AIP1 functions as an endogenous inhibitor of VEGFR2-mediated signaling and inflammatory angiogenesis in mice

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ASK1-interacting protein-1 (AIP1), a recently identified member of the Ras GTPase-activating protein family, is highly expressed in vascular ECs and regulates EC apoptosis in vitro. However, its function in vivo has not been established. To study this, we generated AIP1-deficient mice (KO mice). Although these mice showed no obvious defects in vascular development, they exhibited dramatically enhanced angiogenesis in 2 models of inflammatory angiogenesis. In one of these models, the enhanced angiogenesis observed in the KO mice was associated with increased VEGF-VEGFR2 signaling. Consistent with this, VEGF-induced ear, cornea, and retina neovascularization were greatly augmented in KO mice and the enhanced retinal angiogenesis was markedly diminished by overexpression of AIP1. In vitro, VEGF-induced EC migration was inhibited by AIP1 overexpression, whereas it was augmented by both AIP1 knockout and knockdown, with the enhanced EC migration caused by AIP1 knockdown being associated with increased VEGF2 signaling. We present mechanistic data that suggest AIP1 is recruited to the VEGFR2-PI3K complex, binding to both VEGFR2 and PI3K p85, at a late phase of the VEGF response, and that this leads to inhibition of VEGF2 signaling. Taken together, our data demonstrate that AIP1 functions as an endogenous inhibitor in VEGF2-mediated adaptive angiogenesis in mice.

Introduction

Angiogenesis, the process of new blood vessel formation, is involved in many physiological and pathological settings such as ischemia, diabetes, atherosclerosis, and cancer (1). The VEGFs and their receptors have been shown to be critical in regulating vessel formation in both normal physiological and pathological processes (2). VEGF primarily utilizes its receptor VEGFR2 (also known as Flk-1 or KDR) to induce angiogenic responses by activating a variety of signaling cascades, including activation of PI3K-Akt, PLC-γ–PKC, and MAPK (3). Given the critical role of VEGFR2 signaling, regulation of VEGFR2 activity/activation may represent an important mechanism for the control of angiogenesis. Many secreted antiangiogenic factors, including vasoactive factors, have been shown to modulate VEGF bioactivity or the binding to VEGFR2 (4). In contrast, proangiogenic factors can positively regulate VEGF-VEGFR2 signaling. Sphingosine-1-phosphate via its receptor S1P1, placental growth factor via its receptor VEGFR1, and laminar flow via Src can transactivate VEGFR2 (5–8). We have also shown that proinflammatory cytokine TNF via its receptor TNFR2 transactivates VEGFR2 (9, 10). VEGFR2 activity is also regulated by direct interactions with other proteins, including co-receptor neuropilins (11), adhesion molecule vascular endothelial–cadherin (VE-cadherin) (12), and integrins (13). For example, VE-cadherin is in complex with VEGFR2 and is critical for VEGF-induced survival (PI3K-Akt) signaling (12).

VE-cadherin–VEGFR2 also regulates EC permeability (14, 15). Interestingly, a complex of VEGF2, VE-cadherin, and PECAM-1 (CD31) has been shown to be a mechanosensor that functions upstream of integrin signaling and transduces shear stress-mediated angiogenesis and vascular remodeling (16). However, negative regulations of VEGFR2 by protein–protein interactions are less understood, and an endogenous inhibitor that directly binds to and modulates VEGFR2 activity has not been identified.

Apoptosis signal-regulating kinase 1–interacting (ASK1-interacting) protein-1 (AIP1), also known as DAB2-interacting protein (DAB2IP), a recently identified, novel member of the Ras GTPase-activating protein family, has been implicated in cell growth inhibition and cell apoptosis (17, 18). AIP1, via its Ras GTPase-activating protein (Ras-GAP) activity, inhibits Ras-mediated cell survival signaling, causing cell growth inhibition (19). On the other hand, AIP1 functions as a positive regulator in cell apoptosis by mediating activation of the apoptotic kinase apoptosis signal-regulating kinase 1 (ASK1) (18). Consistent with its role as an inhibitor of cell survival and growth, AIP1 expression is often downregulated in various human cancers (17, 20, 21). Promoter analyses indicate that an epigenetic mechanism regulates AIP1 transcription (17). Recently, it has been reported that genetic variation in AIP1 gene may predict the risk of aggressive prostate cancer based on single nucleotide polymorphism (22, 23). Moreover, human AIP1 (alias for AFQ34) has been identified as a novel fusion partner from an acute myeloid leukemia patient with a t(9;11)(q34;q23) chromosomal translocation; the intron 9 of the MLL gene is translocated into the exon 2 of AIP1 and causes the disruption of the pleckstrin homology (PH) domain in the AIP1 protein and possible disruption of its functions in cell growth inhibition and apoptosis (24). These data suggest that AIP1 may function as a tumor suppressor gene.
However, the in vivo function of AIP1 has not been established. In order to define the in vivo function of AIP1, we created mice that were genetically deficient of the AIP1 gene. AIP1 is highly expressed in vascular ECs. However, AIP1-deficient mice (KO mice) show normal vascular development. We determined the function of AIP1 using several models of inflammatory angiogenesis. Our studies suggest that AIP1 functions as an endogenous inhibitor of adaptive angiogenesis, in part, by binding to the VEGFR2-PI3K complex and inhibiting the VEGFR2-dependent angiogenic signaling.

Results

Generation of KO mice. To explore the role of AIP1 in vivo, we created AIP1-knockout mice (KO mice) using homologous recombination (see Figure 1, A–D, and Methods for details). KO mice were backcrossed with C57BL/6 mice for 6 generations and they were viable and fertile. All experiments were performed with littermates AIP1+/+ (WT) and AIP1−/− (KO) mice. AIP1 protein expression was detected by Western blot with anti-AIP1 antibodies recognizing either the N-terminal PH domain or the C-terminal end of AIP1. AIP1 protein in various tissues was further determined by immunohistochemistry. Consistent with previous findings that AIP1 is highly expressed in vascular ECs (18), AIP1 protein was detected in vascular endothelium of muscle and aorta. Given that AIP1 is highly expressed in vascular endothelium, we decided to determine the function of AIP1 in adaptive angiogenesis. This end, WT and KO mice were subjected to commonly used angiogenesis models, including hind limb ischemia and foreign body response to implanted sponge. WT and KO male mice were subjected to femoral artery ligation and various analyses at different time points as described recently (10). Blood flow was

![Figure 1](http://www.jci.org)

**Figure 1**

Generation of AIP1-flox and knockout mice. (A) Schematic diagram for strategy to generate AIP1-flox mice. The targeting vector contains exons 3/4 at 5′ arm, exons 5/6 in the targeting region, and exon 7 at 3′ arm. PCR primers P1–P6 are shown. The 5′ probe with HindIII digest, and 3′ probe with Scal digest are indicated. (B and C) Screening of ES clones. Genomic DNA from positive clones was extracted and used for Southern blot, using 5′ probe after HindIII digest (B) and 3′ probe after Scal digest (C). Clone 51 was positive and used for blastocyst injection to generate chimera mice. (D) Generation of AIP1+/lox and AIP1−/− mice. The chimeras were mated with C57BL/6 for germline transmission to obtain AIP1+/lox. AIP1−/− mice were further mated with β-actin Cre mice to delete both the targeting region (exons 5/6) and the Neo gene (AIP1−/− mice). KO mice were obtained by mating between the heterozygous (AIP1+/−) male and female. (E and F) AIP1 deletion in tissues. Muscle and aorta from WT and KO mice were homogenized and AIP1 expression was detected by Western blot with anti-AIP1 antibody (E). AIP1 expression in paraffin section of muscle and aorta was determined by immunohistochemistry with anti-AIP1 antibody (F). For muscle tissue, arrows indicate capillaries and the arrowhead indicates a large vessel. For aorta tissue, the arrow indicates endothelium and the arrowhead indicates smooth muscle layer.
measured, and ischemic and nonischemic limb perfusion were measured before surgery and after surgery as indicated (3 days, 2 weeks, and 4 weeks after surgery). Data (mean ± SEM) are presented as the ratio of perfusion unit from nonischemic leg to ischemic leg. *P < 0.05. (B and C) Angiographic evidence for enhanced arteriogenesis in KO mice. Arterial phase angiograms from WT and KO mice after 4 weeks of ischemia (B) were determined by quantitative angiography (C). Data are mean values from 2 angiograms. *P < 0.05. (D–F) Enhanced angiogenesis in KO mice. Capillary density and pericyte recruitment were immunostained with CD31 (an EC marker). Representative sections from nonischemic and ischemic groups of WT and KO mice on day 28 after ischemia are shown in D. Quantification of capillary density (number/mm² muscle area) and ratio of CD31/myocyte are shown in E and F. Data are mean ± SEM from 10 fields per section (3 sections/mouse; n = 4 for each strain). *P < 0.05.

Figure 2
KO mice showed enhanced arteriogenesis and angiogenesis in vivo. (A) KO mice showed augmented recovery of limb perfusion compared with WT mice. Blood flow of ischemic and nonischemic limbs were measured in gastrocnemius muscle at before surgery and 30 minutes, 3 days, 2 weeks, and 4 weeks after surgery. Data (mean ± SEM) are presented as the ratio of perfusion unit from nonischemic leg to ischemic leg. *P < 0.05. (B and C) Angiographic evidence for enhanced arteriogenesis in KO mice. Arterial phase angiograms from WT and KO mice after 4 weeks of ischemia were determined by quantitative angiography (C). Data are mean values from 2 angiograms. *P < 0.05. (D–F) Enhanced angiogenesis in KO mice. Capillary density and pericyte recruitment were immunostained with CD31 (an EC marker). Representative sections from nonischemic and ischemic groups of WT and KO mice on day 28 after ischemia are shown in D. Quantification of capillary density (number/mm² muscle area) and ratio of CD31/myocyte are shown in E and F. Data are mean ± SEM from 10 fields per section (3 sections/mouse; n = 4 for each strain). *P < 0.05.

The role of AIP1 in angiogenesis was further determined in a sponge granuloma model (26, 27). In the early phase (1–3 weeks), the invasion of the sponge by cells and the formation of new tissue permit the quantification of neovascularization. Polyvinyl alcohol sponges were implanted subcutaneously into WT and KO mice and harvested on days 14 or 21. The sponges were subjected to trichrome staining. KO mice had increased invasion by cells and sponge encapsulation (Figure 3A, indicated by arrows). Moreover, the sponges from KO mice had dramatic increases in number of red blood cells (Figure 3A with quantification in Figure 3B), an indicator of new blood vessel formation. We further determined macrophage infiltration and blood vessel formation by immunostaining. KO mice had enhanced macrophage infiltration (measured by anti-F4/80 staining) and neovascularization (measured by anti-CD31 staining) (Figure 3C with quantification in Figure 3, D and E). Interestingly, red blood cells in sponges from WT mice were localized inside vessels (arrowheads in Figure 3A). In contrast, red blood cells in sponges from KO mice were outside the vasculature (Figure 3A), suggesting enhanced vascular leakage. This observation prompted us to examine activation of VEGF-VEGFR2–mediated vascular leakage (15). Consistent with mice that underwent enhanced macrophage infiltration, KO mice had increased VEGF production as determined by Western blot with anti-VEGF (Figure 3F). Similarly, total and phosphorylated VEGFR2 in KO mice.
were also dramatically increased as detected by Western blot with a phospho-specific antibody (Figure 3F). These results suggest that KO mice had augmented VEGF-VEGFR2 signaling.

**VEGF-induced ear, retina, and cornea neovascularization were greatly augmented in KO mice.** The hind limb ischemia and the sponge granuloma models represent pathological settings, involving inflammation, multiple cytokines, and proangiogenic factors (28). To directly determine the role of AIP1 in regulating VEGF-VEGFR2-dependent angiogenesis in vivo, we performed VEGF-induced retina, ear, and cornea angiogenesis in KO mice. For the retina angiogenesis, adenovirus encoding VEGF164 or β-gal was injected intravenously. Five days after viral injection, the vasculature was visualized by isoelectin staining as described previously (29). VEGF-induced retina angiogenesis was significantly augmented in KO mice (Figure 4A with quantification in Figure 4B). For the ear angiogenesis model, adenovirus encoding VEGF164 or β-gal was injected intradermally into the ears. VEGF-induced angiogenesis was accessed by direct microscopy (Supplemental Figure 4A). Ear permeability was measured by Evans blue dye (EBD) assay (Supplemental Figure 4B). Ear vasculature was also visualized by a whole-mount staining with PE-conjugated anti-CD31 (Supplemental Figure 4C with quantification in Supplemental Figure 4D). We also examined angiogenesis in the avascular cornea, into which recombinant VEGF protein formulated into Hydron pellets was implanted as described previously (30) (Figure 4, C and D). In these assays, VEGF-induced angiogenesis was dramatically augmented in KO mice. To determine if the loss of AIP1 is responsible for enhanced VEGF responses, AIP1 was reintroduced into retina of KO mice by an adenoviral expression system. AIP1 overexpression in KO mice diminished augmented VEGF responses (Supplemental Figure 5). These data suggest that AIP1 is critical for “normal” VEGF response and may function as a specific endogenous inhibitor in VEGF-induced angiogenesis.

**VEGF-induced EC migration and tube formation were inhibited by AIP1 overexpression but augmented by AIP1 deficiency.** Next we examined VEGF-dependent angiogenesis using in vitro models for EC migration and tube formation under normal and AIP1-deficient conditions.
migration and tube formation. Primary human ECs (HUVECs) were infected with an inducible adenoviral vector expressing AIP1 (Figure 5A), and the effects of AIP1 overexpression on EC migration were determined in a transwell migration assay (Figure 5B) and a monolayer “wound injury” assay (Figure 5, C and D). In both assays, VEGF-induced EC migration was markedly inhibited by AIP1 overexpression. The effects of AIP1 overexpression on EC tube formation were also determined in a matrigel tube assay. Similar to results from EC migration, AIP1 overexpression significantly inhibits EC tube formation (Figure 5, E and F). Conversely, the effects of AIP1 deficiency on EC angiogenesis were also determined in both AIP1 knockdown HUVECs by siRNA and mouse lung microvessel ECs isolated from KO mice. AIP1 deficiency strongly augmented EC migration and tube formation in identical assays (Figure 5, G–J, for mouse lung microvessel ECs; Supplemental Figure 6A for HUVEC). Similar results were obtained in HUVECs by AIP1 siRNA knockdown (Supplemental Figure 6B). However, AIP1 deletion appeared to have no effect on bFGF-induced signaling (phosphorylation of PLC-γ and Akt) (Supplemental Figure 7), suggesting that AIP1 may have a specific inhibitory effect on VEGF-induced angiogenic response. In contrast, AIP1 deficiency in KO cells showed a reduced response in TNF-induced JNK activation as we recently shown in the same cell line (31).

We then determined whether AIP1 inhibits VEGFR2 signaling by directly associating with VEGFR2 receptor complexes. To this end, HUVECs were treated with VEGF for various times (Figure 6B), and endogenous the AIP1-VEGFR2 complex was determined by a coimmunoprecipitation assay with anti-VEGFR2, followed by Western blot with anti-AIP1. Phosphorylation of VEGFR2 and formation of VEGFR2–PLC-γ and VEGFR2-PI3K complexes peaked at 5 minutes in response to VEGF. However, the AIP1-VEGFR2 complex was only weakly detected at 5 minutes, but dramatically increased after 15–30 minutes (Figure 6B). These data suggest that AIP1 is recruited to VEGFR2 at a late phase of VEGF response when activation of VEGFR2-PI3K/PLC-γ declines. We reasoned that AIP1 associates with an active phosphorylated
form of VEGFR2. To test our hypothesis, ECs were treated with VEGF in the absence or presence of VEGFR2-specific inhibitor SU1498. Indeed, SU1498 specifically blocked phosphorylation of VEGFR2 as well as the formation of the AIP1-VEGFR2 complex (Figure 6C). We also examined the effects of PI3K-specific inhibitor LY294002, PLC-γ inhibitor U73122, and its inactive homolog U73343 on VEGFR2-AIP1 association. As expected, LY294002 and U73122 specifically blocked VEGF-induced phospho-Akt and phospho–PLC-γ, respectively (Figure 6C). Interestingly, PI3K inhibitor (PLC-γ inhibitor) strongly enhanced VEGF-induced association of AIP1 with VEGFR2 as well as the VEGFR2-PI3K p85 complex (Figure 6C). Similar results were obtained with another PI3K-specific inhibitor, wortmannin (Figure 6D). It is conceivable that PI3K inhibition enhances the formation of VEGFR2-PI3K-AIP1 complex. These data support that AIP1 is recruited to the VEGFR2-PI3K complex at a late phase of VEGF response, leading to an inhibition of VEGFR2-dependent signaling.

AIP1 via its proline-rich (PR) motif binds to the Src homology region 3 (SH3) domain of PI3K, while through the C2 domain, AIP1 binds to a tyrosine-phosphorylated form of VEGFR2. We further investigated the VEGFR2-PI3K-AIP1 complex by mapping the critical domains of AIP1 for the interactions. AIP1 contains the N-terminal (AIP1-N) and C-terminal (AIP1-C) half and a PR motif at the C-terminal (AIP1-C-PR) half, which we predicted to bind to

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**Figure 5**

VEGF-induced EC migration and tube formation were inhibited by AIP1 overexpression but augmented by AIP1 deletion. (A–D) AIP1 overexpression inhibits EC migration. HUVECs were infected with adenovirus expressing WT (Ad-AIP1) in the presence of 4 μM doxycycline for 24 hours. (A) AIP1 expression was determined by Western blot with anti-AIP1. (B) Cells were cultured in serum-free media for overnight and subjected to a transwell migration assay and (C) with quantification in D) monolayer “wound injury” assay in response to VEGF (10 ng/ml). (E and F) AIP1 overexpression inhibits EC tube formation. HUVECs were infected with Ad-AIP1 and subjected to a Matrigel tube formation assay VEGF (10 ng/ml) (E with quantification in F). (G–J) AIP1 deficiency augmented on EC migration and tube formation. WT and KO mouse lung microvessel ECs (MLECs) were cultured in 0.5% FBS overnight and subjected to EC migration and tube formation as described above. (G) AIP1 expression was determined by Western blot with anti-AIP1. Quantification for EC migration in transwell assay (H), “wound injury” assay (I), and tube formation assay (J) are shown. Data presented are mean ± SEM of the triplicates from 3 independent experiments. *P < 0.05.
the SH3 domain of the regulatory p85 subunit of PI3K (Figure 7A). We tested this possibility using an in vitro pulldown assay with glutathione-S-transferase–p85-SH3 (GST-p85-SH3) fusion protein as bait. GST-fusion proteins containing the SH3 domains from Src and α-spectrin protein were used as controls. AIP1-C-PR containing the PR motif (Figure 7A) from 293T expression lysate strongly bound to PI3K p85 and weakly bound to Src but not to spectrin. In contrast, AIP1-C with a deletion of the PR region (AIP1-C-ΔPR) was not pulled down by GST-SH3 of p85 (Figure 7B), confirming the PR motif on AIP1 for PI3K binding. To determine the critical domains of AIP1 for VEGFR2 binding, an expression vector encoding for a full-length AIP1 (AIP1-F), AIP1-N, or AIP1-C (Figure 7A) was coexpressed with VEGFR2, and AIP1-VEGFR2 association was determined by a communoprecipitation assay. AIP1-N, but not AIP1-F or AIP1-C, bound to VEGFR2 (Figure 7C). AIP1-F did not associate with VEGFR2, consistent with our previous observations that AIP1 normally exists in a closed inactive form by an intramolecular interaction between the N- and the C-terminal domains (18). AIP1-N contains several structural domains: PH for the targeting on membrane, C2 for the interactions with proteins (e.g., ASK1), and GAP for the inhibition on Ras (Figure 7A). We then determined whether these domains of AIP1 are critical for associating with an active form of VEGFR2 in the late phase of VEGF response. (A) WT and KO mouse lung microvessel ECs were cultured overnight in 0.5% FBS, followed by VEGF treatment (10 ng/ml) for 0–30 minutes as indicated. Phosphorylation of VEGFR2, PLC-γ, and Akt and the total proteins were determined by Western blot with respective antibodies. (B) Formation of the VEGFR2-PI3K-AIP1 complex. HUVECs were treated with VEGF for 0–30 minutes as indicated. Proteins of p-VEGFR2, total VEGFR2, PLC-γ, and AIP1 in the input were determined by Western blot with respective antibodies. Association of endogenous VEGFR2 with AIP1, PLC-γ, and PI3K p85 was determined by immunoprecipitation with anti-VEGFR2, followed by Western blot with anti-AIP1, anti-PLC-γ, and PI3K p85, respectively. (C and D) Effects of inhibitors on the VEGFR2-PI3K-AIP1 complex formation. HUVECs were serum starved for 16 hours and pretreated with the following inhibitors: U73122 (U22), U73343 (U43), SU1498 (SU), or LY294002 (LY) (each at 10 μM for 30 minutes in C; LY294002 and wortmannin (wort) at 1 μM for 30 minutes in D). –, mock. Cells were stimulated with VEGF (10 ng/ml) for 15 minutes, and proteins of p-VEGFR2, total VEGFR2, PLC-γ, and AIP1 in the input were determined by Western blot with respective antibodies. Association of endogenous VEGFR2 with AIP1 and PI3K p85 was determined as above.

Figure 6

AIP1 inhibits VEGFR2-dependent signaling by associating with an active form of VEGFR2 in the late phase of VEGF response. (A) WT and KO mouse lung microvessel ECs were cultured overnight in 0.5% FBS, followed by VEGF treatment (10 ng/ml) for 0–30 minutes as indicated. Phosphorylation of VEGFR2, PLC-γ, and Akt and the total proteins were determined by Western blot with respective antibodies. (B) Formation of the VEGFR2-PI3K-AIP1 complex. HUVECs were treated with VEGF for 0–30 minutes as indicated. Proteins of p-VEGFR2, total VEGFR2, PLC-γ, and AIP1 in the input were determined by Western blot with respective antibodies. Association of endogenous VEGFR2 with AIP1, PLC-γ, and PI3K p85 was determined by immunoprecipitation with anti-VEGFR2, followed by Western blot with anti-AIP1, anti-PLC-γ, and PI3K p85, respectively. (C and D) Effects of inhibitors on the VEGFR2-PI3K-AIP1 complex formation. HUVECs were serum starved for 16 hours and pretreated with the following inhibitors: U73122 (U22), U73343 (U43), SU1498 (SU), or LY294002 (LY) (each at 10 μM for 30 minutes in C; LY294002 and wortmannin (wort) at 1 μM for 30 minutes in D). –, mock. Cells were stimulated with VEGF (10 ng/ml) for 15 minutes, and proteins of p-VEGFR2, total VEGFR2, PLC-γ, and AIP1 in the input were determined by Western blot with respective antibodies. Association of endogenous VEGFR2 with AIP1 and PI3K p85 was determined as above.
The positive charge residues with the C2 domain are critical for protein-protein interactions. We analyzed the effects of mutations of the 2 lysine clusters (Figure 7A), between which the second lysine cluster has been shown to be important for ASK1 binding (18). Results showed that a mutation at the first lysine cluster (KA1), but not that at the second cluster (KA2), reduced the binding of AIP1 to VEGFR2. KA3 contains both mutations and did not show further reduction in the binding (Figure 7E). These data strongly support that the AIP1 via its first lysine cluster associates with VEGFR2.

Effects of AIP1 mutants on p-VEGFR2-PI3K-Akt signaling. Finally, we determined a direct inhibitory effect of various AIP1 mutants on VEGFR2-dependent signaling. To this end, VEGFR2 was coexpressed with an AIP1 mutant (AIP1-N, AIP1-PR, PHC2) in 293T cells that do not express endogenous VEGFR2. Overexpression of VEGFR2 (but not the kinase-inactive form) induced autophosphorylation as well as activation of downstream targets PI3K-Akt and PLC-γ as determined by Western blot with phospho-specific antibodies (Figure 7A). Coexpression of AIP1-N and PHC2 containing the C2 domain strongly inhibits phospho-VEGFR2 and its downstream targets phospho-Akt and phospho-PLC-γ. AIP1-PHC2 contains a deletion of the GAP1 domain, suggesting that the GAP activity is not required for the inhibition of AIP1 on VEGFR2. We further showed that AIP1-N-R289L, with a mutation at the catalytic site, similar to AIP1-N, could bind to VEGFR2 and inhibit phosphorylation of VEGFR2 (Figure 7B). In contrast, AIP1-C-PR specifically blocked VEGFR2-induced phospho-Akt but not phospho-VEGFR2 or phospho-PLC-γ.
These results strongly support that AIP1 via different domains targets both VEGFR2 and PI3K p85, leading to inhibition of VEGFR2-dependent angiogenic signaling.

**Discussion**

In the present study, we show that KO mice exhibit dramatically enhanced ischemia and inflammatory angiogenesis in several in vivo models (ischemic hind limb and sponge granuloma models). Furthermore, VEGF-induced neovascularization is greatly augmented in KO mice, which can be suppressed by re-expression of AIP1. In cultured ECs, overexpression of AIP1 inhibits, while knockout or knockdown of AIP1 in ECs enhances, VEGF-induced migration and tube formation. Mechanistic studies suggest that AIP1 is recruited to the VEGFR2-PI3K complex at a late phase of VEGF response. AIP1 via its C2 domain associates with VEGFR2 while via its PR domain binds to the SH3 domain of PI3K p85, leading to an inhibition of VEGFR2-mediated angiogenic signaling (Figure 8C). Our in vivo and in vitro studies demonstrate that AIP1 functions as an endogenous scaffold in inflammatory angiogenesis by suppressing VEGFR2-Akt–dependent signaling.

The ischemic hind limb model was created to study peripheral arterial disease and has been extensively used to assess a role of a gene in adult arteriogenesis and angiogenesis. The model represents pathological settings, involving inflammation, cytokine/proangiogenic factors, and altered systemic/local blood flow (28). We first assessed the phenotype of KO mice in this model, in which KO mice showed impaired arteriogenesis and angiogenesis. We then determined the role of AIP1 in more "simplified" in vivo angiogenesis models, in which only limited components were involved. For example, inflammation (but not ischemia) is predominant in the sponge granuloma model (26). In contrast, less inflammation is involved in the ear and retina angiogenesis assay, in which a single proangiogenic factor could be applied. In these assays, we have shown that AIP1 specifically inhibits
adenoviral-expressed VEGF-induced angiogenesis in the ear and retina angiogenesis assays. Moreover, recombinant VEGF-induced cornea angiogenesis is also augmented in KO mice.

Our study here demonstrates that AIP1 is dispensable in vascular development but suppresses VEGF-induced angiogenesis in adult tissues using these animal models. The mechanisms by which KO mice show normal development, birth, growth, and breeding compared with WT mice are not known. The close members of Ras-GAP family include p120RasGAP and nGAP (Ras protein activator-like 1). It has been reported that deletion of p120RasGAP in mice results in early embryonic lethality associated with defective vasculogenesis and increased apoptosis of neuronal cells, indicating the importance of this gene during vascular embryogenesis (32).

Effect on VEGFR2 signaling. Nevertheless, our data support that AIP1 may directly bind to and inhibit VEGFR2 activity. Association of AIP1 with VEGFR2 is diminished by a PKC inhibitor, indicating that VEGFR2 signaling is required for the recruitment of AIP1. In contrast, AIP1-VEGFR2 as well as AIP1-P13K and VEGFR2-P13K complexes are strongly enhanced by a PI3K inhibitor. These results may support a model in which AIP1 retains VEGFR2 in an inactive state.

Methods

Targeted inactivation of AIP1 gene by homologous recombination

We have recently reported the KO strategy (31). Briefly, the AIP1 targeting arms were isolated from a BAC clone identified by screening a BAC library from Research Genetics (Invitrogen), using AIP1 cDNA as a probe. The targeting vector was constructed in pEASYflox backbone to contain a loxp site inserted upstream of AIP1 exon 5 and a neomycin cassette (Neo), flanked by 2 loxp sites downstream of exon 6, using standard molecular procedures. The linearized targeting construct was electroporated into 129/C57B/6 ES cells, and the targeted clones were
selected with G418 and ganciclovir. Resistant clones were screened for homologous recombination by PCR and confirmed by Southern blot analysis. Two independent AIP1−/− ES clones were injected into WT blastocysts. Chimeras were further bred with WT females for germline transmission. AIP1−/− mice were mated with β-actin Cre mice to generate AIP1−/− mice with a deletion of both the targeting region (the exons 5/6) and the Neo gene. KO mice were obtained by mating between the heterozygous (AIP1+/−) male and female.

**In vivo angiogenesis models**

All animal studies were approved by the institutional animal care and use committees of Yale University. Eight- to twelve-week-old male littermates of WT and KO mice were used for all experiments.

**Mouse hind limb ischemic model.** The mouse hind limb ischemia model was used as described recently (10, 25). Briefly, following anesthesia (79.5 mg/kg ketamine; 9.1 mg/kg xylazine), the left femoral artery was exposed under a dissection microscope. The proximal of femoral artery and the distal portion of saphenous artery were ligated. All branches between these 2 sites were ligated or cauterized, and arteriectomy was performed. The sham operation was without any femoral artery ligation but skin incision. Blood flow was measured by PeriFlux system with Laser Doppler Perfusion Module (LDPU) unit (Perimed Inc.). A deep measurement probe was placed directly on gastrocnemius muscle to ensure a deep muscle blood flow measurement. Ischemic and nonischemic limb perfusion was measured before and 3 days, 2 weeks, and 4 weeks after surgery. The final blood flow values were expressed as the ratio of ischemic to nonischemic hind limb perfusion. Arteriogenesis was also analyzed by quantitative angiography (45). Briefly, 2 and 4 weeks after surgery, mice were anesthetized and heparinized. Mice were perfused with PBS containing vasodilators (4 mg/l papaverine; 1 g/l adenosine) for 3 minutes at physiological pressure through the descending aorta, and blood was drained from the inferior vena cava. The vasculature was fixed with 2% paraformaldehyde (in PBS) for 5 minutes, flushed with PBS for 2 minutes, and infused with contrast agent (bismuth oxychloride in saline and 10% gelatin in PBS; 1:1). Mice were then immersed in ice to solidify the contrast agent. Microangiography was taken with Faxitron x-ray machine (Hewlett-Packard) at 25 kV and 3.25 mA for 3 minutes. Upper limb vascular density (pixel density), vessel length (average length of vessels with diameter >1 pixel), and fractal dimension were analyzed by modified ImageJ software (http://rsbweb.nih.gov/ij/) and MATLAB software (TheMathWorks).

**Sponge granuloma model.** A polyvinyl alcohol sponge (Ultracell PVA sponge; Ultracell Medical Technologies Inc.) was implanted subcutaneously into 8- to 10-week-old WT and KO mice. Implants were excised at 2 and 3 weeks following implantation for histological analysis and cell recovery. We chose 2–3 weeks because inflammatory angiogenesis peaks during this period whereas the foreign body response begins at 4 weeks. Mice were monitored during this time, but they were not subjected to any additional procedures. The sponge was harvested, fixed in 1% paraformaldehyde for 4.5 hours on ice, and embedded in paraffin. The paraffin section (5 μM) was used for immunostain with anti–CD31, anti–α-SMA, and anti-F4/80 as described previously (10, 25). Bound primary antibodies were detected using avidin-biotin-peroxidase (NovaRED Substrate Kit; Vector Laboratories). Pictures from 4 random areas of each section and 5 sections per mouse were taken using a Kodak digital camera mounted on a light microscope (×40 objective). Images were quantified using the MATLAB software (The MathWorks) as described recently (10, 25). TUNEL assay was performed according to the instruction provided by the manufacturer’s protocol (Roche) (25).

**Plasmids and adenovirus**

Mammalian expression plasmids for VEGFR2 (Flk-1/KDR) and mutants were described previously (9). Adenoviruses expressing LacZ and VEGF were generated in our laboratory. To construct AIP1 in an inducible angiogenesis system, 2 vectors, pShuttleTetTripLac-Bam and pAdEasy-Tet-ON, were obtained from G. Akusjrav (Uppsala University, Uppsala, Sweden). First, 2 pairs of primers, 5′-GATCCGGCGCGCTCGAGACGGATCGTCTTAGATCAGATCAACCGTGAGATGTATCTAGACG-3′ and 5′-GATCTGATATCTAGACGATCGTCGAGCGGCACGACGGAGCAGCCGC-3′, were annealed and ligated into BamHI digested pShuttleTetTripLac-Bam to generate pShuttleTetTripLac-Bam-MCS, which contains multiple cloning sites. Two pairs of primers, 5′-CTGCAGCTGCGGAGACTACAACGAGGCCG-3′ and 5′-CGTCTAGATGAAATTTGCTGCTGTTTCTGAACTC-3′, were used to amplify AIP1-F by PCR using AIP1/2AB as a template. The PCR fragments were digested by Bcl I and Xba I and ligated to the same enzyme-digested pShuttleTetTripLac-Bam-MCS. The resulting construct containing FLAG-tagged AIP1-F in the Shuttle vector were designated pShuttleTetTripLac-AIP1. pShuttleTetTripLac-AIP1 and pAdEasy-Tet-ON were cotransfected into BJS183 to generate recombinant pAd-AIP1. pAd-AIP1 plasmid was further amplified in recombinant-deficient XL10-Gold strain (Stratagene). pAd-AIP1 virus was amplified in 293T cells and purified by ultracentrifugation. Expression of AIP1 was induced by adding 4 μM doxycycline into pAd-AIP1 infected cells for 16 hours (49).
Cell culture, inhibitors, cytokines, and transfection

HUVECs were from Yale Endothelial Cell Facility (Yale University). Bovine aortic ECs were purchased from Clonetics and were cultured in DMEM medium containing 10% FBS. Mouse EC isolation from muscle and lung tissues was performed as described previously (25). Isolation of primary mouse brain endothelial cells was performed as follows. Meninges of 3-week-old mouse was carefully removed from the forebrain and gray matter was minced and then digested with 1 mg/ml collagenase CLS2 (Worthington) in DMEM (Sigma-Aldrich) containing 50 mg/ml gentamicin and 2 mM glutamine in a shaker at 2 hours at 37°C. The cell pellet was separated by centrifugation in 20% BSA–DMEM (1,000 g, 20 minutes). The microvessels obtained in the pellet were further digested with 1 mg/ml collagenase-dispase (Roche) in DMEM for 1.5 hours at 37°C. Microvessel endothelial cell clusters were separated on a 30% continuous Percoll gradient and washed twice in DMEM before planting on collagen type IV and human fibronectin-coated 35-mm plastic dishes. Cultures were maintained in DMEM supplemented with 20% FBS and 1% non-essential amino acids. mouse aortic ECs were purchased from Clonetics and were cultured in DMEM medium containing 10% FBS. Mouse EC isolation from muscle and lung tissues was performed as described previously (25). Briefly, the under surface of the filters were coated overnight at 4°C with fibronectin (20 μg/ml) in PBS (pH 7.4). The coating solution was removed from the lower chamber before filling with migration medium. The lower chambers were filled with DMEM containing 0.5% BSA, with or without VEGF (50 ng/ml). Cells were harvested in PBS containing 5 mM EDTA, and after washing with DMEM containing 5 mM MgCl2, cells were resuspended in 100 μl of protein A/G PLUS-agarose (Santa Cruz Biotechnology) and incubated with the first protein-specific antiserum (e.g., anti-VEGFR2) on ice for 1.5 hours. Then, 10 μl of protein A/G PLUS-agarose (Santa Cruz Biotechnology Inc.) was added and incubated for 2 hours with rotation. Immune complexes were collected after each immunoprecipitation by centrifugation at 13,000 g for 10 minutes followed by 3–5 washes with lysis buffer. The immune complexes were subjected to SDS-PAGE, followed by immunoblot with the second protein (e.g., phosphotyrosine antibody; Upstate). The chemiluminescence was detected using an ECL kit according to the instructions of the manufacturer (Amer sham Life Science). For detection of FLAG-tagged proteins (AIP1), anti-FLAG M2 antibody was used for immunoblot. Antibodies against phospho-VEGFR2 (pY1175), phospho-Akt (Ser-473), Akt, phospho–PLC-γ (pY783) and PLC-γ, and phospho-JNK and JNK were from Cell Signaling Technology Inc. Anti–FLAG M2 antibody was from Sigma-Aldrich. Protein A/G PLUS-agarose and anti–β-tubulin were from Santa Cruz Biotechnology Inc.

In vitro EC migration and tube formation assays

A transwell migration assay. Cell migration was examined using Transwell fitted with polycarbonate filters (8-μm pore size) (Corning Inc.). Briefly, the under surface of the filters were coated overnight at 4°C with fibronectin (20 μg/ml) in PBS (pH 7.4). The coating solution was removed from the lower chamber before filling with migration medium. The lower chambers were filled with DMEM containing 0.5% BSA, with or without VEGF (50 ng/ml). Cells were harvested in PBS containing 5 mM EDTA, and after washing with DMEM containing 5 mM MgCl2, cells were resuspended in 100 μl of protein A/G PLUS-agarose (Santa Cruz Biotechnology) and incubated with the first protein-specific antiserum (e.g., anti-VEGFR2) on ice for 1.5 hours. Then, 10 μl of protein A/G PLUS-agarose (Santa Cruz Biotechnology Inc.) was added and incubated for 2 hours with rotation. Immune complexes were collected after each immunoprecipitation by centrifugation at 13,000 g for 10 minutes followed by 3–5 washes with lysis buffer. The immune complexes were subjected to SDS-PAGE, followed by immunoblot with the second protein (e.g., phosphotyrosine antibody; Upstate). The chemiluminescence was detected using an ECL kit according to the instructions of the manufacturer (Amer sham Life Science). For detection of FLAG-tagged proteins (AIP1), anti-FLAG M2 antibody was used for immunoblot. Antibodies against phospho-VEGFR2, phospho-VEGFR2 (pY1054/1059), and AIP1 were described previously (9, 18).

Monolayer EC migration. Monolayer EC migration was modified from the method described previously (25). Briefly, infected ECs were cultured in 0.5% FBS overnight and subjected to “wound injury” assay with a plastic pipette tip (25). Cells were plated with fresh media and were further cultured for 12–24 hours. The EC migration in culture was determined by measuring wound areas in cell monolayers. Three different images from each well along the wound were captured by a digital camera under a microscope (×4). Wound area (μm²) was measured and analyzed by NIH Image 1.60 (http://rsbweb.nih.gov/nih-image/).

EC tube formation. EC tube formation in Matrigel was performed as follows: 24-well tissue culture plates were coated with 0.2 ml of Matrigel per well and incubated at 37°C for 1 hour (Matrigel; BD). HUVECs were serum starved overnight and 2,000 cells were plated on Matrigel (0.2 ml) in a 24-well plate. At 30 minutes after plating, fresh medium containing the indicated concentrations (see the legend for Figure 6, C and D) of inhibitors in 1% serum was added and further incubated for 24 hours. A branched EC network was visualized under microscope and photographs were taken using a Leica phase contrast microscope. The area covered by branched cells (μm²) was measured and analyzed by NIH Image 1.60 (http://rsbweb.nih.gov/nih-image/).

Statistics

All data are expressed as mean ± SEM. Statistical differences were measured by the nonparametric Mann-Whitney U test. A value of P < 0.05 was considered as statistically significant.

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