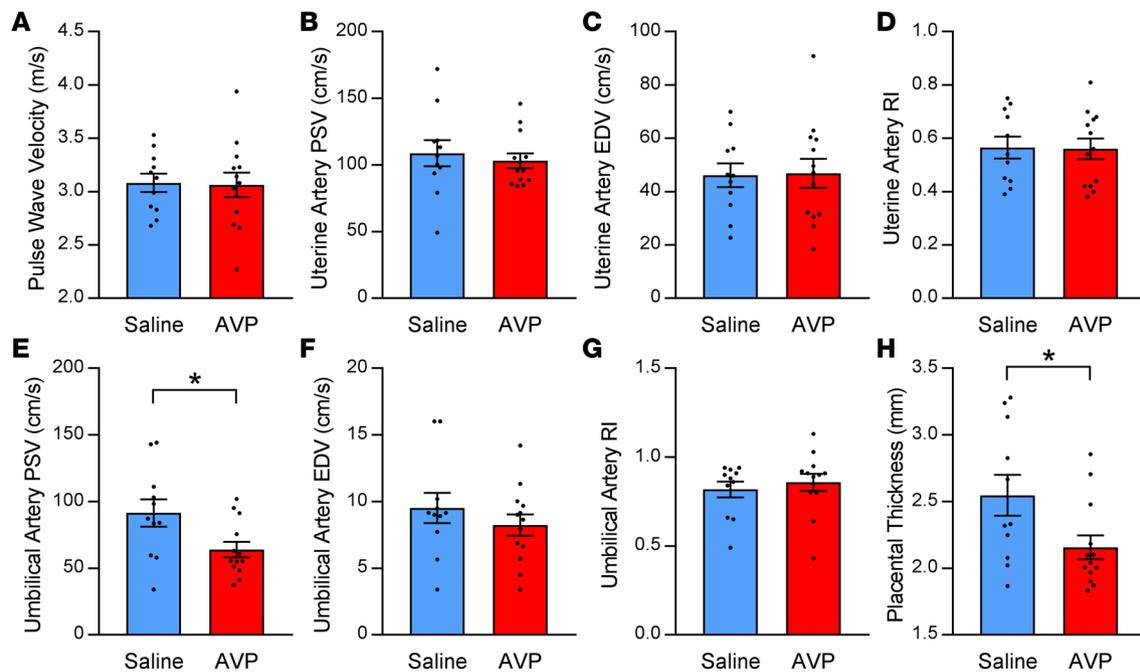
**Figure 1. Gestational arginine vasopressin (AVP) infusion causes an isolated systolic hypertension.** Effects of pregnancy and AVP on (A and E) systolic blood pressure (SBP), (B and F) diastolic blood pressure (DBP), (C and G) pulse pressure (PP), and (D and H) heart rate (HR). Analyses by 2-way ANOVA with repeated measures and Tukey's test for multiple comparisons. Data are expressed as the mean  $\pm$  SEM. \* $P < 0.05$ . Baseline  $n = 10$ , saline  $n = 6$ , AVP  $n = 6$ .

index (RI) (Figure 2D) were similarly unaffected by AVP infusion. In addition to uterine artery dysfunction, defects in umbilical artery flow have also been associated with adverse pregnancy outcomes (28–31). Specifically, multiple groups have demonstrated associations between decreased umbilical PSV and PE (32, 33), with others demonstrating associations between decreased, absent, or even reversed diastolic umbilical velocity and increased RI with uteroplacental insufficiency seen in PE (34, 35). In pregnant mice receiving AVP infusion, umbilical artery PSV (Figure 2E) was decreased and EDV (Figure 2F) and RI (Figure 2G) were unchanged.

**Placental development.** Interestingly, ultrasonography detected a significant decrease in placental thickness in mice receiving AVP infusion (Figure 2H). Therefore, we examined placental morphology at GD 12.5. According to Carnegie-stage comparisons (36), this time in mouse development approximately corresponds to day 40 of human embryologic development when many of the early mechanistic changes in PE are observed, and GD 12.5 has also been used previously to examine placental morphology in mouse models of PE (27). AVP-infused mice in this cohort exhibited early- and mid-gestational elevations in SBP ( $\Delta$ SBP from prepregnancy baseline: saline:  $-7.6 \pm 3.1$ , vs. AVP:  $+3.5 \pm 2.2$  mmHg,  $P = 0.01$ ) and spot urine protein concentration (saline:  $27.1 \pm 2.7$ , vs. AVP:  $44.1 \pm 2.5$  mg/ml,  $P < 0.01$ ). Placental layer thicknesses of the decidua and junctional zone at GD 12.5 were not changed by AVP, but the labyrinth layer, which contains fetus-derived capillaries, was smaller (Figure 3A). Spiral artery diameter (Figure 3B) and number per section (Figure 3C) were both decreased by AVP. Representative images of histological measurements are shown in Supplemental Figure 3.

**Placental mechanisms.** PE is typically characterized by poor spiral artery remodeling (37–40), possibly due to reductions in the angiogenic factor placental growth factor (PGF) (41–43) and increases in its inhibitor, fms-like tyrosine kinase-1 (FLT1) (44). Although PGF was decreased by AVP infusion (Figure 3D), placental FLT1 was also significantly downregulated by AVP infusion (Figure 3E). mRNA levels also appeared to decrease for both genes (*Pgf*: saline  $n = 18$ , 1.0 [0.7- to 1.3-fold] vs. *Avp*  $n = 21$ , 0.3-fold [0.2- to 0.4-fold] [ $\pm 1$  SEM],  $P < 0.05$ ; total *Flt1*: saline  $n = 18$ , 1.0 [0.9- to 1.2-fold] vs. *Avp*  $n = 21$ , 0.5-fold [0.4- to 0.7-fold] [ $\pm 1$  SEM],  $P = 0.06$ ).

Given the association of spiral artery morphology and decreased PGF with placental hypoxia (45–48), we hypothesized that infusion of AVP would lead to placental hypoxia, despite overall normal uterine artery velocity and resistance. We first examined the subcellular localization of hypoxia-inducible factor-1 $\alpha$

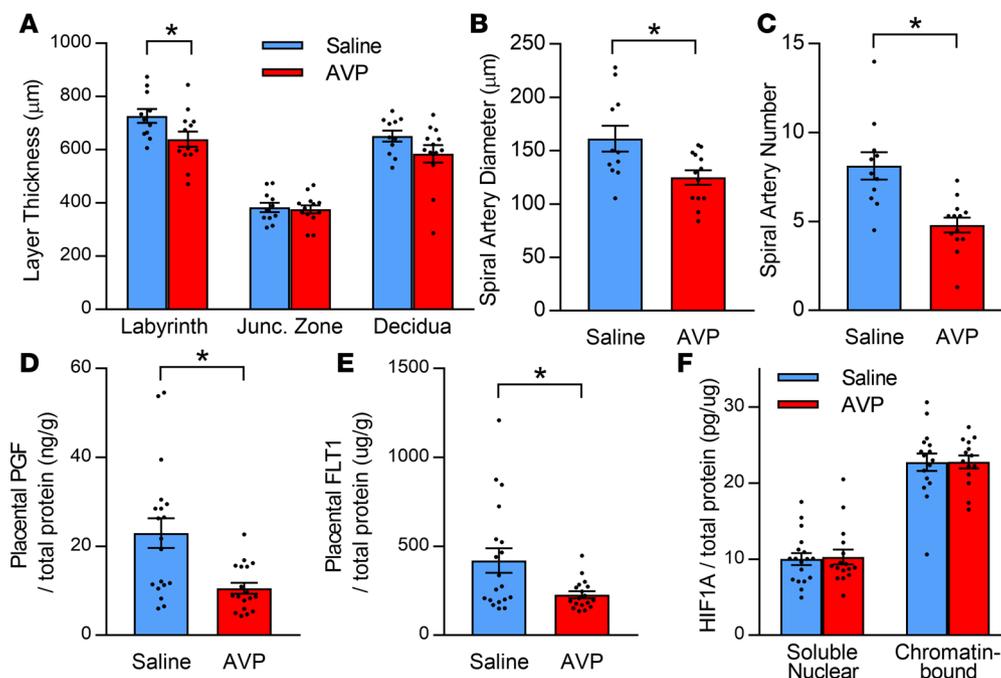


**Figure 2. Arginine vasopressin (AVP) has minor or no effect on blood flow velocity and resistance in uterine or umbilical arteries.** Effects of AVP on (A) aortic pulse wave velocity (PWV) as well as uterine artery (B) peak systolic velocity (PSV), (C) end diastolic velocity (EDV), and (D) resistive index (RI). Umbilical artery (E) PSV, (F) EDV, and (G) RI. (H) Fetal placental thickness by Doppler ultrasonography. Analyses by Student's *t* test. Data are expressed as the mean ± SEM. \**P* < 0.05. Saline *n* = 11, AVP *n* = 13.

(HIF1A), an important transcription factor that translocates to the nucleus and binds to DNA to initiate transcription in the absence of oxygen, in placental tissue at GD 12.5. We found no differences in HIF1A concentrations within soluble nuclear or chromatin-bound subcellular fractions between placentas from saline- and AVP-infused mouse pregnancies (Figure 3F). In addition, transcriptional markers of HIF1A activity were also unchanged or decreased by AVP at GD 12.5 or 17.5 (Supplemental Figure 4). Similarly, at GD 12.5 AVP had no effect on maximal invasion depth of cytokeratin-8-immunopositive cytotrophoblasts migrating into the decidual layer (Supplemental Figure 5), a process that has previously been demonstrated to be stimulated by local hypoxia (49–51).

**Placental gene expression.** To explore whether AVP affected gene expression and pathways implicated in PE, we performed RNA sequencing (RNA-Seq) on placentas from mice treated with saline or AVP. First, gene set enrichment analysis (GSEA) was performed to further explore whether AVP infusion resulted in placental hypoxia. Supporting our above results, we found no evidence of placental hypoxia using 2 independent hypoxia signatures (Table 1 and Figure 4A) (52). Subsequent GSEA of publicly available microarray data from human placentas compiled by Leavey et al. (GSE75010) (53) revealed that chronically hypertensive women with superimposed PE also did not exhibit enrichment for markers of placental hypoxia (Table 1), supporting the concept that placental hypoxia is variable across subtypes of human PE. Interestingly, despite the lack of enrichment for markers of hypoxia in the placentas from mice infused with AVP, GSEA found significant enrichment for markers of oxidative stress in the placentas (FDR *q* value = 0.05, family-wise error rate [FWER] *P* value = 0.02, normalized enrichment score [NES] = 1.39) (Figure 4B).

Next, using differential gene expression analysis, we identified 87 differentially expressed transcripts corresponding to 82 unique genes as well as 3 noncoding RNAs (Figure 4C). Of note, many of these genes were already implicated in human PE, as determined either by comparing against multiple previously published gene expression studies of human placental tissues from pregnancies complicated by preeclampsia (Figure 4D), and as determined by performing manual literature searches against the 82 genes that were differentially expressed in response to AVP treatment. Of the 82 differentially expressed genes (DEGs) reviewed manually, all but one exhibited directional changes similar to those documented in human PE (Table 2). That gene, *NEAT5*, is known to be elevated by hypoxia as well as during hyperosmolality (54), neither of which are detected in the AVP infusion model (Table 1 and Supplemental Figure 6, respectively).

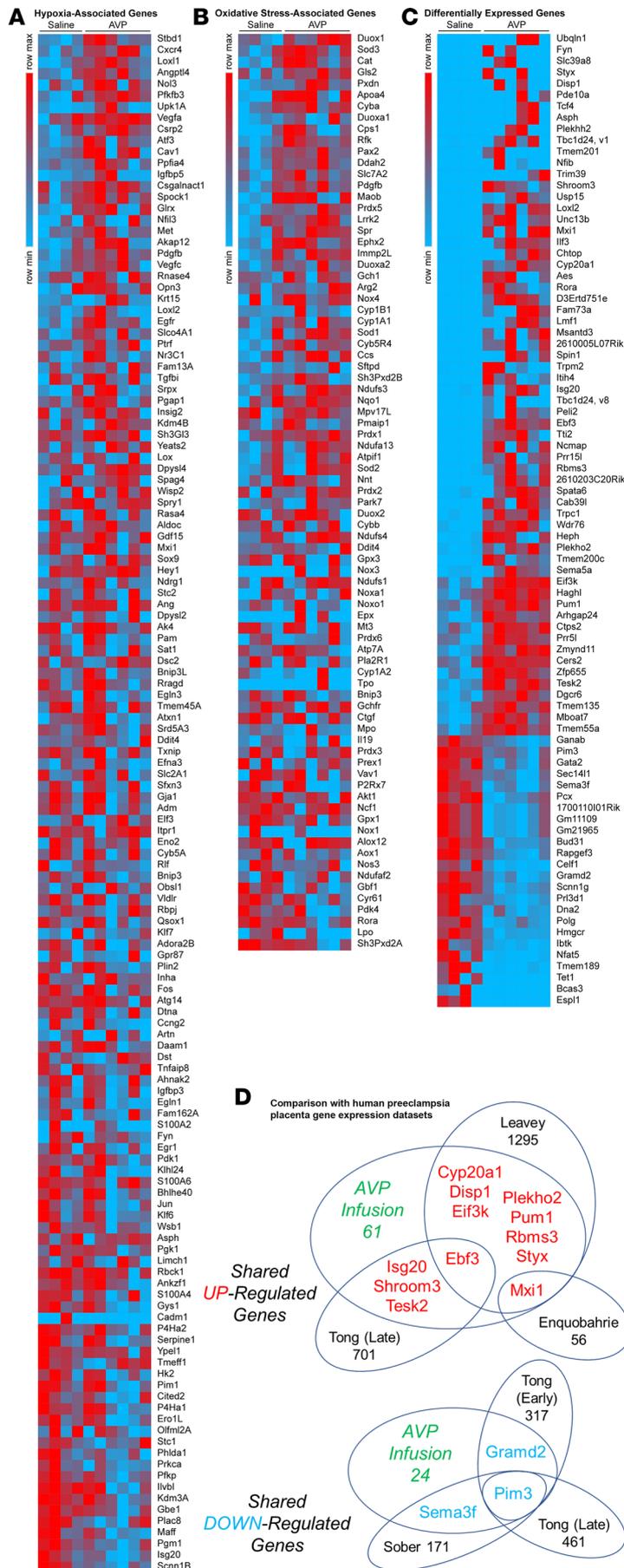


**Figure 3. Arginine vasopressin (AVP) alters placental morphology and angiogenesis at gestational day 12.5.** (A) Effects of AVP on thickness of placental labyrinth, junctional zone, and decidua (saline  $n = 11$ , AVP  $n = 13$ ). Measures of angiogenesis, including (B) spiral artery diameter and (C) spiral artery number (saline  $n = 11$ , AVP  $n = 13$ ), as well as angiogenic markers placental (D) PGF and (E) FLT1 (saline  $n = 20$ , AVP  $n = 20$ ). (F) HIF1A cellular localization in fractionated placentas collected from mice receiving saline or AVP (saline nuclear  $n = 18$ , chromatin  $n = 16$ ; AVP nuclear  $n = 16$ , chromatin  $n = 14$ ). Analyses by Student's  $t$  test. Data are expressed as the mean  $\pm$  SEM. \* $P < 0.05$ .

We also utilized Ingenuity Pathway Analysis (IPA) to identify molecular pathways that are modified in the GD 12.5 placenta by infusion of AVP (Table 3). We found that signatures for interleukin-2 (IL-2) signaling and calcium signaling, each of which have been implicated in PE (55–57), were both significantly enriched in placentas from AVP-infused pregnancies (Table 3). IPA also identified methylglyoxal pathways (58, 59), base excision repair mechanisms (60–67), and mevalonate pathways (68), which have also been associated with PE. Further, IPA revealed that the disease and functional annotations that were significantly enriched in placentas from AVP-infused pregnancies included hypertension, preeclampsia, as well as cell death (Supplemental Table 1). Finally, IPA identified upstream mediators that are expected to cause effects on the transcriptome similar to those observed with AVP infusion. As expected, this list

**Table 1. Enrichment for hypoxia signatures in RNA from GD 12.5 placentas of C57BL/6J mice infused with AVP (24 ng/hr) or saline, and placentas from human pregnancies reported in GSE75010**

Subgroup	Control	AVP or PE	GRD Hypoxia Score		Harris Hypoxia Score (52)	
			NES (PE vs. Con)	FDR $q$ value	NES (PE vs. Con)	FDR $q$ value
<u>Mouse</u>						
AVP-infusion	4	6	0.79	0.69	0.93	0.36
<u>Human</u>						
<b>No Chronic Hypertension</b>						
Preterm	12	19	1.71	0.01	1.57	<0.01
Term	18	6	1.73	0.02	1.63	0.02
<b>With Chronic Hypertension</b>						
Preterm	10	11	1.17	0.31	0.96	0.54
Term	10	3	1.05	0.43	1.11	0.30



**Figure 4. Gene expression profiling of placentas from dams infused with saline ( $n = 4$ ) or arginine vasopressin (AVP) ( $n = 6$ ) dams. (A)** Heatmap illustrating relative expression of genes associated with hypoxia (GRD Hypoxia gene set). **(B)** Heatmap illustrating relative expression of genes associated with oxidative stress (M15990 gene set). **(C)** Heatmap illustrating relative expression of 87 differentially expressed genes identified by DESeq2 (FDR < 0.1). **(D)** Venn diagrams illustrating up- and downregulated genes in AVP-infused mouse placenta (AVP Infusion) that are similarly changed in human placenta in pregnancies complicated by preeclampsia, as described in the text. Numbers indicate total number of genes significantly changed as noted in individual data sets (Leavey [ref. 5], early- or late-onset preeclampsia subsets by Tong [ref. 102], Enquobahrie [ref. 103], and Sober [ref. 103]), whereas genes noted in shared spaces of Venn diagrams are shared by at least 2 overlapping data sets.

included AVP and desmopressin (an AVPR2 receptor agonist) (Supplemental Table 2). It also identified calcium as a potential upstream mediator.

Next, to determine whether AVP could act directly upon the placenta, we measured expression of AVP receptors and other mediators in human and mouse placentas. Through in silico reanalysis of GSE75010, we confirmed that genes for AVP receptors AVPR1A, AVPR1B, AVPR2, cullin-5 (CUL5), and the oxytocin receptor (OXTR), to which AVP can bind and activate, are all present in human placentas (Figure 5A). We also confirmed that AVP, oxytocin (OXT), and LNPEP, the enzyme that degrades AVP, are expressed by human placentas (Figure 5A). Of these, AVP, AVPR1A, and OXTR were increased in pregnancies affected by PE, demonstrating dysregulated local AVP synthesis and signaling in PE (Figure 5B). In mouse placenta, the various AVP receptors, Oxt, and Lnpep were also expressed (Figure 5C), but none were modified by AVP infusion (Figure 5D). This may indicate that increases in human placental AVP system gene expression during PE are secondary to other stimuli such as genetics, epigenetics, hypoxia, or other mechanisms. HTR8/SVneo cells, an immortalized cell line derived from first-trimester, extravillous invasive human placental trophoblasts (69), also express AVP, AVPR1A, AVPR2, CUL5, and OXTR (Figure 5E). Importantly, a dose-dependent increase in intracellular  $Ca^{2+}$  concentration ( $[Ca^{2+}]_i$ ) was observed in response to AVP (Figure 5F) ( $EC_{50} = 440 \pm 200$  nmol/l), demonstrating that AVP can directly activate intracellular calcium signaling cascades in trophoblasts.

**Receptor and time dependency of AVP action: BP.** We next examined how AVP infusion leads to PE phenotypes by clarifying the role(s) of various AVP receptors and the times of gestation they are activated during pregnancy. To do so, we performed pharmacological studies to block specific AVP receptors as outlined in Figure 6A. Briefly, we infused saline (Early) with or without the AVP receptor antagonists, conivaptan (nonselective AVPR1A and AVPR2 antagonist), relcovaptan (AVPR1A antagonist), and tolvaptan (AVPR2 antagonist) through GD 17.5. In other treatment groups, we infused AVP only through GD 2.5 or GD 9.5 to determine whether AVP action early in pregnancy is sufficient to initiate the development of PE phenotypes. Mice infused with

**Table 2. Differentially expressed RNA transcripts within AVP-treated GD 12.5 placenta that have previously been experimentally associated with human PE**

Transcript	AVP model	Human PE	References
<i>Trpm2</i>	↑	Increased in maternal urine at 20 weeks. Increased in maternal plasma in first and second trimesters and also maternal serum at term, most strongly in cases with IUGR; Protein evident in syncytiotrophoblasts and villous endothelial cells and also increased in PE placenta	(105–110)
<i>Itih4</i>	↑	Evaluated within a panel of predictive biomarkers within maternal serum	(111)
<i>Ncmamp</i>	↑	Increased in microarray analysis of PE placenta	(112)
<i>Sema3f</i>	↓	Reduced in PE placenta, maternal serum, amniotic fluid, and cord blood at delivery; Correlates with fetal and placental masses; At 16–18 weeks, reduced in amniotic fluid and predictive of PE (AUC = 0.941)	(113)
<i>Scnn1g</i>	↓	Reduced in PE placenta	(114)
<i>Nfat5</i>	↓	Increased in hypoxic human PE placenta, in animal models of hypoxic PE, and immortalized trophoblasts with hypoxic or hyperosmotic stimuli	(54, 115, 116)

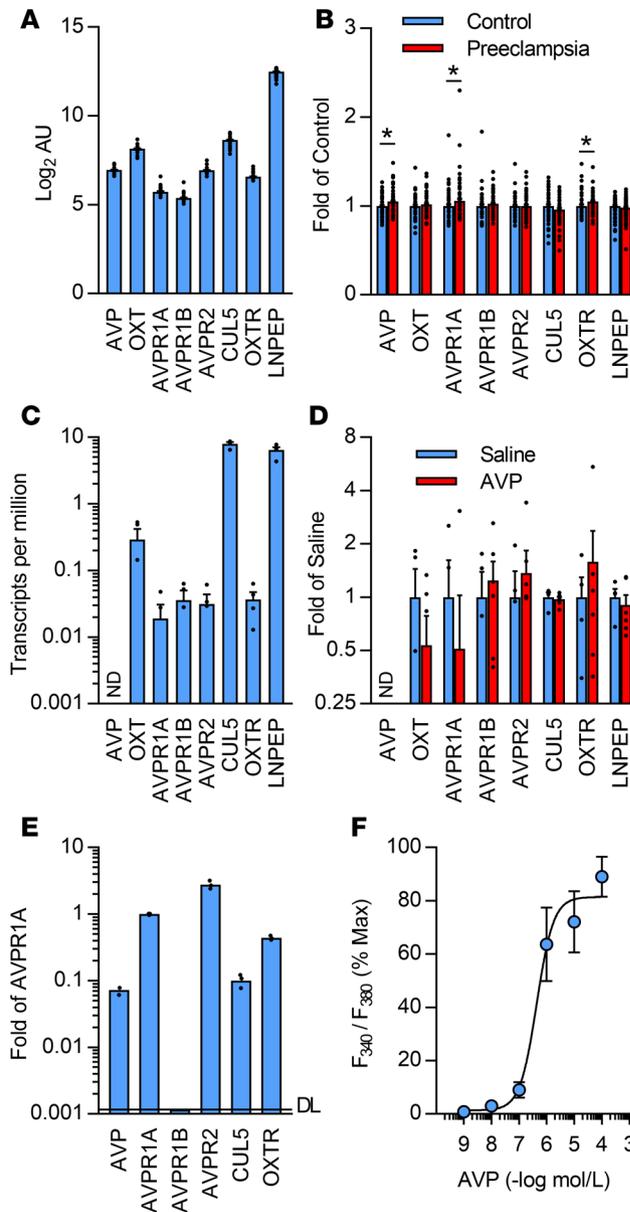
saline exhibited a mid-gestational decrease in SBP that returned to baseline levels at the end of pregnancy. In contrast, SBPs of mice infused with AVP failed to decrease in mid-gestation and then increased above baseline at the end of gestation (Figure 6B). Chronic coadministration of the nonspecific AVPR1A+AVPR2 receptor antagonist, conivaptan, completely prevented AVP-induced changes in SBP. However, coadministration of the selective AVPR1A antagonist, relcovaptan, had no effect on SBP in AVP-infused mice. Interestingly, coadministration of the selective AVPR2 antagonist, tolvaptan, also had no effect on SBP in AVP-infused mice in mid-gestation, but completely ameliorated the SBP elevations in late-gestation. Although SBP was elevated in mice solely infused with tolvaptan in late-gestation, chronic infusion of receptor antagonists in the absence of exogenous AVP infusion had no major effect (Supplemental Figure 7). Collectively, these findings support the concept that AVP elevates SBP in pregnancy through a combination of AVPR1A+AVPR2 receptor activation in early- and mid-gestation and AVPR2-specific activation in late-gestation.

To further examine the gestational time-dependent effects of AVP on SBP, we examined the consequences of only infusing AVP during specific phases of gestation. Whereas infusion of AVP throughout pregnancy (to GD 17.5) resulted in both a failure to reduce SBP in mid-gestation and an elevation in SBP in late-gestation, infusion of AVP only up to GD 2.5 had no effect on SBP at any examined time point (Figure 6C). Interestingly, infusion of AVP only through GD 9.5 resulted in a failure to reduce SBP in mid-gestation (similar to AVP infusion throughout pregnancy), but SBP returned to match that of saline-infused controls by late-gestation. These findings support the idea that SBP is increased by AVP throughout gestation and that the continued action of AVP is required to maintain increases in SBP.

*Receptor and time dependency of AVP action: Renal function.* To characterize the receptor- and time-dependent effects of AVP on renal and fetal phenotypes, we measured urine protein, assessed glomerular damage by electron microscopy, and measured fetal weight near the end of gestation (GD 17.5). As previously demonstrated, GD 17.5 urine protein concentration was elevated by AVP infusion throughout pregnancy (Figure 7A). Unexpectedly, given the established diuretic effect of AVP in the nonpregnant state (70),

**Table 3. Canonical pathways changed by AVP infusion**

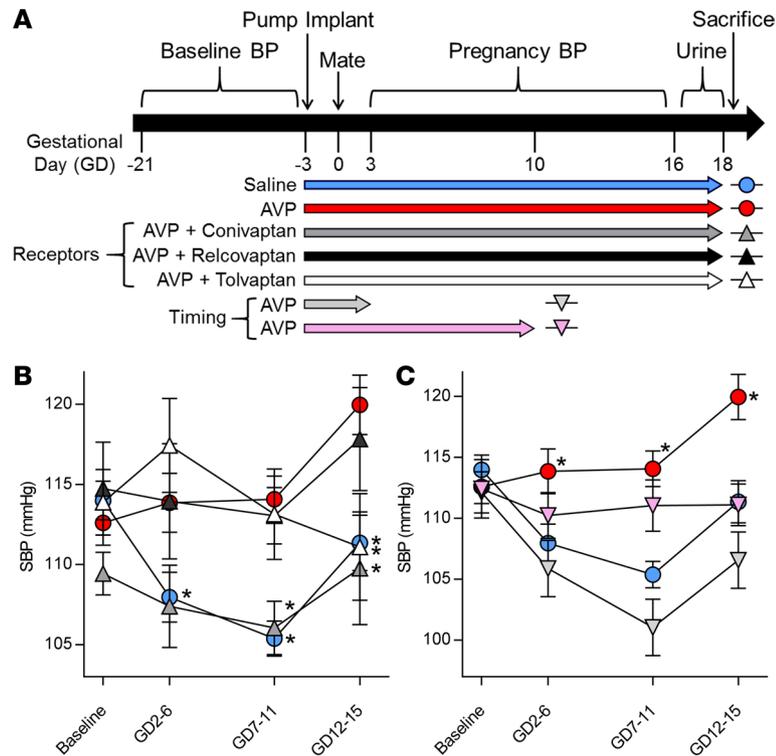
Ingenuity Canonical Pathways	P value
Methylglyoxal Degradation I	0.010
Calcium Signaling	0.026
Regulation of IL-2 Expression in Activated and Anergic T Lymphocytes	0.032
Base Excision Repair (BER) pathway	0.042
Mevalonate Pathway I	0.045
Superpathway of Inositol Phosphate Compounds	0.050



**Figure 5. Components of arginine vasopressin (AVP) system are present in placentas and trophoblasts.** (A) AVP-system components are present in human placentas. (B) Relative abundance of AVP-system components in preeclampsia compared with control pregnancies (control  $n = 77$ , preeclampsia  $n = 80$ , GSE75010). (C) Presence of AVP-system components in mouse placenta and (D) response of those components to AVP (saline  $n = 4$ , AVP  $n = 6$ ). (E) Presence of AVP-system components in HTR8 human placental trophoblasts and (F) intracellular  $Ca^{2+}$  response of HTR8 cells to AVP ( $n = 4$  distinct passages). Increases in intracellular  $Ca^{2+}$  concentration ( $F_{340}/F_{380}$ ) were evoked by application of increasing concentrations of AVP. For each sample,  $F_{340}/F_{380}$  responses for each cell were normalized to the amplitude of maximal AVP concentration. Analyses by Student's  $t$  test. Data are expressed as the mean  $\pm$  SEM. \* $P < 0.05$ .

this dose of AVP significantly reduced water intake (Supplemental Figure 8A) without affecting urine volume (Supplemental Figure 8B). Elevations in urine protein concentration were completely prevented by relcovaptan but not tolvaptan, demonstrating an important role for AVPR1A in this phenotype. Relcovaptan also prevented increases in 24-hour urine protein caused by AVP (Figure 7B). Urine protein effects of conivaptan were not interpretable, as conivaptan independently caused renal damage (Supplemental Figure 8, C and E). In the absence of AVP, relcovaptan and tolvaptan had no effect on urine protein or glomerular morphology (Supplemental Figure 8, C–E). Interestingly, AVP infusion through GD 9.5 but not 2.5 was sufficient to elevate urine protein at GD 17.5 (Figure 7A), although there were no differences in 24-hour urine protein (Figure 7B). Finally, blood was collected via submandibular lance into tubes coated with lithium-heparin from a subset of GD 17.5 dams to examine common markers of renal function. AVP infusion had no significant effect on major electrolytes, glucose, or markers of renal function, as determined by a handheld clinical chemistry analyzer (iSTAT) (Supplemental Table 3).

*Receptor and time dependency of AVP action: Fetal development.* AVP infusion led to decreases in fetal weight that were prevented by tolvaptan (Figure 7C). Low fetal weight appears to be dependent on late-gestation AVP action, as infusion of AVP only through GD 2.5 or 9.5 had no effect on fetal weight. Interestingly, although AVP alone had no effect on placental mass, tolvaptan coinfusion reduced placental



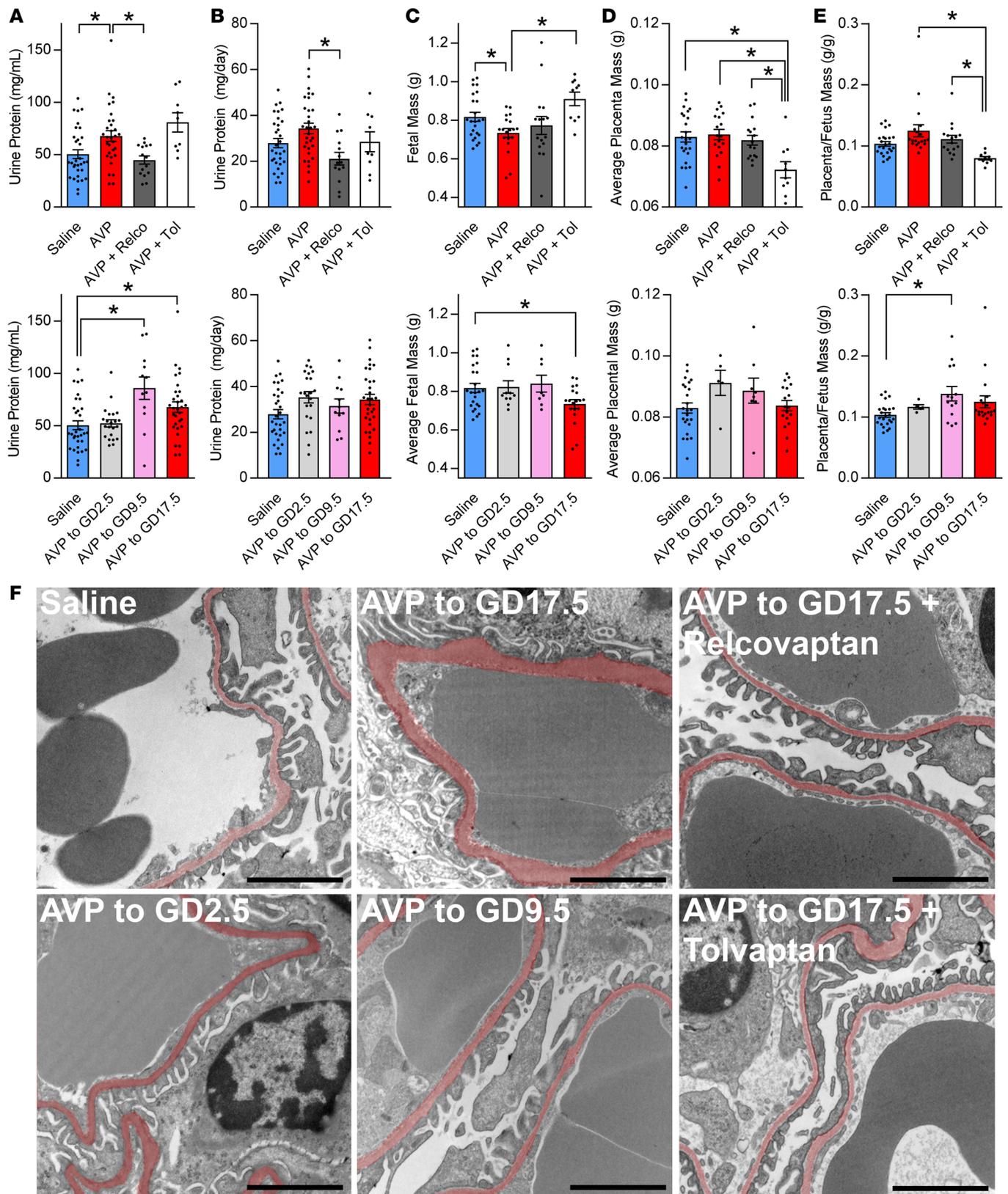
**Figure 6. Experimental design, and blood pressure effects of arginine vasopressin (AVP) and AVP-antagonist coinfusion.** Our experimental design is shown in **A** and described in the text. **(B)** Effects of saline ( $n = 40$ ), AVP ( $n = 27$ ), AVP + conivaptan ( $n = 11$ ), AVP + relcovaptan ( $n = 9$ ), and AVP + tolvaptan ( $n = 11$ ) throughout pregnancy. Analyses by 3-way ANOVA followed by Tukey's test for multiple comparisons. \* $P < 0.05$  versus saline. **(C)** AVP infusion through only GD 2.5 ( $n = 18$ ) and GD 9.5 ( $n = 12$ ). Saline and AVP shown again for clarity. Analyses by 3-way ANOVA followed by Tukey's test for multiple comparisons. Data are expressed as the mean  $\pm$  SEM. \* $P < 0.05$  versus AVP.

mass compared with all other treatment groups (Figure 7D). Tolvaptan therefore significantly reduced the fetal/placental mass ratio when coinfused with AVP throughout gestation (Figure 7E).

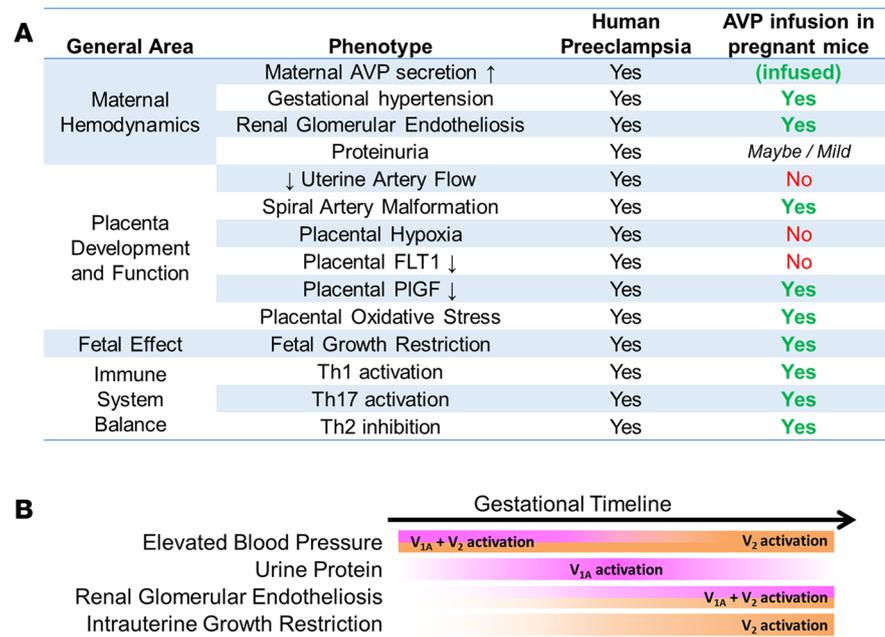
*Receptor and time dependency of AVP action: Renal structure.* RGE, a pathognomonic histological observation for PE characterized by endothelial swelling in maternal glomeruli, was again observed in mice receiving AVP throughout pregnancy (Figure 7F), similar to our previous report (10), when glomeruli were examined at high magnification by electron microscopy. Interestingly, mice coinfused with AVP and relcovaptan had no detectable glomerular damage, whereas mice coinfused with AVP and tolvaptan exhibited a modest protection of glomerular structure. Additionally, AVP infusion through GD 2.5 or 9.5 was not sufficient to induce RGE, indicating that the action of AVPR1A (and to a lesser degree, AVPR2) late in pregnancy is required for RGE in this model. Unfortunately, nonspecific combined antagonism of both AVPR1A and AVPR2 was again uninterpretable, as conivaptan itself without AVP coadministration caused renal ultrastructural damage (Supplemental Figure 8E). Major structural changes to the glomeruli were not as obviously evident when examined using hematoxylin and eosin (H&E) staining and light microscopic, low-magnification imaging methods (Supplemental Figure 9).

*Hepatic complications.* Finally, AVP is known to act through its AVPR1A receptor to modulate protein catabolism, lipid metabolism, bile acid synthesis, and glucose homeostasis through actions at the liver (71–73), and thus we tested the possibility that excess AVP may contribute to hepatic dysfunction during pregnancy, observed during complications such as HELLP syndrome. No differences were observed in ALT or AST activities between plasmas from saline- or AVP-infused dams on GD 17.5 (Supplemental Figure 10), leading us to conclude that the 24 ng/hr, s.c. dosing schedule of AVP is insufficient to model hepatic complications and the HELLP syndrome.

*Summary.* These data, along with findings we recently reported regarding the effect of the 24 ng/hr infusion of AVP upon the immune system (74), are summarized in Figure 8A. Further, our interpretation of the gestational time-dependent involvement of various AVP receptors in the development and maintenance of



**Figure 7. Renal and fetal effects of arginine vasopressin (AVP) and AVP-antagonist coinfusion. (A)** Effects of saline ( $n = 32$ ), AVP ( $n = 30$ ), AVP + relcovaptan ( $n = 15$ ), AVP + tolvaptan ( $n = 9$ ), and AVP to GD 2.5 ( $n = 22$ ) and GD 9.5 ( $n = 12$ ) on urine protein concentration. **(B)** Twenty-four-hour urine protein, **(C)** fetal mass, **(D)** placental mass, and **(E)** placenta/fetus mass ratio effects of saline ( $n = 24$ ), AVP ( $n = 19$ ), and receptor combinations (same  $n$ 's as panel **A**). For panels **A-E**, saline and AVP-treated mice are reported twice for clarity. **(F)** RGE with AVP, as indicated by the thickened and distorted glomerular basement membrane (pink) that was prevented by relcovaptan ( $n = 3$  for all groups). Scale bars: 2  $\mu$ m. Analyses for receptor inhibitor studies by 2-way ANOVA with Dunnett's multiple-comparisons procedure, compared with AVP. For timing studies, analyses by 1-way ANOVA and Dunnett's multiple-comparisons procedure, compared with saline. Data are expressed as the mean  $\pm$  SEM. \* $P < 0.05$ .



**Figure 8. Summary and working model.** (A) Table summarizing major phenotypes of human preeclampsia, and the sufficiency of the AVP infusion (24 ng/hr, s.c.) in pregnant wild-type C57BL/6J mice to phenocopy each, as evaluated in the current manuscript plus our recent publications (10, 104). (B) Working model, based on data presented herein, illustrating the relative involvement of AVPR1A and AVPR2 receptors at various stages of gestation, in the development of blood pressure, renal, and fetal phenotypes in the AVP infusion model.

major clinical features of PE are graphically depicted in Figure 8B. Elevations in SBP and PP appear to be mediated by AVPR1A and AVPR2 in early- to mid-gestation, and this shifts to only AVPR2 in late gestation. Urine protein concentration seems to be increased by mid-gestational AVPR1A activation, while RGE requires both AVPR1A and AVPR2 activation near the end of gestation. Finally, fetal growth restriction appears to be driven primarily by late-pregnancy AVPR2 activation.

### Discussion

AVP secretion, as measured by plasma copeptin, has now been shown by multiple groups around the world to be elevated in the first trimester and throughout gestation in PE (10–17). While it is difficult to directly test the hypothesis that AVP can cause human PE, insights gained from studies of obstetric outcomes in subjects with AVP-associated disorders support a potential association between elevated AVP secretion and PE. For example, Nawathe and Govind recently reviewed cases of syndrome of inappropriate antidiuretic hormone (SIADH) during pregnancy and found that 13 of 18 reported cases developed PE (75). In addition, other disorders associated with elevated AVP secretion, such as diabetes mellitus (76), obesity (77), and hypertension (78) are all established risk factors for PE. These findings support an association between AVP and PE and prompt studies to test a causal role for AVP in the development of PE. Here we show that low-dose infusion of AVP in mice is sufficient to cause the physiological phenotypes of PE, including hypertension, RGE, and intrauterine growth restriction (IUGR), and also induces many of the cellular and molecular phenotypes of PE, including decreased placental PGF, placental oxidative stress, and decreased spiral artery number and diameter. Additionally, we documented a complex participation of AVPR1A and AVPR2 receptors at specific time points throughout gestation in the development of physiological phenotypes of PE in this model.

Despite observing decreases in spiral artery diameter as well as number and the known effects of AVP on the vasculature in the nonpregnant state (79–81), we were surprised that AVP did not induce placental hypoxia or changes in uterine artery velocity or resistance. Placental hypoxia has long been associated with PE (82) and is an essential etiology for multiple models of the disease (6–8). However, recent studies by Leavey et al. have identified populations of patients with PE that do not exhibit molecular signatures consistent with hypoxia (5). Acknowledging the relatively small sample size

included, reanalysis of the GSE75010 data set similarly supports differential enrichment of markers for hypoxia in subsets of humans with PE, such as those with or without chronic hypertension before pregnancy (Table 1). Therefore, there appears to be heterogeneity in the development of placental hypoxia during PE, indicating that additional complementary mechanisms must also contribute to the pathogenesis of PE.

Thus, our data support the conclusion that AVP is sufficient, independently from hypoxia-mediated mechanisms, to induce clinical phenotypes of PE. This conclusion, along with the observations that most pregnancies complicated by PE are characterized by both some degree of placental hypoxia and elevated secretion of AVP, supports the concept that AVP and hypoxia represent dissociable mechanisms that may independently and/or synergistically contribute to the pathogenesis of human PE. These data and concepts fit well within the current understanding of the molecular mechanisms of PE, specifically that aspects thought to be important in the pathogenesis of PE, such as proinflammatory and dysangiogenic mechanisms, are independent of hypoxia (83–85). In addition, hypoxia markers like HIF1A can be induced in placental cells under normoxic conditions via inflammatory pathways (86) in the absence of hypoxia. Clinically, the existence of at least 2 major, independently contributing mechanisms may therefore help explain the heterogeneity of symptom development in human PE.

The lack of evidence for the induction of placental hypoxia by chronic AVP infusion during pregnancy in mice indicates that this manipulation does not fully model typical human PE. On average, human PE is characterized by placental hypoxia, but that observation does not preclude the existence of individual PE patients who do not exhibit placental hypoxia. It is important to appreciate that both in the current study (Supplemental Figure 2) and in our previous study (10), chronic low-dose (24 ng/hr, s.c.) infusion of AVP into nonpregnant female mice did not cause hypertension; this effect is only unmasked in the context of pregnancy. Thus, whether AVP infusion represents a comprehensive model of a subtype of human PE or a partial model of all types of PE (when performed in the context of placental hypoxia) requires further investigation, though the utility of the AVP infusion intervention to model pregnancy-related hypertensive disease and its sequelae remains.

The determination that both AVPR1A and AVPR2 have nonoverlapping, complementary, and time-dissociable actions to mediate the development of PE phenotypes in mice implies that treatment regimens targeting patients with AVP-induced PE symptoms would need to antagonize combinations of both AVPR1A and AVPR2 throughout gestation. While other AVP receptors, such as AVPR1B, may play a role in the development of PE phenotypes, the robust prevention of PE phenotypes in mice by AVPR1A and AVPR2 antagonists supports only minor roles for these alternative receptors. This finding may support relcovaptan and tolvaptan as novel preventative agents against AVP-mediated subtype(s) of PE. However, the effects of these antagonists in human pregnancies and whether phenotype reversal (rather than prevention, as in our studies) is possible are important questions that need to be answered before definitive clinical trials could be initiated.

Instead of therapeutically targeting AVPR1A and AVPR2, it may be simpler and more direct to target synthesis or release of AVP. It is known that AVP secretion increases early in normal pregnancy, but the mechanisms behind this process are not clear and may be modified in PE (19). Future studies aimed at understanding the mechanism(s) of exaggerated AVP secretion during human PE may therefore lead to the discovery of earlier and even more robust biomarkers, and also better inform AVP-centric treatment approaches. In particular, the development of conditional (i.e., floxed) alleles for each of the AVP receptors is required to permit tissue-specific, complete disruption of individual receptors to implicate specific receptor and tissue targets in the development of individual phenotypes. For example, such methods are required to selectively disrupt AVP receptors in individual cell types of the placenta to probe the role of these receptors in the development of PE phenotypes in response to AVP infusion. Such studies can be directed based on insights gained by the pharmacological approaches utilized in the current study.

In summary, we have determined that infusion of AVP throughout mouse pregnancy is sufficient to initiate many of the major phenotypes of PE. Phenotype development is mediated through synergistic activation of multiple AVP receptors at various stages throughout gestation, can be prevented by AVP antagonists, and is independent from placental hypoxia. These data support the concept that interference with AVP secretion or action may provide a therapeutic approach in patients with PE.

## Methods

**Animal subjects.** Experiments were conducted in 10- to 20-week-old female C57BL/6J mice obtained from the Jackson Laboratories. Animals were maintained in conventional housing, including a standard 12-hour light-dark cycle and an ambient temperature of 25°C, with ad libitum access to tap water and chow (Teklad, 7013).

**BP by radiotelemetry.** BP was recorded by telemetric probe as previously described (87–91). Radiotelemetric probes (DSI, model TA11PA-C10), were implanted into the common carotid artery under 2% isoflurane anesthesia and animals were allowed to recover for at least 1 week before recording.

**Pregnancy studies.** Baseline BPs were recorded via tail-cuff plethysmography as previously described (10). Mice were individually placed in restraint tubes and warmed to promote tail blood flow. Mice were acclimated for 10 days before 5 days of baseline measurements, which were averaged within subject for a final baseline SBP measurement. Mice were balanced between groups based on SBP, implanted with a micro-osmotic pump, and allowed to recover for 3 days before mating. Males were put in female cages for 1 night and BP recordings by tail-cuff resumed on GD 2.5. On GD 15.5, mice were transferred to metabolic cages for 48 hours for two 24-hour urine collections and then sacrificed on GD 17.5 for tissue and plasma collection.

**Drug preparation and infusion.** AVP (24 ng/hr, Sigma-Aldrich, V9879), conivaptan (Vaprisol, 22 ng/hr, Baxter Healthcare), relcovaptan (22 ng/hr, SR 49059) (Sigma-Aldrich, S5701), and tolvaptan (22 ng/hr, Sigma-Aldrich, T7455) were kept at –20°C until reconstituted. AVP and tolvaptan were reconstituted and aliquoted in saline, while relcovaptan was reconstituted and aliquoted in saline with 10% DMSO. Reconstituted drugs were kept at –80°C until use. Dose of conivaptan was determined by dose-response studies (data not shown) and relcovaptan and tolvaptan doses were calculated to correspond to the  $K_i$  of conivaptan at AVPR1A and AVPR2, respectively. Micro-osmotic pumps (Alzet model 1004) were filled with corresponding drug for appropriate infusion rate per manufacturer instructions and implanted 3 days prior to mating to allow for surgical recovery. For mice receiving AVP plus an antagonist, 2 pumps were placed, one containing AVP and the other containing antagonist, on opposite flanks. For studies infusing AVP only through GD 2.5 or GD 9.5, micro-osmotic pumps were used that only infused AVP for 1 week (Alzet model 1007D) or 2 weeks (Alzet model 1002), respectively.

**PWV measurement.** PWV was measured in mice between GD 13 and 19 using Doppler ultrasound (MouseDoppler, Indus Instruments) as previously described (92, 93). Mice were placed in a supine position under 2% isoflurane/1% oxygen anesthesia and continuous ECG recordings were collected from the paws. A 20-MHz probe was used to image at the descending aorta and, simultaneously, at the abdominal aorta 1 mm above the exit to the left renal artery. Arrival and transit times were calculated over 5–7 cardiac cycles. The distance between the probes was measured using a caliper. Distance between probes (m) divided by transit time (s) gave PWV (m/s).

**Doppler ultrasonography.** Abdominal ultrasounds were performed on GD 11–14 by a single blinded investigator. Immediately prior to ultrasound, mice were sedated with midazolam 0.1 mg by s.c. injection. The anterior thorax and abdomen were shaved. The animal was grasped gently by the nape of the neck and cradled in the left lateral recumbent position in the imager's hand. Warmed gel was applied to the ultrasound probe to improve the acoustic interface. Uterine and fetal sonography was performed using a 40-MHz linear array probe coupled to a Vevo 2100R imaging system (VisualSonics), generating approximately 180–200 two-dimensional (2-D) frames per second. Fetal cardiac activity and maternal uterine and umbilical artery flow velocity were visualized using 2-D images and color Doppler. Pulse-wave Doppler evaluation was used to measure flow velocity in the umbilical arteries, and in small branches of the uterine arteries adjacent to the gestational sacs. Resistive index was calculated as  $1 - (1/\text{pulsatility index})$ , with pulsatility index defined as peak systolic divided by end diastolic velocity by Doppler. All imaging and image analyses were performed by expert technicians blinded to treatment groups (KAZ and NCB).

**AVP measurement.** Mice at GD 12.5 were sacrificed by live decapitation and collected blood was placed in EDTA tubes before being quickly aliquoted into tubes containing aprotinin (0.6 trypsin inhibitory units per 1 ml blood) according to protocol (Phoenix Pharmaceuticals, RK-APRO). After 30 minutes on ice, tubes were centrifuged at 4°C at 5,000 g for 10 minutes and supernatant collected.

Plasma was then extracted by reverse-phase column. Briefly, we combined 100  $\mu$ l of 2 similarly treated plasma samples and diluted the combined plasma with an equal volume of 8% acetic acid. This was then loaded onto a C18 Sep-Pak cartridge (1 ml, 50 mg; Waters) that had been primed with 3 ml methanol followed by 3 ml deionized water using a vacuum manifold. After plasma sample was loaded, the cartridge was washed with 3 ml 4% acetic acid and peptides were eluted with 2 ml of a 3:1 acetonitrile/4% acetic acid solution.

Extract was frozen overnight and then dried with a SpeedVac (Savant Instruments). Extract was reconstituted in RIA diluent and AVP measured in samples by radioimmunoassay (Phoenix Pharmaceuticals, RK-065-07).

**Placental histology.** Morphology of the placenta was assessed using methods similar to those reported by Monk, et al. (94). Briefly, mice were euthanized by CO<sub>2</sub> asphyxiation and fetoplacental units fixed within the uterus via immersion in 10% neutral buffered formalin (NBF). Fixed tissues were grossly sectioned in half to see all layers of the placenta followed by routine processing and embedding. Tissues were sectioned at 5 μm thickness, and routinely stained with H&E. Placental morphology was evaluated using an Olympus BX51 with attached microscope camera (Olympus DP71) and Olympus CellSens software. Placental layer thicknesses were determined on 2 separate yet adjacent 5-μm-thick tissue sections. Four to 7 fetoplacental units per dam, and 4 to 6 dams per group were analyzed. The same tool was utilized to measure the widest diameter of the spiral arteries on the same slides. The number of spiral arteries within the decidual layer was manually counted on low magnification (×2) throughout the entire section of placenta. All slides were evaluated by a board-certified veterinary pathologist (KNGC) blinded to treatment conditions.

**Cell culture and [Ca<sup>2+</sup>]<sub>i</sub> single-cell imaging.** HTR8/SVneo (HTR8) cells, purchased from ATCC (CRL-3271) (69, 95), were cultured in RPMI medium supplemented with 10% FBS and 1% penicillin/streptomycin. Cells were grown in standard growth conditions at 37°C and 5% CO<sub>2</sub> and were plated on poly-D-lysine-coated (Sigma-Aldrich, P6407) glass cover slides 48 hours prior to the beginning of the experiment. On the day of experiment, cells were loaded with the ratiometric Ca<sup>2+</sup> indicator dye Fura-2/AM (2 μM) for 25 minutes. Cells were then placed in a flow-through perfusion chamber mounted on an inverted IX-71 microscope (Olympus) and perfused with standard extracellular HEPES-buffered Hanks' salt solution (HH buffer) prior to stimulation with AVP. Fura-2 fluorescence was alternately excited at 340 and 380 nm (12-mm bandpass) using the Polychrome V monochromator (TILL Photonics) and a ×20 objective (NA 0.75; Olympus). Emitted fluorescence was collected at 510 nm (84-nm bandpass) using an IMAGO CCD camera (TILL Photonics) as previously described (96). The fluorescence ratio ( $R = F_{340}/F_{380}$ ) was used as an index of [Ca<sup>2+</sup>]<sub>i</sub>.

**Renal ultrastructure.** Kidneys were collected on GD 17.5 and placed in glutaraldehyde solution. Electron microscopic analysis of maternal kidneys was performed at the University of Iowa Department of Pathology using a JEOL JEM-1011 transmission electron microscope and images were interpreted by a board-certified veterinary pathologist (KNGC) blinded to treatment conditions.

**HIF1A localization.** Placentas from mice receiving saline or AVP throughout pregnancy were collected on GD 17.5 and underwent subcellular fractionation per kit protocol (Thermo Fisher Scientific, 87790). HIF1A was subsequently detected by ELISA (LifeSpan Biosciences).

**Plasma osmolality.** Osmolality was measured using a Fiske 2400 Multi-sample Osmometer. Plasma was collected as described above, but in the absence of aprotinin.

**Real-time quantitative PCR.** RNA from placentas of pregnant GD 12.5 mice were isolated using a *mirVana* RNA isolation kit (Thermo Fisher Scientific, AM1560) per manufacturer's instructions. Quality assessment and RNA concentration was determined using a Nanodrop Spectrophotometer 2000. Quantitative real-time PCR assays were performed using TaqMan Gene Expression Assays (Thermo Fisher Scientific) or SYBR Green (IDT) primer sets. TaqMan primers: *18S* (catalog 4333760F), *Pgf* (catalog Mm00435613\_m1), *Flt1* (catalog Mm01210866\_m1). SYBR Green primers: *18S* (F: 5'-ACCTTCGATGGTAGTCGCCG-3', R: 5'-CCTTGGATGTGGTAGCCGTTT-3'), *Avp* (F: 5'-GCCTACTGGCCTTCTCCTC-3', R: 5'-GGCAGGTAGTTCTCCTCCT-3'), *Avpr1a* (F: 5'-GTGCAGAGCAAGCGGGTGTG-3', R: 5'-CGAGTCCTTCCACATACCCGT-3'), *Avpr1b* (F: 5'-CCAAGATCCGAACAGTGAAGAT-3', R: 5'-GCTGTTGAAGCCCATGTAGA-3'), *Avpr2* (F: 5'-GGCCAAGACTGTGAGGATGA-3', R: 5'-ACACGCTGCTGCTGAAAGAT-3'), *Oxtr* (F: 5'-CTCTACTCCAAGCCAAGAT-3', R: 5'-GTCTGCCCTCAGGTAGC-3'), *Cul5* (F: 5'-GTTTCAGCTCGCTGTATTGTTT-3', R: 5'-TGTGCTCTATTAGCCACTCTATTT-3'), *Hk2* (F: 5'-CTGGGTTTCACCTTCTC-GTT-3', R: 5'-GATCAGATCCACCACATCTCTG-3'), *Ddit4* (F: 5'-TCCGCAATCTTCGCTGAC-3', R: 5'-AGGAAGAGGAGGACGAGAAA-3'), *Hif1b* (F: 5'-TAGGTCCGACAGCCAATTTATC-3', R: 5'-TGGACAGAAACCTGGGAATG-3'). Gene expression was analyzed using the Livak method (97).

**Protein levels.** Total protein lysate of placentas was generated by homogenization of tissues in buffer containing 5 M NaCl, 1 M Tris, 0.5 M EDTA, NP-40, protease inhibitor (Roche), and phosphatase inhibitor (Roche). A commercially available bicinchoninic acid (BCA) protein assay kit (Thermo Fisher Scientific, 23225) was utilized per manufacturer's protocol to assess total protein concentration. Commercially available ELISAs were used to detect PGF (Thermo Fisher Scientific, catalog EMPGF) and total FLT1 protein (R&D Systems, catalog DVR100C).

**RNA-Seq.** RNA from placentas of pregnant GD 12.5 mice were isolated using a *mirVana* RNA isolation kit per manufacturer's instructions. Transcriptome profiling using RNA-Seq was performed by the University of Iowa Genomics Division according to manufacturer's recommended protocols. Briefly, 500 ng total RNA was fragmented, converted to cDNA, and ligated to sequencing adaptors containing indexes using the Illumina TruSeq stranded mRNA sample preparation kit (catalog RS-122-2101). The molar concentrations of the indexed libraries were measured using the 2100 Agilent Bioanalyzer and combined equally into pools for sequencing. The concentration of the pools were measured using the Illumina Library Quantification Kit (KAPA Biosystems) and sequenced on the Illumina HiSeq 4000 genome sequencer using a 75-bp paired-end SBS chemistry.

**Transcriptomic data analysis.** Following RNA-Seq, reads were pseudoaligned to the mouse transcriptome (GRCm38/mm10, University of California, Santa Cruz [UCSC] Genome Browser) and transcripts were quantified using Kallisto v0.43.1 (98). Transcripts were then mapped to their corresponding genes by querying the UCSC database. The gene-level RNA-Seq counts were used to determine DEGs using the open source R/BioConductor software (<http://www.bioconductor.org/>) and the DESeq2 package (99). An FDR threshold of 10% was used to determine if a gene was differentially expressed between the placentas from AVP- and saline-infused pregnancies (AVP vs. saline). The transcript per million (TPM) count for each DEG was transformed into a logarithmic value using the formula  $\log_{10}(\text{TPM} + 1)$ , and these values were used as input to generate a heatmap using Morpheus (Broad Institute, <https://software.broadinstitute.org/morpheus/>). The DEGs were also used as input for further pathway analyses with IPA. The RNA-Seq data have been deposited in the NCBI Gene Expression Omnibus database (GEO GSE116751).

**GSEA.** Expression and phenotype files were prepared as described previously (100). Analysis was carried out using GSEA software from the Broad Institute (v3.0). The GRD hypoxia-inducible (GRD\_Hypoxia) gene set consisted of 146 genes curated from the following gene sets obtained from Molecular Signature Database V6.0 (<http://software.broadinstitute.org/gsea/msigdb/>): (a) genes downregulated in MCF7 cells incubated with siHIF1A and siHIF2A (systemic name: M6189, 104 genes), (b) genes upregulated in MCF7 cells treated with the hypoxia mimetic DMOG (systemic name: M9197, 130 genes), and (c) genes upregulated in MCF7 cells cultured under hypoxic condition (1% O<sub>2</sub>) (systemic name: M7363, 171 genes). A stringent criterion was applied to select only genes that appear at least twice in the aforementioned 3 gene sets and those genes were used to compose the GRD hypoxia-inducible gene set; genes are shown in Supplemental Table 4. The Harris hypoxia gene set (systemic name: M10508, 81 genes) and oxidative stress gene set (systemic name: M15990, 96 genes) were downloaded from MSigDB 6.0.

**Statistics.** All quantitative data were analyzed using Student's *t* test or ANOVA followed by Tukey's or Dunnett's tests for multiple comparisons as indicated. All tests were 2-tailed unless otherwise noted. Data are shown as the mean  $\pm$  SEM. *P* values less than 0.05 were considered statistically significant.

**Study approval.** University of Iowa IACUC approval was obtained before any studies were performed. All studies were carried out in accordance with the 8th Guide for the Care and Use of Laboratory Animals (101).

## Author contributions

JAS, DAS, CDS, KNGC, GLP, MKS, and JLG conceived of the project. JAS, GD, DWL, SMS, KJP, ARN, TEN, SYZ, LNA, JW, HLK, MCN, NAP, KAZ, RMW, NCB, YMU, DAS, MJP, KNGC, MKS, and JLG collected and analyzed data. JAS and JLG drafted the manuscript and figures, and all authors reviewed and approved the final manuscript.

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