Macrophage Sult2b1 promotes pathological neovascularization in age-related macular degeneration

Yafang Wang¹, Yang Liu¹, Yan Wang², Yidong Wu¹, Zhixuan Chen¹, Feng Wang⁷, Xiaoling Wan¹,³, Fenghua Wang¹,³,⁴,⁵,⁶, Xiaodong Sun¹,³,⁴,⁵,⁶

Disordered immune responses and cholesterol metabolism have been implicated in age-related macular degeneration (AMD), the leading cause of blindness in elderly individuals. SULT2B1, the key enzyme of sterol sulfonation, plays important roles in inflammation and cholesterol metabolism. However, the role and underlying mechanism of SULT2B1 in AMD have not been investigated thus far. Here, we report that SULT2B1 is specifically expressed in macrophages in choroidal neovascularization lesions. Sult2b1 deficiency significantly reduced leakage areas and inhibited pathological angiogenesis by inhibiting M2 macrophage activation in vivo and in vitro. Mechanistically, loss of Sult2b1 activated LXRs and subsequently increased ABCA1 and ABCG1. SULF2B1, STS, an enzyme of sterol desulfonation, protected against pathological angiogenesis by activating LXR–ABCA1/G1 signalling to block M2 polarization. Collectively, these data reveal a cholesterol metabolism axis related to macrophage polarization in neovascular AMD.

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Introduction

Age-related macular degeneration (AMD), the leading cause of blindness among elderly people (1), occurs in two main end-stage forms: non-neovascular (dry) AMD and neovascular (wet) AMD (nAMD). Wet AMD, which accounts for ~90% of cases of severe vision loss (2, 3, 4), is characterized by the abnormal extension of blood vessels into the retina, termed choroidal neovascularization (CNV). Given the key role of VEGF in the pathology of nAMD, anti-VEGF therapy is currently the gold standard of disease management (3), but treatment responses vary, and long-term anti-VEGF treatment may cause severe retinal adverse events (5, 6, 7, 8). Thus, there is an urgent need to explore alternative approaches to treat nAMD.

The accumulation of lipid-rich deposits, namely, drusen, between the retinal pigment epithelium (RPE) and Bruch’s membrane is a pathological hallmark in the early and intermediate stages of AMD (2, 9). Drusen participates in nAMD progression, resulting in immune cell recruitment and activation (10). In the infiltrating innate immune cells in nAMD lesions, we and other groups have demonstrated that macrophages are the main cell type, indicating that they play an essential role in the pathogenesis of this disease (11, 12, 13, 14). In response to various tissue-derived or environmental stimuli, macrophages can undergo polarization to the pro-inflammatory (M1) or proangiogenic (M2) phenotype (14, 15). Compared with M1 macrophages, M2 macrophages are the predominant participant in the pathological angiogenesis of nAMD (16, 17, 18). Modulation of this phenotype change may be a novel therapeutic strategy for the treatment of nAMD.

Recent studies have shown that cholesterol metabolism is a key regulator of macrophage activation (19, 20). Given the high-cholesterol content of drusen in subretinal lesions, it is crucial to better understand the role of abnormal cholesterol metabolism in macrophage polarization in nAMD. Sulfotransferase family 2b member 1 (SULT2B1) is the key enzyme in the sulfate conjugation of 3β-hydroxysteroids such as cholesterol, oxysterol, and pregnenolone (21, 22, 23), a process that ultimately affects cholesterol metabolism. The desulfation process is catalysed by steroid sulfatase (STS). Our previous study showed that Sult2b1 deficiency promoted pro-inflammatory macrophage polarization and exacerbated ischaemic stroke in the brain (24), the occurrence of which is positively associated with increased cholesterol (25). However, the potential role and underlying mechanism of Sult2b1 in nAMD remain unclear.

In this study, we show that Sult2b1 deficiency attenuates CNV development by inhibiting proangiogenic M2 macrophage polarization...
in vivo and in vitro. Moreover, loss of Sult2b1 activated liver X receptors (LXR)-ABCA1 and ABCG1 (ABCA1/G1) signalling for efficient cholesterol efflux, consequently preventing M2 macrophage polarization. In contrast to the effects of Sult2b1 deficiency, knockout of Sts inactivated LXR–ABCA1/G1 signalling and promoted both M2 macrophage polarization and pathologic neovascularization. Our findings highlight the importance of SULT2B1 and STS in regulating macrophage polarization and cholesterol metabolism in the pathologic neovascularization of nAMD, which might provide a potential therapeutic strategy for nAMD.

Results

Sult2b1 deficiency decreases the CNV lesion area in vivo

To better understand the role of Sult2b1 in nAMD in vivo, we measured Sult2b1 mRNA levels in RPE–choroid tissue in a laser-induced mouse model of nAMD. During CNV lesion development, Sult2b1 mRNA levels were significantly higher (CNV3d, P < 0.001; CNV5d, P < 0.01; CNV7d, P < 0.05) in the laser-induced CNV group than in the control group (Fig S1), indicating that Sult2b1 may play an important role in CNV development.

Then, we performed laser photocoagulation of the ocular fundus in WT- and Sult2b1-deficient (Sult2b1+/−) mice to assess whether Sult2b1 deficiency alters CNV, as represented by leakage or neovascularization areas. The fundus fluorescein angiography (FFA) results showed that 7 d after laser photocoagulation, the leakage area of the laser-induced lesions was significantly smaller (P < 0.0001) in Sult2b1+/− mice than in WT mice (Fig 1A and B). The neovascularization area (labelled with isoelectin) was also significantly smaller (P < 0.001) in Sult2b1+/− mice than in WT mice (Fig 1C and D). Consistently, the mRNA levels of proangiogenic growth markers (Vegfa, Pdgfb, and Tek), an endothelial cell marker (Tek), and profibrotic factors (Tgfb1, Pdgfb, and Fgf2) were lower in the CNV lesions of Sult2b1+/− mice than in those of WT mice (Fig 1E).

Figure 1. Sult2b1 deficiency attenuates CNV in a mouse model of nAMD.
(A, B) Representative images of fundus fluorescein angiography (A) and quantification of the leakage area (red circle, (B)) in WT and Sult2b1+/− mice (n = 10 per group). Scale bar, 300 μm. (C, D) Representative isoelectin (green) staining (C) of RPE–choroid flat mounts and quantification of the CNV area (yellow circle, (D)) in WT and Sult2b1+/− mice (n = 10 per group). Scale bar, 300 μm. (E) Relative mRNA levels of proangiogenic growth markers (Vegfa, Pdgfb, and Tek), an endothelial cell marker (Tek), and profibrotic factors (Tgfb1, Pdgfb, and Fgf2) in the RPE–choroid flat mounts of WT and Sult2b1+/− mice (n = 5 per group). Data information: in (B, D, E), data are presented as the mean ± SD. **P < 0.01, ***P < 0.001, ****P < 0.0001 (unpaired two-tailed t test).
Together, these results suggest that Sult2b1 facilitates the progression of laser-induced CNV.

SULT2B1+ macrophages are required for CNV progression in vivo

To determine which cell type is responsible for CNV development, we first examined the expression pattern of SULT2B1 in CNV lesions. Interestingly, SULT2B1 was specifically expressed in macrophages (F4/80+) in the CNV areas of WT mice but not of Sult2b1−/− mice (Fig 2A). Moreover, we noted less macrophage infiltration in the CNV lesions of Sult2b1−/− mice than in those of WT mice according to F4/80 staining of RPE–choroid flat mounts (Fig 2B and C), suggesting that SULT2B1+ macrophages are involved in the laser-induced CNV.

To verify the hypothesis, we depleted macrophages in WT and Sult2b1−/− mice by intraperitoneal injection (IP) of a Csf1r and c-Kit inhibitor (pexidartinib, PLX3397) that selectively induces macrophage death (26). After a continuous 7-d treatment with PLX3397, we observed a significant reduction (P < 0.01) in macrophages in the spleen compared with the untreated group (Fig 2D and E). After PLX3397 treatment, there was no significant difference in CNV area between Sult2b1−/− and WT mice (Fig 2F–I). Therefore, these results suggest that SULT2B1+ macrophages contribute to laser-induced CNV.

Sult2b1 drives M2 polarization to promote pathological angiogenesis

Given that alternatively activated M2 macrophages can promote pathological angiogenesis (16, 17), we investigated whether Sult2b1 expression affects M2 macrophage polarization in vivo and in vitro. Immunofluorescence analysis of CNV lesions showed that the percentage of M2 macrophages (indicated by YM1 or ARG1 positivity) was lower in the CNV lesions of Sult2b1−/− mice than in those of WT mice (Fig 3A–D). Then, we applied BMDMs in vitro and employed IL-4 to induce M2 polarization (27). Consistently, the SULT2B1 protein level in BMDMs showed a marked increase after IL-4 stimulation (Figs 3E and S2), which indicated that Sult2b1 might play a role in M2 polarization. Moreover, YM1 and ARG1 protein levels were significantly lower in Sult2b1−/− BMDMs than in WT BMDMs after the induction of M2 polarization (Fig 3F). Taken together, these results indicate that Sult2b1 deletion inhibits differentiation to M2 macrophages.

To test our hypothesis that Sult2b1 is related to M2-mediated vessel formation in CNV lesions, we measured the angiogenic activity of M2 BMDMs harvested from Sult2b1−/− and WT mice. To this end, we incubated C166 cells and HUVECs with the conditioned medium of WT or Sult2b1−/− M2 BMDMs and performed tube formation assays to assess vessel formation. The M2 BMDMs from Sult2b1−/− mice formed significantly fewer vessels in the three-dimensional (3D) matrix than those from WT mice (Fig 3G and I), consistent with the in vivo CNV results. Thus, Sult2b1 deficiency negatively affects the ability of macrophages to regulate pathological angiogenesis by inhibiting M2 polarization.

Figure 2. SULT2B1-expressing macrophages are involved in laser-induced CNV. (A) Immunostaining of F4/80 (green) and SULT2B1 (red) in retinal sections from WT and Sult2b1−/− mice 7 d after laser application (n = 5 per group). Blue indicates DAPI. INL, inner nuclear layer; ONL, outer nuclear layer; RPE, retinal pigment epithelium. (B, C) Immunostaining of F4/80 (green) in the flat-mounted RPE–choroid complexes (B) and quantification of macrophage infiltration in the CNV lesions (C) of WT and Sult2b1−/− mice (n = 5 per group). Scale bars, 100 μm. (D, E) Flow cytometry dot plots (D) and a bar graph (E) of F4/80+ cells collected from the spleens of mice treated with or without PLX3397 (n = 5 per group). **P < 0.01. (F) and quantification (G) of the leakage area (red circle) of PLX3397 (a Csf1r and c-Kit inhibitor, 16 mg/kg, IP)-treated WT and Sult2b1−/− mice (n = 10 per group). Scale bars, 100 μm. (H, I) Representative images from the fundus fluorescein angiography analysis (H) and quantification (I) of the leakage area (yellow circle) in WT and Sult2b1−/− mice after PLX3397 treatment (n = 10 per group). Scale bars, 500 μm. Data information: in (C, E, G, I), data are presented as the mean ± SD. **P < 0.01, ****P < 0.0001, no significance (ns) (unpaired two-tailed t test).
Figure 3. Sult2b1 drives macrophages towards M2 polarization and promotes neovascularization. (A, B) Representative images of double staining for F4/80 (green) and YM1 (red) (A) and quantification (B) of YM1-positive macrophages in CNV lesions of WT and Sult2b1−/− mice (n = 6 per group). (C, D) Representative images of double staining for F4/80 (green) and ARG1 (red) (C) and quantification (D) of ARG1-positive macrophages in CNV lesions of WT and Sult2b1−/− mice (n = 6 per group). Blue indicates DAPI. INL, inner nuclear layer; ONL, outer nuclear layer; RPE, retinal pigment epithelium. (E) Western blot analysis of SULT2B1 in WT BMDMs treated with or without IL-4. GAPDH was used as a loading control. (F) Western blot analysis of YM1 and ARG1 in BMDMs from WT and Sult2b1−/− mice treated with or without IL-4. β-Actin was used as a loading control. (G) Representative images of tube formation by C166 cells cultured with conditioned medium from WT or Sult2b1−/− M2 BMDMs. Scale bar, 100 μm. (H) Quantification of tube formation by C166 cells (n = 5). (I, J) Representative images of tube formation by HUVECs cultured with a conditioned medium from WT or Sult2b1−/− M2 BMDMs. Scale bar, 100 μm. (I) Quantification of tube formation by HUVECs (I) (n = 5). Data information: in (B, D, H, J), data are presented as the mean ± SD. **P < 0.01, ***P < 0.001, ****P < 0.0001 (unpaired two-tailed t test). Source data are available for this figure.
Cholesterol efflux mediated by LXR–ABCA1/G1 signalling is increased in Sult2b1−/− macrophages

Having pinpointed that Sult2b1 is involved in M2 polarization, we next sought to explore the mechanism by which Sult2b1 skew macrophage polarization towards the M2 phenotype during CNV development. LXR, a key regulator of cholesterol homeostasis, can be antagonized by Sult2b1 through the sulfation of oxysterols (23). In addition, the induction of efficient cholesterol efflux from senescent macrophages via treatment with an LXR agonist can regulate M2 polarization and pathological angiogenesis (28, 29). Hence, we speculated that Sult2b1 regulates pathological angiogenesis through LXR signalling. To test this hypothesis, we carried out immunofluorescence analyses of macrophages from Sult2b1−/− CNV lesions and found that LXRα and LXRβ protein levels were elevated (Fig 4A and B). Furthermore, the proteins encoded by LXR target genes, including the cholesterol reverse transporters ATP-binding cassette subfamily members A1 (ABCA1) and G1 (ABCG1), were at significantly higher levels in macrophages from Sult2b1−/− CNV lesions than in those from WT lesions, as shown by immunofluorescence (Fig 4C and D). Consistently, LXRα, LXRβ, ABCA1, and ABCG1 protein levels were increased in Sult2b1−/− M2 BMDMs (Fig 4E). Therefore, signalling through LXR is suppressed by Sult2b1 deficiency in macrophages.

Moreover, GSK2033 treatment resulted in decreased protein levels of LXRα, LXRβ, ABCA1, and ABCG1 in WT BMDMs (Fig 6A and B) and increased the protein levels of YMI and ARG1 in WT M2 BMDMs (Fig 6C and D), indicating that LXR inhibition could prevent M2 polarization regardless of Sult2b1 expression status. Notably, the expression levels of LXRs, LXR targets, and M2 markers in Sult2b1−/− macrophages treated with the LXR antagonist did not significantly differ from those in treated WT macrophages (Fig 6A–D). We also examined the effect of the LXR antagonist on the tube-formation ability of WT and Sult2b1−/− macrophages in vitro. The result showed that there was no significant difference in forming vessels between WT and Sult2b1−/− macrophages treated by GSK2033 (Figs 6E and F and S3A and B), which were consistent with those observed in vivo (Fig S5 and H). Further analysis of cholesterol content and cholesterol efflux revealed that treatment with the LXR antagonist reversed both the decrease in cholesterol content and the increase in cholesterol efflux in Sult2b1-deficient macrophages (Fig 6G and H). It may suggest that the effect of Sult2b1 on cholesterol efflux capacity relies on LXRs. Overall, our data suggest that the anti-angiogenic characteristics of Sult2b1−/− macrophages are most likely attributable to the activation of LXRs.

Sls deficiency aggravates CNV by promoting M2 macrophage differentiation

In contrast to Sult2b1, Sls catalyses the conversion of sulfated oxysterol to oxysterol (30, 31). Therefore, we sought to determine whether the loss of Sls plays an opposing role in the pathological angiogenesis of CNV. To accomplish this, we performed laser photocoagulation in WT- and Sls−/− mice (Fig 5A) and examined CNV lesions and infiltrating macrophages. Sls and SULT2B1 were specifically expressed in macrophages within the CNV lesion (Fig 5A). Notably, the percentage of M2 macrophages (indicated by YM1 or ARG1 positivity) in Sls−/− CNV lesions was higher than that in WT lesions (Fig 5F). Moreover, the number of infiltrating macrophages was significantly increased in the CNV lesions of Sls−/− mice, as determined by analysis of RPE–choroid flat mounts (Fig S5A and B). The percentage of M2 macrophages (indicated by YMI or ARG1 positivity) in Sls−/− CNV lesions was higher than that in WT lesions (Fig S5C–E). In addition, the expression levels of LXRs and their targets ABCA1 and ABCG1 in macrophages were lower in CNV lesions from Sls−/− mice than in those from WT mice (Fig S6A–D). These results demonstrate that Sls protects against CNV development by activating LXRA/ABCA1/G1 signalling to block M2 macrophage differentiation, indicating that its role is opposite to that of Sult2b1.

Discussion

The findings of our present study demonstrate that Sult2b1 plays an important role in CNV development by promoting M2 polarization. Specifically, we found that Sult2b1 deficiency leads to enhanced cholesterol efflux via activation of oxysterol-derived LXR signalling.
and that treatment with an LXR antagonist reverses these changes, resulting in decreased cholesterol efflux and increased cholesterol accumulation in the Sult2b1-/- macrophage. Conversely, SIs, which converts sulfated oxysterols to oxysterols, has a protective role against pathological neovascularization. These results shed light on the involvement of the cholesterol metabolism axis in macrophage polarization in nAMD, which may serve as a promising new therapeutic strategy in nAMD.

SULT2B1 has been previously shown to be expressed in multiple cell types, including hepatocytes, lymphocytes, and endothelial cells, in different tissues (32, 33). Here, we showed that SULT2B1 is specifically expressed in the infiltrating macrophages of CNV...
lesions. Our previous work assessing BMDM differentiation in a mouse model of ischaemic stroke suggested that Sutl2b1 inhibited pro-inflammatory (M1) macrophage polarization (24). In this study, we found that Sutl2b1 promoted proangiogenic (M2) macrophage polarization in a laser-induced CNV mouse model. Collectively, our data indicated that the effect of Sutl2b1 on macrophage polarization and function depends on the pathological context.

Oxysterols are the natural ligands of the LXR nuclear receptors and play crucial roles in cholesterol homeostasis (31, 34). SULT2B1-mediated sulfation of oxysterols and STS-mediated desulfation of sulfated oxysterols are opposing processes that can inactivate and activate LXRs, respectively. Previous studies suggest pivotal roles for SULT2B1/STS-mediated oxysterol-derived LXR signalling in the cholesterol metabolism of the

Figure 5. LXR antagonist treatment decreases the capacity of Sult2b1−/− macrophages to regulate CNV in vivo. (A, B, C, D) Macrophages (F4/80+, green) in CNV lesions of WT and Sult2b1−/− mice with GSK2033 (the LXR antagonist, 500 ng/μl, IV) were stained for (A) LXRα (red), (B) LXRβ (red), (C) ABCA1 (red), and (D) ABCG1 (red) (n = 5 per group). Scale bar, 100 μm. (E) Representative images of fundus fluorescein angiography and quantification of the leakage area (red circle) in WT and Sult2b1−/− mice (n = 10 per group) with or without GSK2033 treatment. Scale bar, 500 μm. (F) Representative images of isolectin (green) staining and quantification of CNV area (yellow circle) in RPE–choroid flat mounts from WT and Sult2b1−/− mice (n = 10 per group) with or without GSK2033 treatment. Scale bar, 500 μm. Data information: in (A, B, C, D, E, F), data are presented as the mean ± SD. **P < 0.01, ***P < 0.001, ****P < 0.0001, no significance (unpaired two-tailed t test).
Figure 6. LXR antagonist treatment reverses the capacity of Sult2b1−/− macrophages to regulate neovascularization in vitro. 
(A) Western blot analysis of LXRα, LXRβ, ABCA1, and ABCG1 levels in WT and Sult2b1−/− M2 BMDMs treated with or without GSK2033. (B) Relative protein expression of LXRα, LXRβ, ABCA1, and ABCG1 (n = 3). (C) Western blot analysis of YM1 and ARG1 levels in WT and Sult2b1−/− BMDMs with or without GSK2033 treatment. (D) Relative protein expression of YM1 and ARG1 (n = 3). (E) Tube formation analysis of C166 cells cultured with a conditioned medium from BMDMs (treated with or without GSK2033) (n = 5). (F) Tube formation analysis of HUVECs cultured with conditioned medium from BMDMs (treated with or without GSK2033) (n = 5). (G) The intracellular cholesterol...
liver but of the eye (30, 35). In our study, knockout of Sult2b1 activated LXR-ABCA1/G1-mediated cholesterol efflux, subsequently inhibiting M2 polarization and attenuating pathologic neovascularization. However, Sts deficiency had the opposite effect on CNV development. In conclusion, our study is the first to explore the roles and mechanisms of Sult2b1 and Sts in nAMD.

ABCA1/G1 facilitates cholesterol efflux to high-density lipoprotein particles for formation and maturation, thereby maintaining cholesterol homoeostasis (36). ABCA1 and ABCG1 are highly expressed in macrophages, especially in response to excess cholesterol accumulation (37, 38). Efficient cholesterol efflux mediated by the cholesterol transporters ABCA1/G1 in senescent macrophages can protect against pathologic angiogenesis (28). One study showed that conditional knockout of Abca1 and Abcg1 in macrophages leads to age-associated cholesterol-rich deposits underneath the retina, suggesting that impaired cholesterol efflux in macrophages initiates the development of AMD (39).

Our study showed that the expression levels of ABCA1 and ABCG1 were increased in Sult2b1−/− macrophages, wherein these proteins promoted cholesterol transport and reduced cholesterol accumulation in CNV lesions. Treatment of Sult2b1−/− macrophages with an LXR antagonist reversed all these phenotypes, resulting in decreased cholesterol efflux capacity and increased cholesterol accumulation. In short, our result suggests that the antiangiogenic characteristics of Sult2b1−/− macrophages are most likely dependent on LXR–ABCA1/G1 signalling.

In summary, the current study demonstrates that Sult2b1 deficiency attenuates CNV by inhibiting M2 macrophage polarization and promoting cholesterol efflux via the LXR–ABCA1/G1 pathway. Our findings shed light on the regulation of macrophage polarization and pathologic neovascularization through cholesterol metabolism.

Materials and Methods

Animals

Animal procedures were approved by the Shanghai Jiao Tong University Institutional Review Board. All animal experiments conformed to the Association for Research in Vision and Ophthalmology (ARVO) Statement for the Use of Animals in Ophthalmic and Vision Research. The C57BL/6j mice were provided by the Laboratory Animal Center of Shanghai General Hospital. The Sult2b1−/− mice were provided by Shanghai Institute of Immunology, as described in a previous study (40). The Sts−/− mice were generated by deleting 91 bp in exon 2 of the Sts gene as described in Fig S4. The target ending with NGG near exon 2 was designed and cut under the action of CRISPR/Cas9. The sgRNA1 sequence of sgRNA1 was GAGGATGATGTCAGCGAGCGTGG, and the sgRNA2 sequence of sgRNA2 was AGCCGAGGTACCCGATGCCAGG. Primer sequences for genotyping are presented in Table S1. Male mice aged between 6 and 8 wk and ~20 g were included in the experiment.

Laser-induced CNV

Mice were anaesthetized with an intraperitoneal injection of 1% pentobarbital sodium (0.1 ml/10 g body weight) after the application of tropicamide (Santen) for pupil dilatation. CNV lesions were induced as described in a previous study (11). Briefly, four laser spots were administered around the optic nerve head by an argon laser (120 mW in intensity, 50 μm in size, and 100 ms in duration) (OcuLight Infrared Laser System 810 nm; Iridex Corp.) in each eye. CNV lesion area was determined by FFA and immunofluorescence in RPE–choroid flat mounts on day 7 after injury.

Intravitreal injection

Mice were anaesthetized as described above. After laser treatment, the mice were immediately divided into two groups for intravitreal injection. For local anaesthesia, benoxinate HCl 0.4% drops (Santen) were administered. Subsequently, 2 μl of either GSK2033 (LXR antagonist, 500 ng/μl) or vehicle (PBS) was delivered into the vitreous cavity using a microsyringe (Hamilton). The operation was conducted under a dissecting microscope (Olympus) in a sterilized environment. After the operation, 0.5% levofloxacin eye drops (Santen) were applied to prevent infection.

FFA

Mice were anaesthetized, and their pupils were dilated as described above. Then, fluorescein sodium (1%, 0.1 ml/kg; Alcon) was injected intraperitoneally, and late-phase angiograms were performed 5 min after the injection. Fluorescein angiography (FA) was performed by a Micron IV System (Phoenix Research Laboratories). The area of each CNV lesion with leakage was quantified using ImageJ software (National Institutes of Health).

Macrophage depletion

Pexidartinib (16 mg/kg, PLX3397; MedChemExpress) was injected intraperitoneally twice daily for seven consecutive days before laser photocoagulation. No obvious behavioural or health problems were observed in animals injected with PLX3397. Splenocytes were collected and stained with F4/80 (123108; BioLegend) on day 7 after PLX3397 treatment. Because cessation of PLX3397 treatment induces macrophage regeneration (41), we administered PLX3397 for 7 d after laser injury.

Cell culture

HUVECs, from ATCC were grown in humidified air (37°C, 5% CO2) in an endothelial cell medium (1001; Solarbio) supplemented with 5%
Figure 7. Sts deficiency promotes pathological angiogenesis.

(A) Cryosections of WT and Sts\(^{-/-}\) mouse CNV lesions stained for F4/80 (green) and STS (red). Blue indicates DAPI. INL, inner nuclear layer; ONL, outer nuclear layer; RPE, retinal pigment epithelium. (B, C) Representative images of fundus fluorescein angiography and leakage area (red circle) (B) and quantification of the leakage area (C) in WT and Sts\(^{-/-}\) mice (n = 10 per group). (D, E) Representative images of isolectin (green) staining and CNV area (yellow circle) in RPE–choroid flat mounts (D) and quantification of the CNV area (E) in WT and Sts\(^{-/-}\) mice (n = 10 per group). (F) Relative mRNA levels of proangiogenic growth markers (Vegfa, Pdgfb, and Tek), an endothelial cell marker (Tek), and profibrotic factors (Tgfb1 and Pdgfb) in the RPE–choroid flat mounts of WT and Sts\(^{-/-}\) mice (n = 4 per group). Data information: In (C, E, F), data are presented as the mean ± SD. **P < 0.01, ***P < 0.001, ****P < 0.0001 (unpaired two-tailed t test).
FBS, 1% penicillin-streptomycin antibiotics and 1% endothelial cell growth supplement. C166 cells (from ATCC) and BMDMs were cultured in DMEM/high glucose (SH30243.01; Hyclone) with 10% FBS and 1% penicillin-streptomycin antibiotics.

**BMDM polarization**

Bone marrow cells were harvested from the femur and tibia and then differentiated with 10 ng/ml recombinant mouse M-CSF (PMC2044; Thermo Fisher Scientific) for 7 d (24). For M2 polarization, the macrophages were stimulated with 10 ng/ml recombinant mouse IL-4 (574302; BioLegend) (42) for 24 h. The conditioned medium and cells were collected separately for the following use.

**Endothelial cell tube formation assay**

Matrigel matrix (354234; Corning) was added to a 96-well plate (50 μl/well) and allowed to polymerize for 30 min at 37°C. Then, 1 × 10⁴ C166 cells or HUVECs and conditioned medium from BMDMs (described above) were mixed and seeded on the Matrigel matrix. After 6 h of incubation at 37°C, tube formation was photographed using a light microscope at 10× magnification. Automated analysis was performed with the Image software (National Institutes of Health, Bethesda, MD, USA). Three parameters (number [Nb] of master junctions, Nb of meshes, and total length) were taken for the quantification experiments.

**Cholesterol quantification assay**

The free-cholesterol content of M2 BMDMs was measured using the cholesterol/cholesteryl ester quantitation assay kit (ab65359; Abcam). In brief, the dried lipids (extracted from 1 × 10⁶ M2 BMDMs) were dissolved in 200 μl assay buffer (supplied in the kit), and then incubated with a cholesterol probe/cholesterol enzyme mix solution (supplied in the kit) at 37°C for 1 h protected from light. Finally, the fluorescence was detected by a fluorescent microplate reader (Ex/Em = 535/587 nm). The free-cholesterol content was quantified using a cholesterol standard curve.

**Cholesterol efflux assay**

The cholesterol efflux was measured using the commercial cholesterol efflux assay kit (cell-based) (ab196985; Abcam). In brief, M2 BMDMs (1 × 10⁵) were incubated with a labelling reagent for 1 h and then with the Equilibration Buffer overnight. The cells were treated with the desired cholesterol acceptor (serum) for 6 h in an incubator (37°C, 5% CO₂). The fluorescence intensity of the media and cell lysates was measured via a fluorescent microplate reader (Ex/Em = 482/515 nm). The cholesterol efflux was calculated by dividing the fluorescence intensity of the media by the total fluorescence intensity of the cell lysate and media.

**Immunofluorescence**

Eyeballs were harvested from mice that were transcardially perfused with 4% paraformaldehyde and then prepared for RPE-choroid flat mounts or cryosections. The antibodies used for staining were anti-isolectin (FL-1201, 1:500; Vector Labs), anti-F4/80 (ab6640, 1:500; Abcam), anti-SULT2B1 (ab254616, 1:500; Abcam), anti-YM1 (1404, 1:500; Stem Cell), anti-ARG1 (156001-1-AP, 1:500; Proteintech), anti-LXRα (ab176323, 1:100; Abcam), anti-LXRβ (ab28479, 1:200; Abcam), anti-ABCA1 (ab18180, 1:200; Abcam), anti-ABCG1 (ab52617, 1:100; Abcam), and anti-STS (17870-1-AP, 1:500; Proteintech). Images were visualized by a fluorescence microscope (Olympus). ImageJ (National Institutes of Health, Bethesda, MD, USA) was used for analysis and quantification.

**RNA extraction and real-time PCR**

Total RNA was extracted from RPE-choroidal tissues (with CNV lesions) using a total RNA extraction kit (Tiangen). RNA quantity and quality were assessed with a NanoDrop spectrophotometer (Thermo Fisher Scientific). cDNA was synthesized with a cDNA synthesis kit (Takara Bio), and gene expression was analysed by real-time PCR (Applied Biosystems). Primer sequences are presented in Table S2. The mRNA levels of the target genes were quantitatively normalized against those of Gapdh using the ΔΔCT method.

**Western blot analysis**

Harvested BMDMs described above were homogenized with lysis buffer. The samples were resolved by 10% SDS-PAGE and transferred to PVDF membranes. After blocking with 5% nonfat dry milk in TBST (TBS containing Tween-20) for 1 h at room temperature, the membranes were incubated at 4°C overnight with primary antibodies against GAPDH (10494-1-AP, 1:1,000; Proteintech), β-actin (3779, 1:1,000; Prosci), YM1 (1404, 1:1,000; Stem Cell), ARG1 (16001-1-AP, 1:5,000; Proteintech), SULT2B1 (ab254616, 1:1,000; Abcam), LXRα (ab176323, 1:1,000; Abcam), LXRβ (ab28479, 1:1,000; Abcam), ABCA1 (ab18180, 1:500; Abcam) or ABCG1 (ab52617, 1:1,000; Abcam). The membranes were washed three times with TBST and then incubated with the corresponding horseradish peroxidase-conjugated secondary antibodies for 1 h at room temperature. Finally, the membranes were washed three times with TBST, and the bands were visualized by chemiluminescence using a molecular imaging system (Amersham Imager 600; GE Healthcare). GAPDH or β-actin was used as loading control. The experiment was replicated three times using independent biological samples.

**Statistical analysis**

Data are shown as the mean ± SD. Each experiment was repeated three times. Differences among groups were analysed using unpaired two-tailed t test (Prism v7.0; Graph-Pad Software, Inc). *P* < 0.05 was set as the criterion for significance.

**Data Availability**

The datasets generated during and/or analysed during the current study are available from the corresponding author on reasonable request.
Supplementary Information

Supplementary Information is available at https://doi.org/10.26508/lsa.20230020.

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Author Contributions

Y Wang: data curation, formal analysis, methodology, and writing—original draft.
Y Liu: formal analysis and validation.
Y Wang: formal analysis and validation.
Y Wu: formal analysis and investigation.
Z Chen: formal analysis and investigation.
F Wang: conceptualization, data curation, validation, investigation, project administration, and writing—review and editing.
X Wan: conceptualization, data curation, and writing—original draft, review, and editing.
F Wang: project administration.
X Sun: project administration.

Conflict of Interest Statement

The authors declare that they have no conflict of interest.

References


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